

An overview of the management of established nonindigenous species in the Great Lakes

Rochelle Sturtevant¹
Lauren Berent²
Thomas Makled²
Whitney Conard²
Abigail Fusaro³
Edward Rutherford⁴

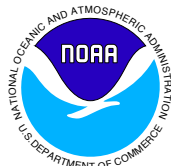
¹NOAA Sea Grant, 4840 S. State Rd., Ann Arbor, MI 48108

²Cooperative Institute for Limnology and Ecosystems Research, 4840 S. State Rd., Ann Arbor, MI 48108

³Wayne State University, 42 W. Warren Ave., Detroit, MI 48202

⁴NOAA Great Lakes Environmental Research Laboratory, 4840 S. State Rd., Ann Arbor, MI 48108

February 2016



UNITED STATES
DEPARTMENT OF COMMERCE

Penny Pritzker
Secretary

NATIONAL OCEANIC AND
ATMOSPHERIC ADMINISTRATION

Kathy Sullivan
Acting, Under Secretary for Oceans & Atmosphere
NOAA Administrator

NOTICE

Mention of a commercial company or product does not constitute an endorsement by the NOAA. Use of information from this publication concerning proprietary products or the tests of such products for publicity or advertising purposes is not authorized. This is GLERL Contribution No. 1795.

This publication is available as a PDF file and can be downloaded from GLERL's web site: www.glerl.noaa.gov. Hard copies can be requested from GLERL Information Services, 4840 S. State Rd., Ann Arbor, MI 48108. pubs.glerl@noaa.gov.

NOAA's Mission – To understand and predict changes in Earth's environment and conserve and manage coastal and marine resources to meet our nation's economic, social, and environmental needs

NOAA's Mission Goals:

- Protect, restore and manage the use of coastal and ocean resources through an ecosystem approach to management
- Understand climate variability and change to enhance society's ability to plan and respond
- Serve society's needs for weather and water information
- Support the Nation's commerce with information for safe, efficient, and environmentally sound transportation
- Provide critical support for NOAA's Mission

TABLE OF CONTENTS

1.	Introduction.....	11
2.	Methods	12
3.	Nonindigenous Species Management.....	16
3.1	Regulations Targeting Pathways	17
3.2	Federal Regulations Targeting Specific Species.....	17
3.3	State Regulations Targeting Specific Species	18
3.3.1	Fish pathogens.....	18
3.3.2	Fish.....	21
3.3.3	Mollusks.....	26
3.3.4	Waterfleas and Mysids.....	27
3.3.5	Plants	29
3.3.6	Miscellaneous taxa covered only under the whitelist approach – Algae and Aquatic Invertebrates.....	33
3.4	Unregulated Species.....	34
4.	Nonindigenous Species Control	35
4.1	Algae	35
4.2	Plants	36
4.3	Fish.....	56
4.4	Mollusks.....	62
4.5	Insects.....	65
4.6	Free-Living Crustaceans, including Waterfleas, Copepods, Mysids and Gammarids.....	65
4.7	Free-Living Worms including Annelids and Platyhelminthes	69
4.8	Bryozoans, Hydrozoans and Testate Amoebae.....	70
4.9	Parasites and Diseases.....	71

4.9.1 Parasitic Platyhelminthes	71
4.9.2 Parasitic copepods.....	72
4.9.3 Protozoan Parasites.....	73
4.9.4 Bacteria.....	75
4.9.5 Viruses	77
5. Discussion	78
6. Literature Cited.....	80
7. Acknowledgements	129
Appendix A. Species Management Profiles	131
A.1 Algae	131
<i>Actinocyclus normanii</i> f. <i>subsalsa</i> (Juhlin-Dannfelt) Hustedt, 1957	131
<i>Bangia atropurpurea</i> (Roth) Agardh, 1824.....	131
<i>Chaetoceros muelleri subsalsum</i> J. R. Johansen and Rushforth, 1985.....	132
<i>Chroodactylon ornatum</i> (C. Agardh) Basson, 1979.....	132
<i>Contricribra guillardii</i> (Hasle) K. Stachura-Suchoples & D.M. Williams.....	132
<i>Cyclotella atomus</i> Hustedt, 1937	132
<i>Cyclotella cryptica</i> Reimann, Lewin, and Guillard, 1963.....	133
<i>Cylindrospermopsis raciborskii</i> (Wolosz.) Seenayya and Subbaraju, 1972	133
<i>Diatoma ehrenbergii</i> Kützing, 1844	134
<i>Discostella pseudostelligera</i> (Hustedt) Houk and Klee, 1939.....	134
<i>Discostella woltereckii</i> Hustedt, 1942.....	135
<i>Hymenomonas roseola</i> Stein, 1878	135
<i>Nitellopsis obtusa</i> (Desvaux in Loiseleur) J. Groves, (1919).....	135
<i>Pleurosira laevis</i> (Ehrenberg) Compère, (1843) 1982.....	135

<i>Skeletonema potamos</i> (Weber) Hasle in Hasle & Evensen, (1970)	136
<i>Skeletonema subsalsum</i> (Cleve-Euler) Bethge, (1912) 1928	136
<i>Sphacelaria fluviatilis</i> Jao, 1943	136
<i>Sphacelaria lacustris</i> Schloesser and Blum, 1980	136
<i>Stephanodiscus binderanus</i> Krieger, 1927	136
<i>Stephanodiscus subtilis</i> (Van Goor) A. Cleve, 1951	137
<i>Thalassiosira baltica</i> Ostenfeld, 1901	137
<i>Thalassiosira lacustris</i> (Grunow) Hasle in Hasle and Fryxell, 1977	137
<i>Thalassiosira pseudonana</i> (Hustedt) Hasle and Heimdal, (1957) 1970	138
<i>Thalassiosira weissflogii</i> (Grunow) G. Fryxell & Hasle, (1896) 1977	138
<i>Ulva (Enteromorpha) flexuosa</i> subsp. <i>flexuosa</i> and <i>flexuosa</i> subsp. <i>paradoxa</i> (Wolfen ex Roth) J. Agardh, 1883	138
<i>Ulva (Enteromorpha) intestinalis</i> Linnaeus, 1753	139
<i>Ulva (Enteromorpha) prolifera</i> O.F. Müller, 1778	139
A.2 Annelids	139
<i>Branchiura sowerbyi</i>	139
<i>Ganius aquaedulcis</i>	140
<i>Potamothrix bedoti</i> (Piquet, 1913)	141
<i>Potamothrix moldaviensis</i> Vejdovsky and Mrazek, 1902	141
<i>Potamothrix vejdoskyi</i> Hrabe, 1941	142
<i>Ripistes parasita</i>	143
A.3 Bacteria	143
<i>Aeromonas salmonicida</i> Emmerich and Weible, 1890	143
<i>Piscirickettsia cf. salmonis</i>	146
<i>Renibacterium (Corynebacterium) salmoninarum</i> Sanders and Fryer, 1980	146

A.4 Bryozoa	148
<i>Lophopodella carteri</i> (Hyatt, 1865)	148
A.5 Coelenterates.....	149
<i>Cordylophora caspia</i> (Pallas, 1771)	149
<i>Craspedacusta sowerbyi</i> Lankester, 1880.....	149
A.6 Amphipods.....	150
<i>Echinogammarus ischnus</i> (Stebbing, 1899)	150
<i>Gammarus tigrinus</i> Sexton 1939.....	151
A.7 Cladocerans	151
<i>Bosmina coregoni</i> Baird, 1857	151
<i>Bythotrephes longimanus</i>	152
<i>Cercopagis pengoi</i>	153
<i>Daphnia galeata galeata</i> Sars, 1864	155
<i>Daphnia lumholtzi</i>	156
<i>Eubosmina maritima</i> P.E. Müller, 1867	156
A.8 Copepods	157
<i>Argulus japonicus</i> Thiele, 1900	157
<i>Cyclops strenuus</i> Fischer, 1851.....	157
<i>Eurytemora affinis</i> Poppe, 1880	158
<i>Heteropsyllus</i> nr. <i>nunni</i> Coull	159
<i>Megacyclops viridis</i> Jurine, 1820	159
<i>Neoergasilus japonicus</i> Harada, 1930.....	160
<i>Nitokra hibernica</i> Brady, 1880	160
<i>Nitokra incerta</i> Richard, 1893	161

<i>Salmincola lotae</i> Olsson, 1877	161
<i>Schizopera borutzkyi</i> Monchenko, 1967	162
<i>Skistodiaptomus pallidus</i> Herrick, 1879	163
A.9 Mysids	163
<i>Hemimysis anomala</i> G.O. Sars, 1907	163
A.10 Fishes	164
<i>Alosa aestivalis</i> Mitchill, 1814.....	164
<i>Alosa pseudoharengus</i> Wilson, 1811.....	165
<i>Apeltes quadracus</i> Mitchill, 1815.....	166
<i>Carassius auratus</i> Linnaeus, 1758	167
<i>Cyprinus carpio</i> Linnaeus, 1758.....	169
<i>Enneacanthus gloriosus</i> Holbrook, 1855	170
<i>Esox niger</i> Lesueur, 1818.....	172
<i>Gambusia affinis</i> Baird and Girard, 1853	173
<i>Gymnocephalus cernua</i> Linnaeus, 1758	173
<i>Ictiobus cyprinellus</i> Valenciennes in Cuvier and Valenciennes, 1844.....	176
<i>Lepisosteus platostomus</i> Rafinesque, 1820.....	176
<i>Lepomis microlophus</i> Günther, 1859	177
<i>Misgurnus anguillicaudatus</i> Cantor, 1842.....	178
<i>Morone americana</i> Gmelin, 1789	179
<i>Neogobius melanostomus</i> Pallas, 1814	180
<i>Notropis buchanani</i> Meek, 1896.....	181
<i>Oncorhynchus gorbuscha</i> Walbaum, 1792	182
<i>Oncorhynchus kisutch</i> Walbaum, 1792.....	184

<i>Oncorhynchus mykiss</i> Walbaum, 1792	186
<i>Oncorhynchus nerka</i> (Walbaum in Artedi, 1792).....	188
<i>Oncorhynchus tshawytscha</i> Walbaum in Artedi, 1792.....	189
<i>Osmerus mordax</i> Mitchill, 1814	191
<i>Petromyzon marinus</i> (Linnaeus, 1758)	193
<i>Phenacobius mirabilis</i> (Girard, 1856).....	195
<i>Proterorhinus semilunaris</i> (Heckel, 1837).....	195
<i>Salmo trutta</i> Linnaeus, 1758	197
<i>Scardinius erythrophthalmus</i> Linnaeus, 1758	199
A.11 Insects.....	200
<i>Acentria ephemerella</i> Olivier, 1791	200
<i>Tanytarsus lemnae</i> Paykull/Fabricius, 1792	200
A.12 Mollusks.....	201
<i>Corbicula fluminea</i> O. F. Müller, 1774	201
<i>Dreissena polymorpha</i> Pallas, 1771	202
<i>Dreissena rostriformis bugensis</i> Andrusov, 1897	203
<i>Lasmigona subviridis</i> Conrad, 1835	205
<i>Pisidium amnicum</i> Müller, 1774	206
<i>Pisidium henslowanum</i> Shepard, 1825.....	206
<i>Pisidium moitessierianum</i> Paladilhe, 1866	207
<i>Pisidium supinum</i> Schmidt 1850	207
<i>Sphaerium corneum</i> Linnaeus, 1758.....	208
<i>Bithynia tentaculata</i>	209
<i>Cipangopaludina chinensis malleata</i>	209

<i>Cipangopaludina japonica</i>	210
<i>Elimia virginica</i>	210
<i>Gillia altilis</i>	211
<i>Potamopyrgus antipodarum</i>	211
<i>Radix auricularia</i>	212
<i>Valvata piscinalis</i>	213
<i>Viviparus georgianus</i> I. Lea, 1834	213
A.13 Plants	214
<i>Agrostis gigantea</i> Roth.....	214
<i>Alnus glutinosa</i> (L.) Gaertn.....	215
<i>Alopecurus geniculatus</i> L.	215
<i>Butomus umbellatus</i> L.....	216
<i>Cabomba caroliniana</i>	217
<i>Carex acutiformis</i> Ehrh.	219
<i>Carex disticha</i> Huds.	220
<i>Chenopodium glaucum</i>	220
<i>Cirsium palustre</i>	220
<i>Conium maculatum</i>	222
<i>Echinochloa crus-galli</i> L. P. Beauvois.....	224
<i>Epilobium hirsutum</i> L.....	226
<i>Frangula alnus</i> P. Mill.....	228
<i>Glyceria maxima</i>	230
<i>Hydrocharis morsus-ranae</i> L.	231
<i>Impatiens glandulifera</i>	232

<i>Iris pseudacorus</i>	233
<i>Juncus compressus</i> Jacq.....	235
<i>Juncus gerardii</i> Loisel.....	235
<i>Juncus inflexus</i> L.	236
<i>Lupinus polyphyllus</i>	237
<i>Lycopus asper</i>	237
<i>Lycopus europaeus</i> L.	237
<i>Lysimachia nummularia</i>	238
<i>Lysimachia vulgaris</i>	238
<i>Lythrum salicaria</i>	239
<i>Marsilea quadrifolia</i>	241
<i>Mentha aquatica</i> L.	242
<i>Mentha gracilis</i> Sole (pro sp.)	242
<i>Mentha spicata</i>	243
<i>Myosotis scorpioides</i> L.	243
<i>Myosoton aquaticum</i> (L.) Moench.....	243
<i>Myriophyllum spicatum</i> L.....	244
<i>Najas marina</i> L.....	246
<i>Najas minor</i> All.	247
<i>Nasturtium officinale</i>	247
<i>Nymphoides peltata</i>	248
<i>Pluchea odorata succulenta</i> (Fern.) Cronq.	249
<i>Pluchea odorata odorata</i> (L.) Cass.	249
<i>Poa trivialis</i> L.	249

<i>Polygonum persicaria</i> L.	250
<i>Potamogeton crispus</i>	251
<i>Puccinellia distans</i> (Jacq.) Parl.	252
<i>Rorippa sylvestris</i> (L.) Bess.	252
<i>Rumex longifolius</i> DC.	253
<i>Rumex obtusifolius</i>	254
<i>Salix alba</i>	255
<i>Salix fragilis</i>	256
<i>Salix purpurea</i>	257
<i>Solanum dulcamara</i>	258
<i>Solidago sempervirens</i> L.	258
<i>Sparganium glomeratum</i> (Laestad.) L. Neum.	259
<i>Trapa natans</i>	259
<i>Typha angustifolia</i> L.	260
<i>Veronica beccabunga</i>	261
A.14 Platyhelminthes	262
<i>Bothriocephalus acheilognathi</i> Yamaguti, 1934	262
<i>Dactylogyrus amphibothrium</i> Wagener or Wegener, 1857	262
<i>Dactylogyrus hemiamphibothrium</i> Ergens, 1956	263
<i>Dugesia polychroa</i> Schmidt, 1861	264
<i>Ichthyocotylurus pileatus</i>	264
<i>Neascus brevicaudatus</i> von Nordmann, 1832	265
<i>Scolex pleuronectis</i> Müller, 1788	265
<i>Timoniella</i> sp.	265

A.15 Protozoa.....	265
<i>Acineta nitocrae</i>	265
<i>Glugea hertwigi</i> Weissenberg, 1911	265
<i>Heterosporis</i> sp.	265
<i>Myxobolus cerebralis</i> Hofer, 1903	266
<i>Psammonobiotus communis</i>	269
<i>Psammonobiotus dziwnowi</i>	269
<i>Psammonobiotus linearis</i>	269
<i>Sphaeromyxa sevastopoli</i>	269
<i>Trypanosoma acerinae</i> Brumpt, 1906	269
A.16 Viruses	270
<i>Novirhabdovirus</i> sp. genotype IV sublineage b	270
<i>Ranavirus</i>	272
<i>Rhabdovirus carpio</i>	272

AN OVERVIEW OF THE MANAGEMENT OF ESTABLISHED NONINDIGENOUS SPECIES IN THE GREAT LAKES

Rochelle Sturtevant, Lauren Berent, Thomas Makled, Abigail Fusaro, and Ed Rutherford

1. Introduction

The Great Lakes are host to thousands of native fishes, invertebrates, plants, and other species that not only provide recreational and economic value to the region, but also hold important ecological value. However, with over 180 documented aquatic nonindigenous species¹ (ANS) and an apparent introduction rate estimated at 1.3-1.8 species·year⁻¹ through 2006, the Great Lakes basin is considered one of the most heavily invaded aquatic systems in the world (Mills et al. 1993, Ricciardi 2006, GLRI Task Force 2010). Some of these nonindigenous species may become invasive (i.e. “species whose introduction does or is likely to cause economic or environmental harm or harm to human health” ([Executive Order 13112, 1999](#))) and threaten the ecological and/or socio-economic value of the Great Lakes. In contrast, other nonindigenous species are capable of contributing value to the Great Lakes. Pacific salmonids, for instance, are stocked annually by the millions to help support the Great Lakes’ multi-billion dollar fishery (Kocik and Jones 1999, USFWS/GLFC 2010, USACE 2012a).

This study provides a snapshot of management of established nonindigenous species in the Great Lakes region as a benchmark for improving management practices. We review the relevant state and federal regulations that impact AIS management as well as management alternatives in a format that allows cross-jurisdictional and cross-taxa analysis. This effort is part of a larger project funded by the Great Lakes Restoration Initiative to enhance the Great Lakes Aquatic Nonindigenous Species Information System (GLANSIS¹), an online database containing information on the identification, distribution, ecology, impact, and management of all established ANS in the Great Lakes. Previously, we assessed the relative ecological and socioeconomic impacts of nonindigenous species (NOAA Technical Memorandum 161 – An Impact Assessment of Great Lakes Aquatic Nonindigenous Species; Sturtevant et al. 2014).

¹ These nonindigenous aquatic species have populations established in the Great Lakes basin below the ordinary high water mark, including connecting channels, wetlands, and waters ordinarily attached to the lakes (see definitions and criteria for listing in the Great Lakes Aquatic Nonindigenous Species Information System at <http://www.glerl.noaa.gov/res/Programs/glansis/glansis.html>).

2. Methods

This document is a synthesis of management data gathered for GLANSIS over the period 2010-2014. GLANSIS fact sheets were primarily researched and written by the authors of this manuscript with some additional support from students mentioned in the Acknowledgements. The 181 species-specific fact sheets, which include information on ecology, invasion history, impact, as well as management were written independently to a common template, but each was reviewed by the core management team for consistency as well as sent to external taxonomic experts (also listed in Acknowledgements) for review prior to posting in GLANSIS. The management sections of each fact sheet were extracted from the GLANSIS database and collectively form the ‘raw’ dataset (included in Appendix A), which is analyzed and summarized in this memo.

Table 1. Great Lakes Established Nonindigenous Species included in GLANSIS

Scientific Name	Common Name
Algae	
<i>Actinocyclus normanii</i> f. subsalsa	Diatom
<i>Bangia atropurpurea</i>	Red Alga
<i>Chaetoceros muelleri</i>	Diatom
<i>Chroodactylon ornatum</i>	Red Alga
<i>Contracribra guillardii</i>	Diatom
<i>Cyclotella atomus</i>	Diatom
<i>Cyclotella cryptica</i>	Diatom
<i>Cylindrospermopsis raciborskii</i>	Cylindro
<i>Diatoma ehrenbergii</i>	Diatom
<i>Discostella pseudostelligera</i>	Diatom
<i>Discostella woltereckii</i>	Diatom
<i>Hymenomonas roseola</i>	Coccolithophorid
<i>Nitellopsis obtusa</i>	Starry Stonewort
<i>Pleurosira laevis</i>	Diatom
<i>Skeletonema potamos</i>	Diatom
<i>Skeletonema subsalsum</i>	Diatom
<i>Sphacelaria fluviatilis</i>	Brown Alga
<i>Sphacelaria lacustris</i>	Brown Alga
<i>Stephanodiscus binderanus</i>	Diatom
<i>Stephanodiscus subtilis</i>	Diatom
<i>Thalassiosira baltica</i>	Diatom
<i>Thalassiosira lacustris</i>	Diatom
<i>Thalassiosira pseudonana</i>	Diatom
<i>Thalassiosira weissflogii</i>	Diatom
<i>Ulva flexuosa</i>	Grass Kelp
<i>Ulva intestinalis</i>	Grass Kelp
<i>Ulva prolifera</i>	Sea Lettuce

Annelids	
<i>Branchiura sowerbyi</i>	Tubificid Worm
<i>Gianius aquaedulcis</i>	Tubificid Worm
<i>Potamotheix bedoti</i>	Tubificid Worm
<i>Potamotheix moldaviensis</i>	Tubificid Worm
<i>Potamotheix vejovskyi</i>	Tubificid Worm
<i>Ripistes parvita</i>	Oligochaete
Bacteria	
<i>Aeromonas salmonicida</i>	Furunculosis
<i>Piscirickettsia cf. salmonis</i>	Muskie Pox
<i>Renibacterium salmoninarum</i>	Bacterial Kidney Disease (BKD)
Bryozoa	
<i>Lophopodella carteri</i>	Freshwater Bryozoan
Coelenterates	
<i>Cordylophora caspia</i>	Freshwater Hydroid
<i>Craspedacusta sowerbyi</i>	Freshwater Jellyfish
Amphipods	
<i>Echinogammarus ischnus</i>	Scud
<i>Gammarus tigrinus</i>	Amphipod
Cladocerans	
<i>Bosmina coregoni</i>	Waterflea
<i>Bythotrephes longimanus</i>	Spiny Waterflea
<i>Cercopagis pengoi</i>	Fishhook Waterflea
<i>Daphnia galeata mendotae</i>	Waterflea
<i>Daphnia lumholzi</i>	Waterflea
<i>Eubosmina meridionalis</i>	Cladoceran
Copepods	
<i>Argulus japonicus</i>	Japanese Fishlouse
<i>Cyclops strenuus</i>	Copepod
<i>Eurytemora affinis</i>	Calanoid Copepod
<i>Heterosyllus nr. ninni</i>	Harpacticoid Copepod
<i>Megacyclops virens</i>	Cyclopoid Copepod
<i>Neogamasius japonicus</i>	Parasitic Copepod
<i>Nitokra hibernica</i>	Harpacticoid Copepod
<i>Nitokra incerta</i>	Harpacticoid Copepod
<i>Salmincola lotae</i>	Parasitic Copepod
<i>Schizopera borutzkyi</i>	Oarsman
<i>Skistodiaptomus pallidus</i>	Calanoid Copepod
Mysids	
<i>Hemimysis anomala</i>	Bloody Red Shrimp
Fish	
<i>Alosa aestivalis</i>	Blueback Herring
<i>Alosa pseudoharengus</i>	Alewife
<i>Apeltes quadracus</i>	Fourspine Stickleback
<i>Carassius auratus</i>	Goldfish

<i>Cyprinus carpio</i>	Common Carp
<i>Enneacanthus gloriosus</i>	Bluespotted Sunfish
<i>Esox niger</i>	Chain Pickerel
<i>Gambusia affinis</i>	Western Mosquitofish
<i>Gymnocephalus cernua</i>	Ruffe
<i>Ictiobus cyprinellus</i>	Bigmouth Buffalo
<i>Lepisosteus platostomus</i>	Shortnose Gar
<i>Lepomis microlophus</i>	Redear Sunfish
<i>Misgurnus anguillicaudatus</i>	Oriental Weatherfish
<i>Morone americana</i>	White Perch
<i>Neogobius melanostomus</i>	Round Goby
<i>Notropis buchanani</i>	Ghost Shiner
<i>Oncorhynchus gorbuscha</i>	Pink Salmon
<i>Oncorhynchus kisutch</i>	Coho Salmon
<i>Oncorhynchus mykiss</i>	Rainbow Trout
<i>Oncorhynchus nerka</i>	Sockeye Salmon
<i>Oncorhynchus tshawytscha</i>	Chinook Salmon
<i>Osmerus mordax</i>	Rainbow Smelt
<i>Petromyzon marinus</i>	Sea Lamprey
<i>Phencobius mirabilis</i>	Suckermouth Minnow
<i>Proterorhinus semilunaris</i>	Tubenose Goby
<i>Salmo trutta</i>	Brown Trout
<i>Scardinius erythrophthalmus</i>	Rudd
Insects	
<i>Acentria ephemerella</i>	European Water Moth
<i>Tanysphyrus lemnae</i>	Duckweed Weevil
Mollusks	
<i>Corbicula fluminea</i>	Asian Clam
<i>Dreissena polymorpha</i>	Zebra Mussel
<i>Dreissena rostriformis bugensis</i>	Quagga Mussel
<i>Lasmigona subviridis</i>	Green Floater
<i>Pisidium amnicum</i>	Greater European Peaclam
<i>Pisidium henslowanum</i>	Henslow's Peaclam
<i>Pisidium moitessierianum</i>	Pygmy Peaclam
<i>Pisidium supinum</i>	Humpbacked Peaclam
<i>Sphaerium corneum</i>	European Fingernail Clam
<i>Bithynia tentaculata</i>	Mud Bithynia, Faucet Snail
<i>Cipangopaludina chinensis malleata</i>	Chinese Mysterysnail
<i>Cipangopaludina japonica</i>	Japanese Mysterysnail
<i>Elimia virginica</i>	Piedmont Elimia
<i>Gillia altilis</i>	Buffalo Pebblesnail
<i>Potamopyrgus antipodarum</i>	New Zealand Mudsnaill
<i>Radix auricularia</i>	European Ear Snail
<i>Valvata piscinalis</i>	European Stream Valvata
<i>Viviparus georgianus</i>	Banded Mysterysnail

Plants	
<i>Agrostis gigantea</i>	Redtop, Black Bent, Water Bentgrass
<i>Alnus glutinosa</i>	Black Alder
<i>Alopecurus geniculatus</i>	Water Foxtail, Marsh Meadow-Foxtail
<i>Butomus umbellatus</i>	Flowering Rush
<i>Cabomba caroliniana</i>	Carolina Fanwort
<i>Carex acutiformis</i>	Swamp Sedge
<i>Carex disticha</i>	Tworank Sedge
<i>Chenopodium glaucum</i>	Oak-leaved Goosefoot
<i>Cirsium palustre</i>	Marsh Thistle
<i>Conium maculatum</i>	Poison Hemlock
<i>Echinochloa crus-galli</i>	Barnyard Grass
<i>Epilobium hirsutum</i>	Great Hairy Willow Herb
<i>Frangula alnus</i>	Glossy Buckthorn
<i>Glyceria maxima</i>	Reed Mannagrass
<i>Hydrocharis morsus-ranae</i>	European Frogbit
<i>Impatiens glandulifera</i>	Ornamental Jewelweed
<i>Iris pseudacorus</i>	Yellow Iris
<i>Juncus compressus</i>	Flattened Rush
<i>Juncus gerardii</i>	Black-grass Rush
<i>Juncus inflexus</i>	European Meadow Rush
<i>Lupinus polyphyllus</i>	Lupine
<i>Lycopus asper</i>	Western Water Horehound
<i>Lycopus europaeus</i>	European Water Horehound
<i>Lysimachia nummularia</i>	Moneywort
<i>Lysimachia vulgaris</i>	Yellow Loosestrife
<i>Lythrum salicaria</i>	Purple Loosestrife
<i>Marsilea quadrifolia</i>	European Water Clover
<i>Mentha aquatica</i>	Watermint
<i>Mentha gracilis</i>	Creeping Whorled Mint, Gingermint
<i>Mentha spicata</i>	Spearmint
<i>Myosotis scorpiodes</i>	True Forget-Me-Not
<i>Myosoton aquaticum</i>	Giant Chickweed
<i>Myriophyllum spicatum</i>	Eurasian Watermilfoil
<i>Najas marina</i>	Spiny Naiad
<i>Najas minor</i>	Brittle Waternymph
<i>Nasturtium officinale</i>	Water-cress
<i>Nymphoides peltata</i>	Yellow Floating Heart
<i>Pluchea odorata succulenta</i>	Sweetscent
<i>Pluchea odorata odorata</i>	Marsh Fleabane
<i>Poa trivialis</i>	Rough-stalked Meadow Grass
<i>Polygonum persicaria</i>	Lady's Thumb, Smartweed, Spotted Knotweed
<i>Potamogeton crispus</i>	Curlyleaf Pondweed
<i>Puccinellia distans</i>	Weeping Alkali Grass
<i>Rorippa sylvestris</i>	Creeping Yellow Cress

<i>Rumex longifolius</i>	Yard Dock
<i>Rumex obtusifolius</i>	Bitter Dock
<i>Salix alba</i>	White Willow
<i>Salix fragilis</i>	Crack Willow
<i>Salix purpurea</i>	Purple Willow
<i>Solanum dulcamara</i>	Bittersweet Nightshade
<i>Solidago sempervirens</i>	Seaside Goldenrod
<i>Sparganium glomeratum</i>	Bur Reed
<i>Trapa natans</i>	Water Chestnut
<i>Typha angustifolia</i>	Narrow-Leaved Cattail
<i>Veronica beccabunga</i>	European Brooklime
Platyhelminthes	
<i>Bothriocephalus acheilognathi</i>	Asian Tapeworm
<i>Dactylogyrus amphibothrium</i>	Monogenetic Fluke
<i>Dactylogyrus hemiamphibothrium</i>	Monogenetic Fluke
<i>Dugesia polychroa</i>	Flatworm
<i>Ichthyocotylurus pileatus</i>	Digenean Fluke
<i>Neascus brevicaudatus</i>	Digenean Fluke, Trematode
<i>Scolex pleuronectis</i>	Cestode
<i>Timoniella</i> sp.	Digenean Fluke, Trematode
Protozoans	
<i>Acineta nitocrae</i>	Suctorian Ciliate
<i>Glugea hertwigi</i>	Microsporidean Parasite
<i>Heterosporis</i> sp.	Microsporidean Parasite
<i>Myxobolus cerebralis</i>	Salmonid Whirling Disease
<i>Psammonobiotus communis</i>	Testate Amoeba
<i>Psammonobiotus dziwnowi</i>	Testate Amoeba
<i>Psammonobiotus linearis</i>	Testate Amoeba
<i>Sphaeromyxa sevastopoli</i>	Myxosporean Parasite
<i>Trypanosoma acerinae</i>	Flagellate Parasite
Viruses	
<i>Novirhabdovirus</i> p. genotype IV sublineage b	Viral Hemorrhagic Septicemia Virus (VHS)
<i>Ranavirus</i>	Largemouth Bass Virus (LMBV)
<i>Rhabdovirus carpio</i>	Spring Viremia of Carp (SVC)

3. Nonindigenous Species Management

There are two fundamentally different regulatory approaches to address nonindigenous species. One approach is to address a particular listed or known species, and another approach targets certain pathways by which a variety of species may be introduced (Corn and Johnson 2013). Both regulatory approaches are employed in the Great Lakes region at both the federal and state levels.

3.1 Regulations Targeting Pathways

A variety of state and federal regulations target the intentional and unintentional movement of species via various pathways without mentioning any particular species by name.

Ballast water regulations are aimed primarily at interdicting species which may be transported and introduced via the shipping ballast pathway. Presently, any ballast-related water in essentially all vessels over 79 feet in length that enter the Great Lakes through the St. Lawrence Seaway must have a salinity over 30 ppt, obtained either by mid-ocean exchange or exchange conducted at approved alternate locations. Any ballast tanks containing water not meeting that criteria are sealed and checked again as the ship leaves the Great Lakes. The United States Coast Guard (USCG) and the United States Environmental Protection Agency (USEPA) are implementing regulations under both the National Invasive Species Act (1996) and the Clean Water Act that will require treatment of ballast water prior to discharge to any United States waters. Discharged ballast water will have to meet numeric discharge standards for live organism content (DHS 2012). Treatment systems must be approved by the USCG for use in United States waters and several treatment systems have been approved on a short-term (five year) basis under the USCG "Alternative Management System" Program.

3.2 Federal Regulations Targeting Specific Species

A handful of aquatic invasive species established in the Great Lakes are called out by name for stringent regulation under federal laws. These species include Zebra and Quagga Mussels (*Dreissena* spp.), Sea Lamprey (*Petromyzon marinus*), noxious weeds (*Sparganium* spp.), and viruses (Viral Hemorrhagic Septicemia (VHS)).

The injurious wildlife provision of the Lacey Act ([1900](#)) bans import and shipment of listed species between the continental United States, the District of Columbia, Hawaii, the Commonwealth of Puerto Rico, or any possession of the United States. A 1990 amendment to the Lacey Act prohibits the possession and transportation of Zebra Mussel (*Dreissena polymorpha*) and Quagga Mussels (*D. rostriformis bugensis*) in the United States unless intended for scientific purposes (by permission of the Secretary of the Interior).

The Great Lakes Fishery Commission was formed in 1955 in part to control Sea Lamprey. The commission, in cooperation with Fisheries and Oceans Canada, the United States Fish and Wildlife Service, and the United States Army Corps of Engineers participates in Sea Lamprey control on the Great Lakes. Fisheries management on the Great Lakes, including management of nonindigenous fishes (as well as any factors affecting fish stocks of common concern), is coordinated through the Great Lakes Fishery Commission under the [Convention on Great Lakes Fisheries of 1954](#), though the states have ultimate authority for management within their borders.

The Federal Noxious Weed Act ([1974](#)) authorizes management of federally listed noxious weeds. Under the synonym, *Sparganium erectum*, *S. glomeratum* is federally listed as a noxious weed ([Code of Federal Regulations, Title 7, 360.200](#)).

The Animal Damage Control Act ([1931](#)) authorizes the United States Department of Agriculture Animal and Plant Health Inspection Service (APHIS) to control damage caused by wildlife to agricultural interests (including aquaculture), and the Federal Seed Act ([1939](#)) adds regulation of noxious weed seeds. Viral Hemorrhagic Septicemia (VHS) is an internationally-reportable fish disease. Permits from APHIS and health inspections were required for interstate movement of VHS-susceptible fish from VHS-infected or at risk states to non-infected states during 2006-2014. Post-2014, the federal order was removed as duplicative of state regulations. A United States Department of Agriculture import permit and a veterinary health certificate must accompany imports of live fish, fertilized eggs, and gametes of species susceptible to Spring Viremia of Carp including Common Carp (*Cyprinus carpio*), Koi (*C. carpio koi*), Grass Carp (*Ctenopharyngodon idella*), Silver Carp (*Hypophthalmichthys molitrix*), Bighead Carp (*H. nobilis*), Crucian Carp (*Carassius carassius*), Goldfish (*Carassius auratus*), Tench (*Tinca tinca*), Ide (*Leuciscus idus*), and Wels Catfish (*Silurus glanis*) (USDA APHIS 2012).

3.3 State Regulations Targeting Specific Species

A much broader array of established nonindigenous aquatic species are targeted for regulation in state laws in the Great Lakes region. Most states in the Great Lakes take a blacklist approach to regulation of aquatic invasive species in which species are regulated only if named in specific legislation/regulation. Illinois takes a whitelist approach to regulation of aquatic invasive species in which all aquatic species – including fish, reptiles, amphibians, mollusks, crustaceans, algae, aquatic plants, and aquatic invertebrates – are regulated unless approved. The whitelist approach results in the effective prohibition on aquaculture, transport, stocking, import and possession of a large number of species that are not specifically named in legislation. Illinois additionally blacklists some species as ‘injurious’ with more stringent regulation and penalties.

3.3.1 Fish pathogens

Five species of fish pathogens, including two bacteria (*Renibacterium salmoninarum* responsible for Bacterial Kidney Disease and *Aeromonas salmonicida* responsible for furunculosis), a myxosporean (*Myxobolus cerebralis* responsible for Whirling Disease), and two viruses (*Novirhabdovirus* sp. responsible for Viral Hemorrhagic Septicemia (VHS) and *Ranavirus* sp. responsible for Large Mouth Bass Virus) are explicitly targeted for regulation by the Great Lakes states. Four of these species (all but *A. salmonicida*) are considered to have high environmental impact to the Great Lakes. *A. salmonicida* is targeted primarily because it is considered a threat to hatchery operations for rearing salmonids. Regulations focus on transportation, stocking, and baitfish as the primary vectors for the movement of fish pathogens.

Table 1. Great Lakes States that regulate fish pathogens.

	MN	WI	IL	IN	MI	OH	PA	NY
<i>Aeromonas salmonicida</i>	√	√	√	√	√	√	√	√
<i>Renibacterium salmoninarum</i>	√	√	√	√	√	√	√	√
<i>Myxobolus cerebralis</i>	√	√	√	√	√	√	√	√
<i>Novirhabdovirus</i> sp. genotype IV sublineage b	√	√	√	√	√	√	√	√
<i>Ranavirus</i> sp.		√	√		√	√		

Regulations that target *Novirhabdovirus* sp. (VHS) focus on transport. Transportation of VHS-susceptible species is prohibited from New York, Pennsylvania, Ohio, Michigan, Indiana, Illinois (including movement of natural water from infested waters), Wisconsin, Minnesota, Ontario, and Quebec unless certain conditions are met (see below; USDA APHIS 2008). Movement of VHS-susceptible species is permitted from the two infected Canadian provinces to the United States if the shipment meets certain requirements and is imported under an APHIS permit for direct slaughter, or during catch and release fishing activities (USDA APHIS 2008). Movement of VHS-susceptible species is permitted between VHS-infected or at-risk states as long as fish are sent directly to state-inspected slaughter facilities that discharge waste water to a municipal sewage system that includes disinfection, or discharge to a non-discharging pond or a settling pond that disinfects according to all applicable United States Environmental Protection Agency (USEPA) and state regulatory criteria, and are accompanied by a valid VS 1-27 (Permit for Movement of Restricted Animals) form issued by an APHIS area office. Remains from slaughter facilities must be rendered or composted (USDA APHIS 2008). Interstate movement of VHS-susceptible fish is permitted from VHS-infected or at risk states to non-infected states as long as the fish are accompanied by appropriate state, tribal, or federal documentation stating the fish have tested negative for the virus (USDA APHIS 2008). Movement of VHS-susceptible species may also be permitted to state, federal, or tribal-authorized research and diagnostic facilities which meet containment and discharge requirements provided that the fish are accompanied by a valid VS 1-27 form issued by an APHIS area office and the remains are disposed of as medical waste adhering to all applicable USEPA and state regulatory criteria (USDA APHIS 2008).

Regulations targeting *Renibacterium salmoninarum*, *Myxobolus cerebralis*, *Aeromonas salmonicida*, and *Ranavirus* sp. focus primarily on stocking. The Great Lakes Fish Disease Control Policy and Model Program has prohibited stocking the Great Lakes and their tributaries with fish from whirling disease-infected farms. Fish imported into north-central-region states must be certified free of whirling disease in order to obtain import permits (Faisal and Garling 2004). Ohio requires out-of-state source facilities to document annual salmonid fish, egg, and

sperm health inspections for one year prior to importation (NCRAC 2010ab). Ohio further requires source facilities outside the Great Lakes basin to document annual health inspections showing no furunculosis, Bacterial Kidney Disease (BKD), or whirling disease occurrences for the previous five years prior to importing salmonids to the Lake Erie watershed (Baird 2005). Indiana requires source facilities within the Great Lakes basin to document they have been free of whirling disease for three consecutive years prior to importing salmonid stock. Source facilities outside the basin must document that salmonid stocks have been free of whirling disease, BKD, and furunculosis consecutively since 2002 (Baird 2005). Salmonids found carrying the pathogen, but asymptomatic, can be sold within states if source facilities are within the Great Lakes basin (Baird 2005). Michigan requires imported aquacultured fish to be accompanied by either an official interstate health certificate, official interstate certificate of veterinary inspection, or a fish disease inspection report. Importing of aquacultured fish is prohibited from source facilities with a record of an emergency disease within the past two years. Fish must be certified free of Largemouth Bass Virus (LMBV) if they are imported from non-Michigan source facilities and intended for stocking in public waters (NCRAC 2010ab). Michigan requires source facilities to document salmonid stocks have been whirling disease free for two consecutive years prior to importation, while Wisconsin requires one. Wisconsin requires source facilities to document fish health inspections targeting LMBV prior to importing live fish and eggs (NCRAC 2010ab). Both Michigan and Wisconsin also target BKD. Illinois requires source facilities of any species of live fish, eggs, and sperm to document they are disease free prior to importation (NCRAC 2010ab) and the testing of these facilities must be to the World Organization for Animal Health (OIE) or the American Fisheries Society (AFS) Bluebook standards and have appropriate HACCP in place. Further, Illinois restricts the import of live fish and gametes from certain geographic locations. Illinois and Minnesota also require imported salmonid health inspections. Minnesota allows the importation of whirling disease-infected eggs, if prior egg treatments are approved (Baird 2005). Prior to placing fish in New York waters, a fish health certification report must document that the fish are furunculosis, BKD, and VHS free.

All eight Great Lakes states have instated similar baitfish regulations to control the spread of *Myxobolus cerebralis* and *Novirhabdovirus* sp. New York, Pennsylvania, Michigan, Illinois, Wisconsin, and Minnesota have instated similar baitfish regulations to control the spread of furunculosis, BKD, and other fish pathogens. New York regulations include that bait harvested from inland waters for personal use is only permitted to be used within the same body of water from which it was taken and cannot be transported overland (with the exception of *Osmerus mordax*, suckers (Catastomidae), *Alosa pseudoharengus*, and *Alosa aestivalis*). Once transported, baitfish cannot be replaced to their original body of water (NYSDEC 2012a). Live or frozen bait harvested from inland New York waters for commercial purposes is only permitted to be sold or possessed on the same body of water from which it was taken and cannot be transported over land unless under a permit and/or accompanied by a fish health certification report. Bait that is preserved and packaged by any method other than freezing, such as salting, can be sold and used wherever the use of bait fish is legal as long as the package is labeled with the name of the

packager-processor, the name of the fish species, the quantity of fish packaged, and the means of preservation (NYSDEC 2012a). Certified bait may be sold for retail and transported overland as long as the consumer maintains a copy of a sales receipt that contains the name of the selling vendor, date sold, species of fish sold, and quantity of fish sold. Bait that has not been certified may still be sold, but the consumer must maintain a sales receipt containing the body of water where the bait fish was collected and a warning that the bait cannot be transported by motor vehicle. Bait sold for resale requires a fish health certification along with a receipt that contains the name of the selling vendor, date sold, species of fish sold, and quantity of fish sold, which must be kept for 30 days or until all bait is sold (NYSDEC 2012a).

3.3.2 Fish

Pacific salmon (*Oncorhynchus gorbuscha*, *O. kisutch*, *O. mykiss*, *O. nerka*, *O. tshawytscha*) were introduced to the Great Lakes in the 1960s to manage Alewife (*Alosa pseudoharengus*) populations and support a fishery (Tody and Tanner 1966). Soon after, the multi-million dollar Great Lakes Pacific salmon sportfishery was established and is now supports one of the largest economic sectors in the region. Therefore, Pacific salmon management objectives are designed to maintain or enhance the health and stability of the fisheries. Pacific salmon management is extremely diverse, integrated, and cascading and is therefore these are the most heavily regulated species (direct and indirectly) in the Great Lakes. Typically, Pacific salmon regulations are not species specific, but rather regulate the salmonid fisheries as a whole. *Salmo trutta*, *Osmerus mordax*, *Morone americana*, *Lepomis microlophus*, and *Cyprinus carpio* are other nonnative species which are managed at least in part to support fisheries (moderate to high beneficial species). *Alosa pseudoharengus* is the prime prey of Pacific salmonids and is managed in part to support the Pacific salmonid fisheries.

Table 2. Nonindigenous fishes managed primarily as beneficial species in Great Lakes States.

	MN	WI	IL	IN	MI	OH	PA	NY
<i>Lepomis microlophus</i>		√			√			
<i>Notropis burchanani</i>							√	
<i>Oncorhynchus gorbuscha</i>	√	√	√	√	√	√	√	√
<i>Oncorhynchus kisutch</i>	√	√	√	√	√	√	√	√
<i>Oncorhynchus mykiss</i>		√			√	√		√
<i>Oncorhynchus tshawytscha</i>	√	√	√	√	√	√	√	√
<i>Salmo trutta</i>		√						

Great Lakes states and provinces have their own specific fishing regulations. Generally, the overall goals and objectives of Pacific salmon (and other) fishing regulations are the same throughout the region. Fishing regulations include daily and season bag limits, size limits, permitted baitfish, manner of take (i.e., snagging or hook and line), and designated season dates

(See New York State DEC, Pennsylvania F&BC, Ohio DNR, Michigan DNR, Indiana DNR, Illinois DNR, Minnesota DNR, Wisconsin DNR websites for specific fishing regulations). Note that Ontario takes a whitelist approach to baitfish – all unlisted species are prohibited to use as bait.

The Ghost Shiner (*Notropis buchanani*) is native south of the Great Lakes basin and is listed as an endangered species in Pennsylvania. Pennsylvania state law prohibits the catch, take, kill, possession, and import to or export from Pennsylvania, sale or offer of sale or purchase of any individual of an endangered species, alive or dead, or any part thereof, without a special permit, ([58 PA Code § 75.1](#)). In locations where it is invasive to the Great Lakes basin, the environmental impacts of *N. buchanani* are unknown and socioeconomic impacts are low. No other state in the Great Lakes region regulates *N. buchanani*.

In addition to the above regulations on fish transport, stocking, baitfish (which target fish pathogens), fishing regulations (targeted at sustaining fisheries), and endangered species protections, there are state regulations for 14 species of nonindigenous fish which focus on controlling or managing fish as invasives – plus one species that is effectively restricted by a whitelist approach. Four of these species, Alewife (*Alosa pseudoharengus*), Common Carp (*Cyprinus carpio*), Rainbow Smelt (*Osmerus mordax*), and White Perch (*Morone americana*) each have favorable benefits as well as negative environmental impacts. The remaining 10 species include a mix of species with high to moderate impacts as well as a few for which impact has not been adequately assessed.

Table 3. Nonindigenous fishes regulated primarily as invasives in Great Lakes states.

	MN	WI	IL	IN	MI	OH	PA	NY
<i>Alosa aestivalis</i>			Restricted			Prohibited exotic		Restricted
<i>Alosa pseudoharengus</i>	Regulated		Approved			Prohibited exotic		Restricted
<i>Apeltes quadracus</i>			Restricted					
<i>Carassius auratus</i>	Regulated	Restricted invasive	Approved				Prohibited bait	Restricted
<i>Cyprinus carpio</i>	Regulated	Restricted invasive	Approved	Regulated		Prohibited exotic	Prohibited bait	Restricted
<i>Enneacanthus gloriosus</i>			Restricted					
<i>Gambusia affinis</i>		Prohibited	Native					
<i>Gymnocephalus cernua</i>	Prohibited	Restricted invasive	Injurious	Regulate	Prohibited	Prohibited	Prohibited	
<i>Lepisosteus platostomus</i>			Native			Permit Required		
<i>Misgurnus anguillicaudatus</i>		Restricted Aquarium	Restricted		Prohibited			
<i>Morone americana</i>	Prohibited		Restricted	Regulated		Prohibited		
<i>Neogobius melanostomus</i>	Prohibited		Injurious	Regulated	Prohibited	Prohibited	Prohibited	Restricted
<i>Oncorhynchus nerka</i>			Restricted					
<i>Osmerus mordax</i>	Regulated	Restricted invasive	Injurious	Regulated sportfish		Commercial unrestricted	Regulated commercial	Restricted
<i>Petromyzon marinus</i>	Prohibited					Prohibited		Restricted
<i>Proterorhinus semilunaris</i>	Prohibited	Restricted invasive	Injurious	Regulated	Prohibited	Prohibited	Prohibited	
<i>Scardinius erythrophthalmus</i>	Prohibited	Prohibited	Injurious	Regulated	Prohibited	Prohibited	Prohibited	

Gambusia affinis and *Scardinius erythrophthalmus* are prohibited in Wisconsin, meaning that no person may transport, possess, transfer, or introduce the species without authorization ([WI](#)

[Administrative Code § NR 40.04](#)). *Gymnocephalus cernua*, *Proterorhinus semilunaris*, *Osmerus mordax*, *Cyprinus carpio* and *Carassius auratus* are restricted invasive species, and therefore cannot be transported, possessed, transferred, or introduced without a permit ([WI Administrative Code § NR 40.05](#)). *Oncorhynchus mykiss*, *Salmo trutta*, and *Lepomis microlophus* are listed as restricted species under the definition “nonnative fish in the aquaculture industry” ([WI Administrative Code § NR 40.02, 40.05](#)) and therefore cannot be transported, possessed, transferred, or introduced without a permit. *Misgurnus anguillicaudatus* is restricted as a nonnative viable fish species in the aquarium trade ([WI Administrative Code § NR 40.02, 40.05](#)).

In Minnesota, *Petromyzon marinus*, *Gymnocephalus cernua*, *Morone americana*, *Scardinius erythrophthalmus*, *Proterorhinus semilunaris*, and *Neogobius melanostomus* are prohibited invasive species, meaning it is unlawful (a misdemeanor) to possess, import, purchase, transport, or introduce an organism except under permit for control, research, or education ([MN Administrative Rules § 6216.0250](#)). *Alosa pseudoharengus*, *Cyprinus carpio*, *Osmerus mordax*, and *Carassius auratus* are regulated invasive species ([MN Administrative Rules § 6216.0260 Regulated](#)), making it illegal to introduce the species without a permit ([MN Administrative Rules § 6216.2060, MN Administrative Rules § 6216.0265](#)).

In Ohio, it is unlawful for any person to possess, import, or sell live *Petromyzon marinus*, *Neogobius melanostomus*, *Proterorhinus semilunaris*, *Gymnocephalus cernua*, or *Morone americana* ([Ohio Administrative Code §1501:31-19](#)). A class B aquaculture permit is required to engage in propagation, culture, or sale of Shortnose Gar (*Lepisosteus platostomus*), and two levels of escapement prevention are required if cultured in the Lake Erie drainage basin ([Ohio Administrative Code § 1501:31-39-01](#)). In Ohio it is illegal for any person to possess, import or sell exotic species of fish including *Alosa aestivalis*, *Alosa pseudoharengus*, and *Cyprinus carpio* and/or hybrids for introduction or to release into any body of water that is hydrologically connected to public waters, or waters of the state, without first having obtained permission ([Ohio Administrative Code §1501:31-19](#)). In Ohio, *Osmerus mordax* is defined as a commercial fish and an unrestricted species under [Ohio Administrative Code 1501 § 31-1-02](#). Commercial fish are permitted to be taken, possessed, bought, or sold unless otherwise restricted in Ohio code.

In Indiana, *Gymnocephalus cernua*, *Neogobius melanostomus*, *Proterorhinus semilunaris*, and *Morone americana* are classified as exotic fish ([312 IN Administrative Code § 9-6-7](#)), meaning except as otherwise provided, no individual can import, possess, propagate, buy, sell, barter, trade, transfer, loan, or release into public or private waters live fish, recently hatched juveniles, viable eggs, or genetic material. Indiana ([312 IN Administrative Code § 9-6-8](#)) prohibits the use of carp as bait. Indiana also has no bag limit for *Cyprinus carpio* and has legalized spearfishing, bowfishing and snaring for this species. In Indiana, *Osmerus mordax* sport fishing season on Lake Michigan is defined as March 1-May 30, with capture allowed only by the use of dip nets, seines, or nets ([312 IN Administrative Code § 9-7-2](#)); otherwise, there is no bag limit, possession limit, or size limit as defined under [312 IN Administrative Code § 9-7-14](#).

In Pennsylvania ([58 PA Code §71.6](#)), it is illegal to possess, import, or introduce *Neogobius melanostomus*, *Proterorhinus semilunaris*, *Scardinius erythrophthalmus* or *Gymnocephalus cernua*. Further, it is unlawful to sell, purchase, offer for sale, or barter for live Ruffe ([58 PA Code § 63.46](#)). Ruffe may not be transported from another state, province, or country into Pennsylvania, liberated into a Pennsylvania watershed, or transferred between Pennsylvania waters without written permission from the Pennsylvania Fish and Boat Commission under [58 PA Code § 73.1](#), Pennsylvania ([58 PA Code §63.44](#)) prohibits the use of carp and Goldfish (*Carassius auratus*) as bait. The use of commercial trap nets under license to capture Rainbow Smelt (*Osmerus mordax*) is regulated by [Pennsylvania Administrative Code § 69.33](#).

In Michigan, *Misgurnus anguillicaudatus*, *Gymnocephalus cernua*, *Scardinius erythrophthalmus*, *Proterorhinus semilunaris*, and *Neogobius melanostomus* are prohibited species ([MI NREPA 451 § 324.41301](#)). Both introduction and possession of these species is prohibited except for education, research, or identification purposes ([MI NREPA 451 § 324.41303](#)). A violation involving a prohibited species is a felony, and a knowing introduction with intent to harm is punishable with up to five years imprisonment and a \$2,000 to \$1,000,000 fine ([MI NREPA § 324.41309](#)). *Lepomis microlophus* and *Oncorhynchus mykiss* are listed as approved species for aquaculture production in Michigan ([Aquaculture Development Act; MCL § 286.875](#)).

Illinois takes a whitelist approach to invasive fish species. Any non-native species not explicitly listed as approved for aquaculture, transportation, stocking, importation, and/or possession is automatically considered to be restricted. *Alosa pseudoharengus*, *Carassius auratus*, and *Cyprinus carpio* are approved. While not native to Lake Michigan, *Gambusia affinis*, *Lepisosteus platostomus*, *Esox niger*, *Ictiobus cyprinellus*, and *Phenacobius mirabilis* are native elsewhere in Illinois and so are not restricted. Though not called out by name, *Alosa aestivalis*, *Misgurnus anguillicaudatus*, *Morone americana*, *Oncorhynchus nerka*, *Apeltes quadracus*, and *Enneacanthus gloriosus* are considered restricted. *Gymnocephalus cernua*, *Proterorhinus semilunaris*, *Neogobius melanostomus*, and *Scardinius erythrophthalmus* are listed as injurious species under [Illinois Administrative Code 17 § 805.20](#). It is unlawful to possess, propagate, buy, sell, barter, or offer to be bought, sold, bartered, transported, traded, transferred, or loaned an injurious species to any person or institution unless a permit is obtained from the Illinois DNR ([IL Administrative Code 17 § 805.30](#)). Rainbow Smelt (*Osmerus mordax*) is regulated as a sportfish; the season for *O. mordax* is defined as March 1-April 30 under [Illinois Administrative Code 17-1 § 810.10](#).

New York, under federal law, is required to follow the Interstate Fishery Management Plan for Shad and River Herring established by the Atlantic States Marine Fisheries Commission ([Enacted May 2009](#); ASMFC). The New York Department of Environmental Conservation is therefore forced to close any non-sustainable commercial and recreational fisheries by January 1, 2012 until it can prove New York fisheries are self-sustaining (NYSDEC 2012b). New York prohibits or restricts the use of several species of nonindigenous fish as bait including *Alosa aestivalis*, *Alosa pseudoharengus*, *Osmerus mordax*, *Carassius auratus* larvae, *Petromyzon*

marinus larvae, carp (presumed all species), and *Neogobius melanostomus* ([6 NY Code, Rules, and Regulations \(NYCRR\) § Part 10, Paragraph 10.1\(c\)\(3\)](#), [6 NYCRR § Part 19, NY Environmental Conservation Law § 11-1315, 6a](#)). Goldfish larvae taken in nets operated pursuant to baitfishing are to be destroyed immediately ([NY Environmental Conservation Law § 11-1315](#)).

3.3.3 Mollusks

Six species of mollusks are state-regulated and an additional 11 species are effectively regulated due to Illinois' whitelist approach. Of these, only Zebra Mussels are regulated in all eight Great Lakes states, though Quagga Mussels are federally regulated and thus are effectively regulated in all eight states. This list includes a mix of species with high impact (Dreissenid mussels), moderate impact (*Corbicula fluminea* and *Potamopyrgus antipodarum*), and unknown impact species (*Cipangopaludina* spp.).

Table 4. Great Lakes states regulating nonindigenous mollusks.

	MN	WI	IL	IN	MI	OH	PA	NY
<i>Cipangopaludina chinensis malleata</i>	√	√	√					
<i>Cipangopaludina japonica</i>	√		√					
<i>Corbicula fluminea</i>		√	√	√				
<i>Dreissena polymorpha</i>	√	√	√	√	√	√	√	√
<i>Dreissena rostriformis bugensis</i>	√	√	√	√	√		√	
<i>Elimia virginica</i>			√					
<i>Gillia altilis</i>			√					
<i>Lasmigona subviridis</i>			√					
<i>Pisidium amnicum</i>			√					
<i>Pisidium henslowanum</i>			√					
<i>Pisidium moitessierianum</i>			√					
<i>Pisidium supinum</i>			√					
<i>Potamopyrgus antipodarum</i>	√	√	√					
<i>Radix auricularia</i>			√					
<i>Sphaerium corneum</i>			√					
<i>Valvata piscinalis</i>			√					

Corbicula fluminea and *Potamopyrgus antipodarum* are prohibited species in Wisconsin ([WI Administrative Code § NR 40.04](#)). *Cipangopaludina chinensis* is a restricted species in Wisconsin ([WI Administrative Code § NR 40.05](#)). In addition to the federal restrictions, in

Wisconsin, it is unlawful to transport, transfer, or introduce Zebra Mussels (*Dreissena polymorpha*) ([WI Administrative Code § NR 40.05](#)).

Potamopyrgus antipodarum are listed as a prohibited species in Minnesota ([MN Administrative Rules § 6216.0250](#)). *Cipangopaludina* spp. are regulated invasive species in Minnesota ([MN Administrative Rules § 6216.0260](#)). In addition to federal regulations, in Minnesota it is unlawful to place or attempt to place into state water a watercraft, trailer, or plant harvesting equipment that has attached Zebra Mussels ([MN Statutes § 84D.10](#)). Persons leaving the state are required to drain boats and related equipment during transportation on a public road ([MN Statutes § 84D.10](#), [MN Administrative Rules 6216.0500](#), Kaminski Leduc 2011).

In Indiana, an individual may not import, possess or release Asiatic clams (including *Corbicula fluminea*) or dreissenid mussels (*Dreissena polymorpha* and *Dreissena rostriformis bugensis*) into public or private waters ([312 IN Administrative Code § 9-9-3](#)).

Illinois also applies the whitelist approach to non-native mollusks. Thus *Corbicula fluminea*, *Cipangopaludina* sp., *Potamopyrgus antipodarum*, *Sphaerium corneum*, and *Valvata piscinalis* are restricted, even though they are not explicitly mentioned in legislation. *Lasmigona subviridis*, *Pisidium amnicum*, *P. henslowanum*, *P. moitessierianum*, *P. supinum*, *Elimia virginica*, *Radix auricularia*, and *Gillia altilis* are also not explicitly mentioned in Illinois' legislation, but are not currently found in Illinois waters – under the whitelist approach import of these species to Illinois is prohibited as is possession in the state. *Viviparus georgianus* and *Bithynia tentaculata* are approved species in Illinois. Illinois lists Zebra Mussels as injurious species ([IL Administrative Code 17 § 805.20](#)).

In addition to federal regulations, the following states have additional restrictions for mollusks specific to *Dreissena* spp. In Pennsylvania, it is unlawful to possess, introduce, import, transport, sell, purchase, offer for sale, or barter Zebra Mussels ([58 PA Code § 63.46](#), [58 PA Code § 71.6](#), [58 PA Code § 73.1](#)). In Michigan, Zebra Mussels are a restricted species ([MI Compiled Laws § 324.41301](#)) and therefore cannot be possessed unless it is to identify, eradicate, or control the species ([MI Compiled Laws § 324.41303](#)). In Ohio, it is unlawful to possess, import, or sell Zebra Mussels ([Ohio Administrative Code § 1501:31-19-01](#)). In New York, it is unlawful to intentionally release Zebra Mussels into state waters ([NY Environmental Conservation Law § 11-0507](#)).

3.3.4 Waterfleas and Mysids

Three species of established nonindigenous waterfleas as well as one mysid are regulated by name by select Great Lakes states. These include two high impact raptorial waterfleas (*Bythotrephes longimanus* and *Cercopagis pengoi*) and two relatively recent invaders whose impact has not been assessed (*Daphnia lumholtzi* and *Hemimysis anomala*). An additional 13 species are covered solely by Illinois' whitelist approach.

In Wisconsin, *Bythotrephes longimanus*, *Cercopagis pengoi*, *Daphnia lumholtzi*, and *Hemimysis anomala* are prohibited invasive species. With certain exceptions, it is unlawful to transport, possess, transfer, or introduce a prohibited invasive species in Wisconsin ([WI Administrative Code § NR 40.04](#)). In Minnesota, *Bythotrephes longimanus* is a regulated invasive species ([MN Administrative Rules § 6216.0260](#)); it is legal to possess, sell, buy, and transport regulated invasive species, but no person may introduce a regulated invasive species without a permit ([MN Administrative Rules § 6216.0265 Subpart 1](#)).

Illinois' whitelist approach also applies to crustaceans, therefore *Bythotrephes longimanus*, *Cercopagis pengoi*, *Daphnia lumholtzi*, *Hemimysis anomala*, *Eurytemora affinis*, *Heteropsyllus* nr. *nunni*, *Nitokra hibernica*, *Schizopera borutzkyi*, *Bosmina maritima*, *Echinogammarus ischnus*, and *Gammarus tigrinus* which are not approved, are considered restricted. *Cyclops strenuous*, *Megacyclops viridis*, *Nitokra incerta*, *Skistodiaptomus pallidus*, and *Daphnia galeata galeata* are also not approved, but not currently found in Illinois waters – possession and importation of these species is prohibited.

Table 5. Great Lakes states regulating nonindigenous crustacean species.

	MN	WI	IL	IN	MI	OH	PA	NY
<i>Bosmina coregoni</i>			√					
<i>Bythotrephes longimanus</i>	√	√	√					
<i>Cercopagis pengoi</i>		√	√					
<i>Daphnia galeata galeata</i>			√					
<i>Daphnia lumholtzi</i>		√	√					
<i>Eubosmina maritima</i>			√					
<i>Cyclops strenuus</i>			√					
<i>Eurytemora affinis</i>			√					
<i>Heteropsyllus</i> nr. <i>Nunni</i>			√					
<i>Megacyclops viridis</i>			√					
<i>Nitokra hibernica</i>			√					
<i>Nitokra incerta</i>			√					
<i>Schizopera borutzkyi</i>			√					
<i>Skistodiaptomus pallidus</i>			√					
<i>Echinogammarus ischnus</i>			√					
<i>Gammarus tigrinus</i>			√					
<i>Hemimysis anomala</i>		√	√					

3.3.5 Plants

In addition to the federal regulations applicable to the noxious weed *Sparganium glomeratum*, the Great Lakes states regulate 26 species of nonindigenous aquatic plants which are established in the basin. Seventy three percent of the listed plants have documented moderate to high environmental and or socioeconomic impacts. An overlapping 35% of plants have documented benefits. No individual plant species is regulated in all eight Great Lakes states. Some species (e.g., Black Alder, *Alnus glutinosa*), which are restricted in one state, are recommended in another. In Pennsylvania, Sweetscent (*Pluchea odorata*) (native to the eastern United States) is listed as an endangered species; no other Great Lakes state regulates this species.

Table 6. Great Lakes states regulating nonindigenous plant species.

	MN	WI	IL	IN	MI	OH	PA	NY
<i>Alnus glutinosa</i>	√	√	Approved					
<i>Alopecurus geniculatus</i>			Restricted					
<i>Butomus umbellatus</i>	√	√	Injurious	√	√	√	√	√
<i>Cabomba caroliniana</i>	√	√	Restricted		√			√
<i>Carex acutiformis</i>			Restricted					
<i>Carex disticha</i>			Restricted					
<i>Chenopodium glaucum</i>			?					
<i>Cirsium palustre</i>	√	√	Restricted		√			√
<i>Conium maculatum</i>		√	Restricted			√		
<i>Echinochloa crus-galli</i>	√		?					
<i>Epilobium hirsutum</i>		√	?			√		√
<i>Frangula alnus</i>	√	√	Regulated Exotic Weed					√
<i>Glyceria maxima</i>		√	Restricted					√
<i>Hydrocharis morsus-ranae</i>	√	√	Injurious	√	√			√
<i>Iris pseudacorus</i>	√		Injurious	√	√	√		√
<i>Juncus compressus</i>			Restricted					
<i>Juncus gerardii</i>			Restricted					
<i>Juncus inflexus</i>			Restricted					
<i>Lycopus europaeus</i>			Restricted					
<i>Lysimachia nummularia</i>			?					√
<i>Lysimachia vulgaris</i>			?					√
<i>Lythrum salicaria</i>	√	√	Regulated Exotic Weed	√	√	√	√	
<i>Marsilea quadrifolia</i>			Restricted					
<i>Myosotis scorpiodes</i>		*	?					
<i>Myriophyllum spicatum</i>	√	√	Injurious	√	√			
<i>Najas marina</i>	√		Approved					
<i>Najas minor</i>	√	√	Injurious	√				√
<i>Nasturtium officinale</i>		√	Approved					
<i>Nymphoides peltata</i>		√	Injurious	√	√			√
<i>Pluchea odorata</i>			Restricted***				Native **	
<i>Potamogeton crispus</i>	√	√	Injurious	√	√			√
<i>Rorippa sylvestris</i>			Restricted					
<i>Solanum dulcamara</i>			?					√
<i>Solidago sempervirens</i>			Restricted					
<i>Sparganium glomeratum</i>	√		Injurious	√				
<i>Trapa natans</i>	√	√	Injurious	√	√			√

<i>Typha angustifolia</i>		√	Approved	√				
<i>Veronica beccabunga</i>			Restricted					

*Proposal pending in WI to list *Myosotis scorpiodes* as a restricted species.

** In PA, *Pluchea odorata* is listed as an endangered species.

****Pluchea odorata succulenta* is native to parts of Illinois and not regulated. *Pluchea odorata odorata* is not native and restricted.

? Plants which are not obligate aquatic species (e.g., facultative and wetland plants) may not be considered ‘aquatic’ and so may not be covered under the IL Whitelist approach.

Cirsium palustre, *Cabomba caroliniana*, *Epilobium hirsutum*, *Glyceria maxima*, *Hydrocharis morsus-ranae*, *Myriophyllum spicatum* (including hybrids and variants), *Najas minor*, *Nymphoides peltata*, and *Trapa natans* are prohibited species in Wisconsin. Wisconsin law exempts some species in some counties (generally where already very well established) from the full restrictions otherwise associated with prohibited species. *Lythrum salicaria* is designated both as a restricted species ([WI Administrative Code § NR 40.05](#)) and as an invasive aquatic plant ([WI Administrative Code § NR 109.07](#)). *Butomus umbellatus*, *Frangula alnus*, *Potamogeton crispus*, and *Typha angustifolia* are listed as restricted species, and may not be transported, transferred, or introduced into any ecosystem. The Robert W. Freckman Herbarium of the University of Wisconsin lists an eradication notice for Poison Hemlock (*Conium maculatum*). However, as of 2007 it was not illegal to sell *C. maculatum* in Wisconsin (Annen 2007). However, the possession, transportation, transfer, or introduction of *C. maculatum* is restricted in most Wisconsin counties (WIDNR 2010a). Even though it is not restricted or prohibited, the Wisconsin Department of Natural Resources acknowledges *Nasturtium officinale* as being highly invasive and recommends its eradication upon detection (Robert W. Freckman Herbarium 2012). *Alnus glutinosa* is listed as a “plant to avoid” in Wisconsin’s planting guide.

Butomus umbellatus, *Hydrocharis morsus-ranae*, *Lythrum salicaria* (including all cultivars) *Myriophyllum spicatum* (including all hybrids and variants), *Najas minor*, *Potamogeton crispus*, *Sparganium glomeratum*, and *Trapa natans* are listed as prohibited species in Minnesota ([MN Administrative Rules § 6216.0250](#)). *Frangula alnus* is listed as a restricted noxious weed, and the importation, sale, or transport this plant is illegal (MNDNR 2009). *Iris pseudacorus* is established and considered a “moderate threat” to local ecosystems in Minnesota; this has led it to be classified as a restricted species that cannot be planted/released without a permit (GLPANS 2008, Minnesota Invasive Species Advisory Council 2009). *Cabomba caroliniana* can be possessed, sold, bought, and transported, but it is illegal to release it into the environment (MN DNR 2013b). Even though it is not present in Minnesota, *Cirsium palustre* is characterized as a severe threat to native ecosystems based on its impact in other locations (Minnesota Invasive Species Advisory Council 2009). *Echinochloa crus-galli* is considered to pose a “minimal” threat to ecosystems: it doesn’t pose significant competition with native species, it may naturalize, it does not significantly alter ecosystems, and has little possibility of spread within or

to other sites (Minnesota Invasive Species Advisory Council 2009). *Najas marina* is listed as a “species of special concern”; meaning it is extremely uncommon and deserves careful monitoring of its status (MN DNR 2013b). *Alnus glutinosa* is a recommended tree for urban environments in Minnesota (Johnson and Himanga 2009).

Trapa natans is prohibited in New York (GLPANS 2008). The New York Invasive Species Council assessed *Cabomba caroliniana*, *Frangula alnus*, *Glyceria maxima*, *Hydrocharis morsus-ranae*, *Iris pseudacorus*, *Lysimachia nummularia*, *Lysimachia vulgaris*, *Nymphoides peltata*, and *Potamogeton crispus* as having a high risk of causing ecological harm and recommend that their use be prohibited (NYISC 2010). The New York Invasive Species Council ranks *Najas minor* a moderate ecological risk and recommends that the species be regulated (NYISC 2010). *Cirsium palustre* and *Epilobium hirsutum* are also considered to be a medium to high threat species in New York (Higman and Campbell 2009, NYISC 2010). *Butomus umbellatus* is on New York State’s Interim Invasive Species Plant List (NYSDEC 2011).

Butomus umbellatus, *Hydrocharis morsus-ranae*, *Iris pseudacorus*, *Myriophyllum spicatum*, *Najas minor*, *Nymphoides peltata*, *Potamogeton crispus*, *Sparganium glomeratum*, and *Trapa natans* are listed as injurious species in Illinois. *Lythrum salicaria* and *Frangula alnus* are listed as exotic weeds in Illinois ([IL Compiled Statutes 525; 10/3 and 10/4](#)) making it illegal to buy, sell, or distribute plants, its seeds, or any part without a permit. Illinois’ whitelist approach applies to aquatic plants, but the definition of aquatic plant used does not correspond completely to the definition used by GLANSIS. Nonindigenous submerged, floating and emergent aquatic plants including *Alopecurus geniculatus*, *Cabomba caroliniana*, *Carex acutiformis*, *Carex disticha*, *Cirsium palustre*, *Conium maculatum*, *Juncus compressus*, *Juncus gerardii*, *Juncus inflexus*, *Lycopus europaeus*, *Marsilea quadrifolia*, *Pluchea odorata odorata*, *Rorippa sylvestris*, and *Veronica beccabunga* are clearly restricted under the Illinois whitelist approach.

Facultative wetland species which can invade aquatic environments but are principally upland species may not be restricted – these include *Agrostis gigantea*, *Alnus glutinosa*, *Chenopodium glaucum*, *Echinochloa crus-galli*, *Epilobium hirsutum*, *Glyceria maxima*, *Impatiens glandulifera*, *Lupinus polyphyllus*, *Lysimachia nummularia*, *Lysimachia vulgaris*, *Mentha aquatica*, *Mentha gracilis*, *Mentha spicata*, *Myosotis scorpiodes*, *Myosoton aquaticum*, *Poa trivialis*, *Polygonum persicaria*, *Rumex longifolius*, *Rumex obtusifolius*, *Salix alba*, *Salix fragilis*, *Salix purpurea*, and *Solanum dulcamara*. The New Invaders Watch Program lists *Glyceria maxima* on its “watch list” for Illinois (Maurer 2009). *Pluchea odorata succulenta*, *Lycopus asper*, *Najas marina*, *Puccinellia distans*, *Solidago sempervirens*, and *Typha angustifolia* are native to parts of Illinois and so not restricted. *Najas marina*, *Typha angustifolia* and *Nasturtium officinale* are approved for purposes of aquaculture.

Butomus umbellatus, *Cabomba caroliniana*, *Hydrocharis morsus-ranae*, *Iris pseudacorus*, *Myriophyllum spicatum*, *Nymphoides peltata*, and *Trapa natans* are prohibited in Michigan (GLPANS 2008). *Potamogeton crispus* and *Lythrum salicaria* (with an exemption for sterile

cultivars) are restricted ([MI NREPA 451 § 324.41301](#)). *Cirsium palustre* is considered to be a medium to high threat species in Michigan (Higman and Campbell 2009).

In Ohio, *Conium maculatum* has been designated as a prohibited noxious weed (Ohio Division of Natural Areas and Preserves and Nature Conservancy 2000, USDA NRCS 2012). Planting or sale of *Lythrum salicaria* without a permit is ([Ohio Revised Code § 927.682](#)), though the director may exempt varieties ‘demonstrated not to be a threat to the environment’. *Butomus umbellatus*, *Epilobium hirsutum* and *Iris pseudacorus* are classified as “well established invasive plants” by the Ohio Department of Natural Resources.

Pennsylvania has designated all nonnative *Lythrum* species and their cultivars as noxious weeds ([7 PA Code § 110.1](#)). *Butomus umbellatus* is labeled as a medium-high threat to native ecosystem. In Pennsylvania, *Pluchea odorata*, which is native to the east coast, is listed as an endangered species (USDA NRCS 2012).

Planting, sale, or other distribution of *Lythrum salicaria* without a permit is also prohibited in Indiana ([312 IN Code § 14-24-12](#)). Indiana recommends that Black Alder be inter-planted to improve soil quality and “protect other valuable trees” (IDNR n.d.) but the state also includes Black Alder on a list of invasive exotics plants whose “use in landscaping and re-vegetation projects should be avoided or limited when possible” (Homoya 2010).

3.3.6 Miscellaneous taxa covered only under the whitelist approach – Algae and Aquatic Invertebrates

The Illinois whitelist approach explicitly includes algae, while not listing any algae species as approved. Therefore all 27 species of nonindigenous algae are restricted by Illinois – including *Actinocyclus normanii* fo *subsalsa*, *Bangia atropurpurea*, *Chaetoceros muelleri*, *Chroodactylon ornatum*, *Controcriba guillardii*, *Cyclotella atomus*, *Cyclotella cryptica*, *Cylindrospermopsis raciborskii*, *Diatoma ehrenbergii*, *Discostella pseudostelligera*, *Discostella wolterecki*, *Hymenomonas roseola*, *Nitellopsis obtusa*, *Pleurosira laevis*, *Skeletonema potamos*, *Skeletonema subsalsum*, *Sphacelaria fluviatilis*, *Sphacelaria lacustris*, *Stephanodiscus binderanus*, *Stephanodiscus subtilis*, *Thalassiosira baltica*, *Thalassiosira lacustris*, *Thalassiosira pseudonana*, *Thalassiosira weissflogii*, *Ulva flexuosa*, *Ulva intestinalis*, and *Ulva prolifera*.

Illinois also includes aquatic invertebrates in its whitelist approach. We interpret this to include restrictions on the free-living annelids (*Branchiura sowerbyi*, *Ganius aquaedulcis*, *Potamotheix bedoti*, *Potamotheix moldaviensis*, *Potamotheix vejovskyi*, and *Ripistes parasita*), Flatworms (*Dugesia polychroa*), insects (*Acentria ephemerella* and *Tanytarsus lemnae*), bryozoan (*Lophopodella carteri*), hydrozoa (*Cordylophora caspia* and *Craspedacusta sowerbyi*), and Amoebae (*Psammobiotus communis*, *Psammobiotus dziwnowii*, and *Psammobiotus linearis*).

3.4 Unregulated Species

Sixty four percent (116 of 181) of nonindigenous species established in the Great Lakes are not mentioned in any state or federal regulation. Seventy-eight of these species (67%) appear to be captured under Illinois' whitelist approach; highlighting the strength of this approach in regulating additional species. This leaves 38 species (20%) entirely unregulated (Table 7).

Free-living micro-plankton and small benthic invertebrates are disproportionally represented among the nonindigenous species not captured by name but potentially regulated under the whitelist approach. There are no federal or state regulations which explicitly target any of the 27 species of algae, eight free-living copepods, six annelids, three testate amoebae, two insects, two amphipods, one freshwater bryozoan, two freshwater hydroids, or one free-living flatworm which number among the Great Lakes nonindigenous species. Additionally, three of the five nonindigenous waterfleas (60%), 12 of 18 mollusks (66%), 28 of 56 plants (50%), and 16 of 21 fish parasites/diseases (80%) are not mentioned by name. In contrast, most established nonindigenous fishes are identified in federal and state regulations; only 6 of the 27 nonindigenous fishes (22%) are not regulated and half of those are captured by the whitelist approach leaving only 3 nonindigenous fish species entirely unregulated.

Table 7. Unregulated nonindigenous species.

Fishes <i>Esox niger</i> <i>Ictiobus cyprinellus</i> <i>Phenacobius mirabilis</i> Fish Parasites and Diseases <i>Argulus japonicus</i> <i>Neoergasilis japonicus</i> <i>Salmincola lotae</i> <i>Bothriocephalus acheilognathi</i> <i>Dactylogyrus amphibothrium</i> <i>Dactylogyrus hemiamphibothrium</i> <i>Ichthyocotylurus pileatus</i> <i>Neascus brevicaudatus</i> <i>Scolex pleuronectis</i> <i>Timoniella</i> sp. <i>Acineta nitocrae</i> <i>Glugea hertwigi</i> <i>Heterosporis</i> sp. <i>Sphaeromyxa sevastopoli</i> <i>Trypanosoma acerinae</i> <i>Piscirickettsia</i> cf. <i>salmonis</i>	Mollusks <i>Bithynia tentaculata</i> <i>Viviparus georgianus</i> Plants <i>Agrostis gigantea</i> <i>Impatiens glandulifera</i> <i>Lupinus polyphyllus</i> <i>Lycopus asper</i> <i>Mentha aquatica</i> <i>Mentha gracilis</i> <i>Mentha spicata</i> <i>Myosotis scorpiodes</i> <i>Myosoton aquaticum</i> <i>Poa trivialis</i> <i>Polygonum persicaria</i> <i>Puccinellia distans</i> <i>Rumex longifolius</i> <i>Rumex obtusifolius</i> <i>Salix alba</i> <i>Salix fragilis</i> <i>Salix purpurea</i>
--	---

Unregulated species are not necessarily innocuous. *Ichthyocotylurus pileatus*, *Heterosporis* sp., and *Bithynia tentaculata* were rated in our previous assessments ([GLERL TM-161](#)) as having high environmental impact while *Polygonum persicaria* has high socioeconomic impact, *Piscirickettsia* cf. *salmonis* has moderate environmental impact and *Salix fragilis* has moderate socioeconomic impact – 6 out of 35 (17%) of unregulated species are thus still responsible for significant impacts.

4. Nonindigenous Species Control

In this section we summarize the control technologies that are available for control of various nonindigenous taxa. Information is drawn from the scientific literature and case studies. Where a particular method has been tried and proven ineffective, this is noted. Absence of information on the effect of a particular method on a particular species indicates only that we were unable to find information, not that the method has proven ineffective.

4.1 Algae

Options for control of established nonindigenous algae in open waters are limited. Only the largest two species (*Ulva prolifera* and *Nitellopsis obtusa*) can be effectively harvested mechanically, even then, they are often the first to recolonize (Pullman and Crawford 2010). Some species, particularly *Cylindrospermopsis raciborskii*, are associated with stratified water columns and mechanical destratification (mixing) may help prevent bloom development (Antenucci et al. 2005).

Nitellopsis obtusa is very sensitive to common algaecides containing copper and endothall based compounds. When *N. obtusa* is still low growing, algaecide can treat the entire organism. However, in taller individuals, the algaecide is absorbed in the top of the plant, killing that portion but leaving the bottom of the plant alive. This type of treatment has been found to be somewhat successful and is called a “hair cut treatment” by managers. The timing of the algaecide treatment is also important. Treatment early in the spring could help open up spawning habitat for native fish species, but *N. obtusa* or other nonnative aquatic plants are likely to recolonize these areas in the early summer. Algicides are generally less effective on planktonic algae, except for limited applications in smaller enclosed ponds. Note, the use of copper-based algicides may inhibit the degradation of cylindrospermopsin (Smith and Alexander 2008) and so are not a good choice when dealing with *Cylindrospermopsis raciborskii*.

Many of the invasive algae proliferate only in high-nutrient (*A. normanii* fo. *subsalsa*, *Cyclotella atomus*, *Diatoma ehrenbergii*, *Discostella pseudostelligera*, *Stephanodiscus binderanus*, *Thalassiosira baltica*, and *Thalassiosira pseudonana*) or high-saline (*Cyclotella cryptica*, *Thalassiosira lacustris*, and *Thalassiosira weissflogii*) conditions or both (*Bangia atropurpurea*, *Chaetoceros muelleri* var. *subsalsum*, *Contricribra guillardii*, *Stephanodiscus subtilis*, *Ulva flexuosa*, *Ulva intestinalis*, and *Ulva prolifera*). Measures to reduce nutrient and salt pollution are usually the most effective options to control these species.

Management of *Cylindrospermopsis raciborskii* often focuses on alleviating impacts due to the toxins that it produces. Potential management tools include absorption by activated carbon at point of use (Westrick et al. 2010), bacterial degradation (Donovan et al. 2008, Ho et al. 2012), and inactivation by chlorine, ozone, and/or hydroxyl radical treatments (Westrick et al. 2010).

There are no known control methods for some planktonic algae, particularly those that are not associated with high-nutrient concentrations or salinity. Fortunately, these species (*Chroodactylon ornatum*, *Hymenomonas roseola*, *Pleurosira laevis*, *Skeletonema potamos*, *Skeletonema subsalsum*, *Sphacelaria fluviatilis*, and *Sphacelaria lacustris*) also tend to have low impact.

4.2 Plants

Although in some cases control options are extremely limited (e.g., to hand pulling) and large, established infestations may be difficult to control, control options are at least available for all the established nonindigenous plants. Many tools are available for control of aquatic plants, though rarely is any one method completely effective in eradicating a particular species. Several species of nonindigenous plants produce toxins that cause skin irritation, contact dermatitis, allergic reactions in sensitive individuals, or even chemical burns (Pitcher 2004, Cooper and Johnson 1984, King County 2010). Proper protective clothing should be used when handling or burning the following: *Conium maculatum*, *Iris pseudacorus*, *Polygonum persicaria*, and *Solanum dulcamara*.

While small, new infestations of most invasive plants can be controlled by hand-pulling before seed-set, most invasive plants have characteristics (rhizomes, tubers, extensive roots, viable seed bank, etc.) which make this unfeasible for control of truly established or extensive infestations. Great care to remove all parts of the plant should be taken when implementing a physical method of control. To ensure removed plants will not sent out shoots, thoroughly dry all plant pieces (Jensen 2011).

Invasive tree species (*Alnus glutinosa*, *Frangula alnus*, *Salix* spp.) can be felled, but usually require application of an herbicide in order to prevent regrowth (USDA NRCS 2006).

Table 8. Physical control methods for invasive trees.

	Handpulling	Felling/girdling + Herbicide	Mowing/cutting/h arvesting	Flooding	Burn
<i>Alnus glutinosa</i>		Effective			
<i>Frangula alnus</i>	Plants <0.5cm	Effective if fruit are also removed/burned	Plants <2 years old. Most effective in spring.	Effective	Repeat every 2-3 years
<i>Salix</i> spp.	Effective for small seedlings	May need to be repeated, for large trees need to have the roots removed with machinery	Effective for seedlings		

Mowing, cutting, or harvesting is frequently employed as a physical control for invasive plants. For most species, mowing should be conducted before seedset. Repeated mowing (often over several years) is recommended for species with extensive roots/rhizomes (*Butomus umbellatus*, *Carex* spp, *Glyceria maxima*, and *Iris pseudacorus*) to deplete those energy reserves (Jensen 2011). Species which reproduce easily by fragmentation (e.g., *Lysimachia* spp. and *Myriophyllum spicatum*) may be spread or promoted by attempts to harvest (Kennay and Fell 2011). In other species, increased mowing promotes germination of the seed bank (*Lythrum salicaria*), aids seedling development (*Rumex* spp.), stimulates growth (*Echinochloa crus-galli* and *Juncus* spp.), or increases competitive capacity of the invasive over natives (*Cirsium palustre* and *Potamogeton crispus*).

Plants whose seeds require light to germinate (*Echinochloa crus-galli* and *Lupinus polyphyllus*) may be controlled by seed burial via tilling or mulching (Cornell University 2012). Tilling may also be effective to kill seedlings (*Alopecurus geniculatus* and *Rumex* spp.).

Water level manipulation is frequently employed to control invasive plants (Toogood et al 2008, Lenseen et al 2000). Some species are sensitive to drying (*Alopecurus geniculatus*, *Cabomba caroliniana*, *Hydrocharis morsus-ranae*, *Myriophyllum spicatum*, *Poa trivialis*, and *Potamogeton crispus*). Other species are sensitive to flooding (*Epilobium hirsutum*, *Frangula alnus*, *Lythrum salicaria*, and *Typha angustifolia*). Note that the length of time necessary and the sensitive season may vary with each species.

Controlled burns (Hanson et al. 2012, DiThomaso 2013) may be effective for some species (*Frangula alnus*, *Lysimachia nummularia*, *Polygonum persicaria*, and *Typha angustifolia*), but promote germination/regrowth in others (*Iris pseudacorus* and *Lupinus polyphyllus*).

Though usually limited to controlling small areas such as around docks or at swimming beaches, use of plastic or geotextile barriers or shading of the sediments (Forest Health Staff 2006) has proven effective for several species (*Glyceria maxima*, *Myriophyllum spicatum*, *Najas minor*, *Polygonum persicaria*, *Potamogeton crispus*, and *Solanum dulcamara*).

Table 9. Physical control methods for invasive floating plants.

	Handpulling	Mowing/cutting/harvesting	Drying/drawdowns
<i>Hydrocharis morsus-ranae</i>		May provide temporary control	After turions have germinated but before extensive growth
<i>Nasturtium officinale</i>	Effective for small populations		
<i>Nymphoides peltata</i>	Effective if all plant pieces are removed	Even with multiple harvests per year, complete control is unlikely.	
<i>Trapa natans</i>	roots easily uplifted and removed in small populations - all fragments must be removed	necessray for larger populations, fragments will regrow	

Table 10. Physical control methods for invasive submerged plants.

	Handpulling	Mowing/cutting/harvesting	Seed burial/Tilling/mulch	Drying/drawdowns	Shading/Barriers
<i>Cabomba caroliniana</i>	avoid late season when the plant is brittle	likely to provide relief only for a few weeks		Will reduce growth but rootball must be dried completely or it will return	
<i>Myriophyllum spicatum</i>	Effective for small populations	Only after all niches filled - otherwise this will enhance the rate of spread. Multiple harvest per season		Most effective if plants exposed to several weeks drying and root crowns exposed to sub-freezing temperatures	Cover sediment with opaque fabric
<i>Najas minor</i>		Physical removal provides only short-term relief			Benthic barriers may be effective
<i>Potamogeton crispus</i>	Effective if crowns, turions and fragments are removed	Use only if monoculture	Use only if monoculture	Autumn drawdown will kill turions	Effective

Table 11. Physical control methods for invasive emergent monocot species.

	Handpulling	Mowing/cutting/harvesting	Seed burial/Tilling/mulch	Drying/drawdowns	Flooding	Burn	Shading/Barriers
<i>Agrostis gigantea</i>	Effective for small populations	<3 inches					
<i>Alopecurus geniculatus</i>			effective only if seed bank not extensive	Effective	Not effective		
<i>Butomus umbellatus</i>	rhizomes must be removed	multiple cuts per year will reduce abundance and prevent spread		Not effective			
<i>Carex</i> spp.		rhizomes must be removed	>7cm	Not effective	Not effective		
<i>Echinochloa crus-galli</i>		Not effective	<1 cm prevents germination				
<i>Glyceria maxima</i>	Effective if rhizomes can be removed	2-3 times per summer may deplete roots and rhizomes			May be used together with other methods		Cover with black plastic for 5-6 weeks
<i>Iris pseudacorus</i>	Rhizomes must be removed.	Remove leaves and stems above water level before flowing. May need to be repeated 3-4 years.				Not recommended as this encourages reprofiting from rhizomes	
<i>Juncus</i> spp.		This may increase		Not effective	Not effective		

		the seed bank as well as promote regrowth.					
<i>Poa trivialis</i>				This species has a low tolerance for drought.			
<i>Sparganium glomeratum</i>	roots are easily lifted from sediment, but must be removed	roots are easily lifted from sediment, but must be removed					
<i>Typha angustifolia</i>		during the growing season and again just before flowers reach maturity			Flooding can be effective if it triggers anaerobic respiration in the sediments, or following cutting	May be effective if repeated several times or rhizomes are exposed	

Table 12. Physical control methods for invasive emergent broadleaf plants.

	Handpulling	Mowing/cutting/harvesting	Seed burial/Tilling/mulch	Flooding	Burn	Shading/Barriers
<i>Chenopodium glaucum</i>			spring and fall			
<i>Cirsium palustre</i>	cut just below surface before flowering	Repeated (>3x per year) close mowing for 3-4 years necessary				
<i>Conium maculatum</i>	Taproot must be removed	Mow before flowering				
<i>Epilobium hirsutum</i>	Effective if rhizomes can be removed			>18 weeks can reduce roots and leave the plant more susceptible to other control methods		
<i>Impatiens glandulifera</i>	Before seed-set	Cut below the lowest node to prevent re-growth. Stems left must be crushed to prevent regrowth.				
<i>Lupinus polyphyllus</i>	Root system should be severed below the crown.	Generally not effective unless very frequent			Not effective, promotes germination	
<i>Lysimachia nummularia</i>	All fragments and rhizomes	May contain the existing population, but will not				

	must be removed to prevent resprouting	eradicate it.				
<i>Lythrum salicaria</i>	Remove entire root system before seedset using a cultivator	Only if followed by flooding, otherwise it promotes germination		> 30cm for over 7 weeks, or following mowing	Not effective	
<i>Mentha</i> spp.	Effective for small populations					Soil barriers (edging) may contain spread via rhizomes
<i>Myosoton aquaticum</i>		Before seeds form				
<i>Polygonum persicaria</i>	Effective				Effective to kill seeds	Solarizing black plastic will kill seeds
<i>Rorippa sylvestris</i>	Small rhizome fragments are difficult to remove					
<i>Rumex</i> spp.		To prevent seedset, but may aid seedling development and promote branching	Repeated will eliminate seedlings			
<i>Solanum dulcamara</i>	all parts of roots must be removed	Not effective unless followed by root removal				Cover with geotextile cloth for 2 years
<i>Solidago sempervirens</i>	Deadheading prior to seedset is effective	Deadheading prior to seedset is effective				

Many chemical herbicides are available for the control of invasive plants. However, only a handful of these are approved for use in open waters. Thus while the following notes herbicides and chemical families that have proven effective for particular species, options in open water are MUCH more limited than this suggests.

Glyphosate is an EPSP Synthase inhibitor (Herbicide Family 9) which leads to the depletion of the aromatic amino acids tryptophan, tyrosine, and phenylalanine (Shaner 2006). This systemic broad-spectrum herbicide is used to control floating-leaved plants like water lilies and shoreline plants like purple loosestrife. It is generally applied as a liquid to the leaves. Glyphosate does not work on underwater plants such as Eurasian watermilfoil. Glyphosate is non-selective and will injure any plant tissue with which it comes in contact. It is toxic to frogs and tadpoles. It is commonly used as a foliar spray and proven effective against *Alnus glutinosa* (stump application), *Carex* spp., *Cirsium palustre* (applied to cut stems), *Conium maculatum*, *Frangula alnus* (applied to basal stems or foliar), *Glyceria maxima*, Ornamental jewelweed (*Impatiens glandulifera*), *Iris pseudacorus*, *Juncus* spp., *Lysimachia* spp., *Lythrum salicaria*, *Mentha* spp., Giant chickweed (*Myosoton aquaticum*), dock (*Rumex*) spp. (at early heading), Willow (*Salix*) spp., and *Typha angustifolia*. It is effective against *Echinochloa crus-galli* if applied to plants <5 cm in height. Because of its non-selective nature, re-seeding (with natives capable of outcompeting seedlings) is required to prevent regrowth of species such as lupine (*Lupinus polyphyllus*) and Rough-stalked meadow grass (*Poa trivialis*). While not directly tested on invasive *Alopecurus geniculatus*, it has been proven effective against related species. It is not effective against *Nasturtium officinale* in flowing water. It will kill above-ground biomass of Great Hairy Willowherb (*Epilobium hirsutum*) but repeated applications are needed to weaken roots. Likewise it is marginally effective against Creeping Yellow Cress (*Rorippa sylvestris*) and *Nymphoides peltata*. Some applicators report control of *Butomus umbellatus* with glyphosate. Glyphosate-resistant strains of Redtop (*Agrostis gigantea*) and Lady's Thumb (*Polygonum persicaria*) are becoming common. Plants can take several weeks to die and a repeat application is often necessary to remove plants that were missed during the first application.

ACCase (acetyl-Coenzyme A carboxylase) inhibitors (Herbicide Family 1) block the first step in fatty acid synthesis in grasses (Armstrong 2012). With a few specific exceptions, herbicides in this family are not approved for use in aquatic environments, but can be used to control these invasives in upland settings. Most of the herbicides in this family are most effective against seedlings when applied early post-emergence. Broadleaf plants have a natural resistance to herbicides in this family. Some herbicides in this family control only specific subsets of grasses. Cyhalofop-butyl is highly toxic to both freshwater and estuarine/marine animals on an acute exposure basis. Data indicate that the major degradation products for cyhalofop-butyl are non-toxic to most aquatic organisms at normal application levels; however for endangered species of estuarine/marine fish, estuarine/marine invertebrates, and freshwater fish this low toxicity is still considered a concern and use prohibited where such species are of concern. It is prohibited for

aquatic use except in rice fields. However, most varieties of rice are now tolerant of cyhalofop-butyl. Clethodim is slightly toxic to fish and aquatic invertebrate species. Clethodim may be highly persistent in the aquatic environment. It can be used to control invasive plants in upland areas, but not for direct aquatic application. Sethoxydim will not harm broadleaf herbs, sedges or woody plants. It is only mildly toxic to aquatic animals, but degrades rapidly (<1 hour) in open water. Dichlofop, fluazifop and haloxyfop are very toxic to aquatic organisms, may cause long-term adverse effects in the aquatic environment. Fluazifop-p cannot be used in Forest Sustainability Certified Areas. Fluazifop controls Water Foxtail (*Alopecurus geniculatus*).

Table 13. Plants susceptible to ACCase inhibitors.

	Cyhalofop	Clethodium	Dichlofop	Fluazifop	Haloxyfop	Sethoxydim
<i>Agrostis gigantea</i>				Proven for related species		
<i>Alopecurus geniculatus</i>				Effective		Effective
<i>Echinochloa crus-galli</i>	Effective	Effective	Effective	Effective	Effective	Effective
<i>Frangula alnus</i>				Effective		
<i>Juncus</i> spp.						Effective

ALS (acetolactate synthase) inhibitors (Herbicide Family 2) block the production of branched chain amino acids (isoleucine, leucine and valine). Herbicide resistance is common in this family (Armstrong 2012). Corn is often susceptible to herbicides in this family thus many are not recommended for use near croplands. Rimsulfuron and DPX-79406 (1:1 premix of nicosulfuron and rimsulfuron) are registered in Ontario and can be used with corn. Bispyribac is registered for use in ricefields. Rimsulfuron is most effective in dry conditions. Chlorsulfuron should not be used on powdery, dry, light or sandy soils. Imazapyr is a systemic, broad-spectrum, slow-acting herbicide. When applied as a liquid it is used to control emergent plants like spartina, reed canarygrass, phragmites, and floating-leaved plants like water lilies but does not work on underwater plants such as Eurasian Watermilfoil. Imazapyr is effective on some sedge species and most effective when water levels are low and during calm weather. Imazapic is similar, but non-selective.

Table 14. Plants susceptible to ALS inhibitors.

	bensulfuron methyl	Bispyribac	chlorsulfuron	Imazapyr	Imazamox/ imazapic/ imazaquin	metalsulfuron	penoxsulam	triflorsulfuron	Sulfometuron	sulfosulfuron
<i>Agrostis gigantea</i>					proven effective on related species			effective if kept dry		
<i>Butomus umbellatus</i>				effective during calm weather						
<i>Carex</i> spp.				proven effective on some sedge species						
<i>Cirsium palustre</i>						effective foliar				
<i>Conium maculatum</i>			effective		effective	effective				
<i>Echinochloa crus-galli</i>		effective postemergent		effective	effective			effective	effective	
<i>Frangula alnus</i>				effective foliar		effective foliar				
<i>Glyceria maxima</i>				effective only when water levels are low						
<i>Hydrocharis morsus-ranae</i>				effective	effective		effective			
<i>Impatiens glandulifera</i>						effective				
<i>Iris pseudacorus</i>				excellent for large infestations						
<i>Juncus</i> spp.					effective					
<i>Lupinus polyphyllus</i>			effective			effective				
<i>Lysimachia vulgaris</i>				effective						

<i>Lythrum salicaria</i>				effective						
<i>Marsilea quadrifolia</i>	used in Japan									
<i>Poa trivialis</i>										currently being labelled for selective control in creeping bentgrass
<i>Rumex</i> spp.			effective on young plants						effective	
<i>Salix</i> spp.						effective on fully leaved-out plants				
<i>Typha angustifolia</i>				effective						

Microtubule protein inhibitors (Herbicide Family 3) interfere with the alignment of the spindle apparatus during mitosis and prevent normal cell division in root tissue; thus they inhibit root growth (Armstrong 2012). Pendamethalin is used for post-emergent control of *Echinochloa crus-galli* and trifluralin for control of *Rorippa sylvestris*. Pendamethalin cannot be used in Forest Sustainability Certified Areas or open waters.

Synthetic auxins (Herbicide Family 4) mimic the natural plant hormone IAA (indole-3-acetic acid). These herbicides affect cell wall plasticity and nucleic acid metabolism which leads to inhibited cell division and growth in the meristem (Armstrong 2012). These herbicides are generally selective for broadleaf control. 2,4-D is approved in both granular and liquid forms; by court-order the butoxy-ethyl-ester formulation of 2,4-D cannot be used in waters with threatened and endangered salmon-bearing waters in the Pacific Northwest. Picloram and Triclopyr have been proven effective in control of *Conium maculatum*, *Frangula alnus*, and *Rumex* spp. Triclopyr also is effective for control of *Alnus glutinosa*, *Impatiens glandulifera*, *Lysimachia vulgaris*, *Lythrum salicaria*, *Myriophyllum spicatum*, *Salix* spp., *Solanum dulcamara*, and *Trapa natans*. There are two formulations of triclopyr. Triclopyr should be used in its amine form in wetlands. The TEA formation of triclopyr is registered for use in aquatic or riparian environments. Many native aquatic species are unaffected by triclopyr; it is very useful for Purple Loosestrife control since native grasses and sedges are unaffected by this herbicide.

Photosystem I inhibitors (Herbicide Family 22) capture electrons from photosystem I and are reduced to form free radicals that destroy cell membranes (Armstrong 2012). Diquat is a fast-acting, non-selective contact herbicide which destroys the vegetative part of the plant but does not kill the roots. It is applied as a liquid. Typically diquat is used primarily for short term (one season) control of a variety of submersed aquatic plants. It is very fast-acting and is suitable for spot treatment. However, turbid water or dense algal blooms can interfere with its effectiveness. Diquat provides effective control of *Hydrocharis morsus-ranae*, *Myriophyllum spicatum*, *Najas minor*, and *Typha angustifolia*. It can be used to control *Potamogeton crispus* if applied before turion production and has been proven in laboratory tests to be effective against *Cabomba caroliniana* (Michigan applicators report it to be ineffective against *Cabomba* in the field). Some applicators report control of submerged *Butomus umbellatus* with diquat in sites that have limited dilution potential. Paraquat is effective against *Echinochloa crus-galli* when applied to plants less than five centimeters in height.

Photosystem II Inhibitors (Herbicide Family 5-7) inhibit photosynthesis by binding to the QB-binding niche on the D1 protein of the photosystem II complex in the chloroplast (Armstrong 2012). It blocks electron flow from QA to QB and stops carbon dioxide fixation and production of ATP and NADPH₂ which are needed for plant growth and development. Death occurs from free radicals destroying cell membranes. Herbicide resistance is increasingly common in this group. Herbicides in this class have proven effective in control of *Agrostis gigantea*, *Conium maculatum*, *Myriophyllum spicatum*, and *Potamogeton crispus*. This family of herbicides is most

effective against *Echinochloa crus-galli* when used post-emergence on plants <5 cm. Hexazinone and simazine cannot be used in Forest Sustainability Certified Areas.

Table 15. Nonindigenous plants susceptible to Photosystem II inhibitors.

	Atrazine	Hexazinone	Linuron	Metribuzin	Simazine	Terbacil	Terbutryn	Propanil	Buthidazole
<i>Agrostis gigantea</i>	Effective								
<i>Conium maculatum</i>		Effective		Effective		Effective			
<i>Echinochloa crus-galli</i>	Effective	Effective	Effective		Effective		Effective	Effective	Effective
<i>Myriophyllum spicatum</i>							Effective		
<i>Potamogeton crispus</i>							Effective		

Pigment synthesis inhibitors (Herbicide Family 12, 13 and 27) are collectively called ‘bleachers’ because of the characteristic whitening of plant tissue (Armstrong 2012). These may inhibit chlorophyll or other pigments such as carotenoids. Fluridone is a slow-acting systemic herbicide used to control Eurasian Watermilfoil and other underwater plants. It may be applied as a pellet or as a liquid. Fluridone can show good control of submersed plants where there is little water movement and an extended time for the treatment. Its use is most applicable to whole-lake or isolated bay treatments where dilution can be minimized. It is not effective for spot treatments of areas less than five acres. It is slow-acting and may take 6-12 weeks before the dying plants fall to the sediment and decompose. Granular formulations of fluridone are proving to be effective when treating areas of higher water exchange or when applicators need to maintain low levels over long time periods. Although fluridone is considered to be a broad spectrum herbicide, when used at very low concentrations, it can be used to selectively remove Eurasian Watermilfoil. Some native aquatic plants, especially pondweeds, are minimally affected by low concentrations of fluridone. *Cabomba caroliniana* has been controlled in one waterbody in Michigan by whole lake application of fluridone (20 ppb).

Table 16. Nonindigenous plants susceptible to pigment synthesis inhibitors.

	Fluridone	Norflurazon	Isoxaflutole	Mesotrione
<i>Agrostis gigantea</i>			Proven effective on related species	Proven effective on related species
<i>Cabomba caroliniana</i>	Effective			
<i>Echinochloa crus-galli</i>		Effective		
<i>Myriophyllum spicatum</i>	Effective			
<i>Najas marina</i>	Effective			
<i>Najas minor</i>	Effective			
<i>Potamogeton crispus</i>	Effective if applied before dieback			

PPO Inhibitors (Herbicide Family 14) inhibit the photoporphyrinogen oxidase, an enzyme that is responsible for chlorophyll and heme biosynthesis (Armstrong 2012). PPO inhibition leads to accumulation of PPIX (protoporphyrin IX), which creates free radical oxygen in the cell and destroys cell membranes. Carfentrazone is proven effective for control of *Myriophyllum spicatum*. Flumioxazin has proven effective against *Cabomba caroliniana* in laboratory tests and in many Michigan waterbodies (200 ppb flumioxazin). LH Some applicators report control of *Nitellopsis obtusa* with 150 ppb flumioxazin.

VLFA Inhibitors (Herbicide Family 15) are herbicides that inhibit very long chain fatty acid synthesis in shoot tissue during germination of sensitive plants. Napropamide can be used for pre-emergent control of *Alnus glutinosa*.

Dichlobenil (Herbicide Family 20) inhibits actively dividing cells by disrupting formation of the cell plate. Germinating seedlings and actively growing plants are most affected by dichlobenil. Dichlobenil is effective against *Myriophyllum spicatum*, *Nymphoides peltata*, *Potamogeton crispus*, and *Rorippa sylvestris*. In rotation, it can be effective against *Polygonum persicaria* (this plant is known for developing resistance).

Endothall's herbicidal mode of action is not clear. It interferes with plant protein synthesis in some way and also affects lipid synthesis and dipeptide and proteinase activities (McDonald et al. 1993). The net result is a wilting of leaf tissue. Endothall may be applied in a granular or liquid form. Typically endothall compounds are used primarily for short-term (one season) control of a variety of aquatic plants. However, there has been some recent research that indicates that when used in low concentrations, endothall can be used to selectively remove

exotic weeds, leaving some native species unaffected. Because it is fast acting, endothall can be used to treat smaller areas effectively. Endothall has been proven effective against *Cabomba caroliniana* (Michigan applicators have reported that *Cabomba* populations in Michigan do not appear to be controlled by endothall in the field), *Myriophyllum spicatum*, and *Najas* spp. It can be effective against *Potamogeton crispus* if used before turion production.

Alkali grasses, such as *Puccinellia distans*, and other salt-tolerant invasives, such as *Solidago sempervirens*, outcompete other grasses in most brackish conditions. Thus, management efforts in the freshwater Great Lakes regions targeted at reducing salt contamination may benefit efforts to control these species.

Biological controls are employed for some plant species. Biological control agents do not completely eliminate the target, but when successful, can suppress populations to a nonsignificant level (Rees et al. 1996).

Grass Carp (*Ctenopharyngodon idella*) are voracious consumers of aquatic plants and frequently employed in the southern states for control of invasive plants. However, diploid (fertile) Grass Carp are illegal for use in most Great Lakes states. Grass Carp are emerging as an “aquatic species of concern” for the Great Lakes region (Great Lakes ANS Panel, Research Coordination Committee). The use of certified triploid (sterile) Grass Carp is allowed in Illinois, New York, and Pennsylvania, with the correct permits (Shiels and Hartle 2012, NYSDEC 2013). Since 1963, the Grass Carp, *Ctenopharyngodon idella* has been released to suppress Eurasian Watermilfoil and other nuisance aquatic plants in numerous sites within North America (Julien and Griffiths 1998, CEH 2004d). It has been found that Grass Carp may only eat Eurasian Watermilfoil after native plants have been consumed (IL DNR 2009a). To achieve control of Eurasian Watermilfoil generally means the total removal of more palatable native aquatic species before the Grass Carp will consume Eurasian Watermilfoil. In situations where Eurasian Watermilfoil is the only aquatic plant species in the lake, this may be acceptable. However, generally Grass Carp are not recommended for Eurasian Watermilfoil control. Likewise, Grass Carp will provide effective control of *Potamogeton crispus*, but may feed on native plants (CEH 2004e). Other bottom feeding fish, such as Common Carp, do not feed on *P. crispus*, but they create turbid water conditions and may prevent the growth of this plant species (CEH 2004e). Grass Carp will eat *Cabomba caroliniana*, but it is not their preferred food source. Grass Carp also feed on *Hydrocharis morsus-ranae*.

In their native range in Europe, ducks have been known to graze extensively on *Butomus umbellatus* (Hroudová et al. 1996). Beaver can have huge impact on willow species and are capable of eradicating entire stands (USDA NRCS 2002). Muskrat (*Ondatra zibethicus*) populations can have a serious impact on *Typha* populations. However, large populations of muskrats can shift to other plants species and have a long-term detrimental effect on the vegetation community (Miklovic 2000).

Van Leeuwen (1983) found that a combination of European grazers (rabbits, the hoverfly *Cheilosia grossa*, and *Epiblema scutula*) resulted in an approximately 30% reduction in flower heads on *Cirsium palustre*. Furthermore, plants that had suffered predation had a reduced stem height, resulting in a reduced seed dispersal distance of surviving achenes (van Leeuwen 1983). Additional research is needed to determine if native species of rabbits and insects could have similar results on controlling *C. palustre* in the Great Lakes. Bitter Dock (*Rumex obtusifolius*) is avoided by rabbits, but docks appear to be a favorite food plant of deer (Amphlett and Rea 1909).

Grazing of domestic herbivores can be an effective method of control for many palatable species. Allowing cattle or sheep access to areas infested with *Impatiens glandulifera* will control the population and the spread of the species either by direct grazing or by trampling of young seedlings (CEH 2004b). Cattle, horses, and sheep graze on *Juncus* spp., but the extent of control gained from grazing is unknown (CEH 2004a, Cosyns et al. 2005). Docks (*Rumex* spp.) are grazed by cattle, sheep, and goats but not by horses. Targetted grazing by sheep has been used as a biocontrol for Purple Loosestrife (Kleppel and LaBarge 2011). Goats are attracted to the flowering stage of many thistles, including *Cirsium palustre*. Only about 0.5% of thistle seeds that pass through their digestive systems remain viable, making it unlikely that they would aid in the spread of this species. Effective grazing could reduce Marsh Thistle populations, although it is unclear whether grazing would ultimately control *C. palustre* via the trampling of rosettes or facilitate its spread through the creation of safe sites for germination (Fraser 2000). Reseeding of native vegetation may enhance the success of prior control efforts. Moreover, goats do not select for Marsh Thistle and may also eat native thistles in intermingled communities (Popay and Field 1996). Heavy grazing will eliminate *Typha* spp. from riparian corridors; however, this technique might also affect other native species (Stevens and Hoag 2006).

Five species of beetles have been approved for the biocontrol of *Lythrum salicaria* (Blossey et al. 1994ab). *Galerucella californiensis* and *G. pusilla* are both leaf-feeding chrysomelids. These beetles defoliate and attack the terminal bud area, drastically reducing seed production. The mortality rate to Purple Loosestrife seedlings is high. Evidence of *Galerucella* spp. damage are round holes in the leaves. Four to six eggs are laid on the stems, axils, or leaf underside. The larvae feed constantly on the leaf underside, leaving only the thin cuticle layer on the top of the leaf. Initial introductions in eastern North America occurred in Virginia, Maryland, Pennsylvania, New York, Minnesota, and southern Ontario in August 1992 (Hight et al. 1995). In 1992 these three beetles were released in Washington. *Galerucella* spp. populations visibly impacted Purple Loosestrife stands by 1996 (Washington State Department of Ecology 2012). In the Great Lakes region, Sea Grant conducted an extensive, multi-state program involving youth in raising and releasing *Galerucella* beetles for control of *L. salicaria* (Michigan Sea Grant 2001).

Hylobius transversovittatus is a root-mining weevil that also eats leaves (Wilson et al. 2004). This beetle eats from the leaf margins, working inward. The female crawls to the lower 2-3

inches of the stem then bores a hole to the pithy area of the stem where 1-3 eggs are laid daily from July to September. Or, the female will dig through the soil to the root, and lay eggs in the soil near the root. The larvae then work their way to the root. *H. transversovittatus* damage is done when xylem and phloem tissue are severed, and the carbohydrate reserves in the root are depleted. Plant size is greatly reduced because of these depleted energy reserves in the root. The presence of larvae is evidenced by zig-zag patterns in the root.

Nanophyes marmoratus and *N. brevis* are seed eating beetles (Blossey 2002a, Wilson et al. 2004). Young adults feed on shoot tips, later feeding on the flowers and closed flower buds. Sixty to one hundred eggs are laid in the immature flower bud. Seed production is reduced by 60%. There were two test sites releases in 1996. Approval to introduce *N. marmoratus* was granted followed by introductions in New York and Minnesota in 1994. Additional releases occurred in New Jersey in 1996. *N. marmoratus* has also been released in Ohio (Ohio EPA 2001). Release of *N. brevis* planned for 1994 was delayed due to contamination of the original shipment with a parasitic nematode (Piper 1997). This infection appeared benign for *N. brevis*, however, due to the potential for non-target effects of the nematode after introduction into North America, only disease free specimens should be introduced, which, at present, effectively precludes the introduction of *N. brevis* (Blossey 2002a). *Bayeriola salicariae*, a gall midge, was studied and screened between 1990 and 1992 (Blossey and Schroeder 1995). Based on results indicating a potential wider host range, the gall midge *B. salicariae* was not proposed for introduction (Blossey and Schroeder 1995).

Many invasive plants are not susceptible to grazing. For example, Lupines (*Lupinus polyphyllus*) may be toxic, and populations often increase in grazed (pasture) systems. Several native insects feed on Lupines, but are considered insufficient for control (DiTomaso 2013). Chemical defense indicates that the use of biocontrol agents on invasive populations of *Cabomba caroliniana* may not be a viable approach (Morrison and Hay 2011). When fed on by crayfish and snails, *C. caroliniana* induces a chemical defense mechanism to deter both herbivores and microbes that typically attack plants via openings left by herbivores (Morrison and Hay 2011).

Research into potential biocontrol via host-specific and native insects is ongoing for many species of invasive aquatic plants. A North American weevil, *Euhrychiopsis lecontei*, may be associated with natural declines of *Myriophyllum spicatum* at northern lakes (Sheldon 1994, Creed and Sheldon 1995). *E. lecontei* feeds on the new growth of *M. spicatum* and can help keep populations under control; it is common for the populations of *E. lecontei* and *M. spicatum* to exhibit the classic predator-prey cycles (Creed and Sheldon 1995, Michigan Sea Grant 2012b). Studies have found the herbivorous weevil causes significant damage to Eurasian water-milfoil while having little impact on native species, suggesting the insect as a potential biocontrol agent (Creed and Sheldon 1995). Female weevils have higher fecundity when raised on *M. spicatum* as opposed to native *M. sibiricum* (Solarz and Newman 1996, Creed 1998, TNC Vermont 1998, Sheldon and Jones 2001).

The defoliating Hemlock Moth (*Agonopterix alstroemeriana*) was accidentally introduced to the United States, but it is now being investigated as a potential biocontrol agent because of its monophagous (feeding on a single food source) association with *Conium maculatum* (Castells and Berenbaum 2006). These moth larvae feed on the young stem tissue, flowers, and seeds (Forest Health Staff 2006d). High densities of *A. alstroemeriana* have been effective drivers of plant mortality in *C. maculatum* stands in the western United States, where several hundred larvae have been reported from a single plant. However, as a chemical defense, alkaloid production appears to increase with *A. alstroemeriana* herbivory, potentially driving surviving populations to higher levels of toxicity over time (Castells et al. 2005). Furthermore, *A. alstroemeriana* was found to be targeted by a predatory wasp (*Euodynerus foraminatus*) in Illinois, suggesting that the effectiveness of biocontrol may be lessened in the Midwest and other locations where *E. foraminatus* exerts top-down pressure on *A. alstroemeriana* (Castells and Berenbaum 2008). Although *A. alstroemeriana* is widespread in the United States, larvae may still be difficult to obtain for biocontrol purposes (Castells and Berenbaum 2006). *Trichoplusia ni*, the cabbage looper, is a generalist lepidopteran that is found throughout the United States and occasionally feeds on *C. maculatum*. Overall growth of *T. ni* is not stunted, but larvae raised on diets enriched with the piperidines found in *C. maculatum* develop slower. A prolonged larval stage makes *T. ni* more vulnerable to predators and could reduce overall biocontrol capabilities (Castells and Berenbaum 2008). *Papilio poluxenes*, black swallowtail butterfly, will lay eggs on *C. maculatum*, but Feeny et al. (1985) found low larvae survivorship in central New York.

While there are no specific biocontrol agents for *Cirsium palustre* (GLIFWC 2006), herbivory by a variety of species may be beneficial but requires additional research. Promising biocontrol candidates include a European seedhead fly, *Terellia ruficauda* (Fraser 2000, OLA, and MAFF 2002); the seed-eating weevil, *Rhinocyllus conicus*, currently undergoing experimental trial in the Robson Valley Forest District, British Columbia (OLA and MAFF 2002, USDA Forest Service 2005b); and the glassy cutworm, *Apamea devastator* (native in New York and Ohio; Volger and Stressler 2011). The latter is an indiscriminate herbivore known to feed on *C. palustre* and may help control Marsh Thistle; however, this moth feeds on a broad spectrum of additional plants. Larvae of the artichoke plume moth (*Platyptilia carduidactyla*) also feed on Marsh Thistle, but as its common name suggests, this species is considered a pest to artichokes (Winston et al. 2008). Furthermore, the moth's native range is south of the Great Lakes. Occasionally *Cheilosia corydon*, a fly native to Italy, feeds on Marsh Thistle (Winston et al. 2008). This fly was released in Oregon in 1991 to control several invasive thistle populations. However, since its release, *C. corydon* populations have attacked native and exotic thistles indiscriminately (ODA Plant Division 2011).

Elephant moth (*Deilephila elpenor*) feeds on *Epilobium hirsutum*, but is not a native to the Great Lakes (Hoskins 2012, Pittaway 2012). Genetic material extracted from *E. hirsutum* individuals displaying phyllody of flowers and/or plant yellowing revealed infection by *epilobium* phyllody

(EpPh) phytoplasma, an obligate, parasitic bacteria that attach to phloem tissue (Alminaité et al. 2002). The ability of this phytoplasma to act as a biocontrol agent is still unknown.

The use of the stem-boring larvae of the weevils *Apion violaceum* and *A. miniatum* has been investigated for controlling *Rumex obtusifolius* (Hopkins 1980, Freese 1995). In the UK and elsewhere, the Chrysomelid Beetle (*Gastrophysa viridula*) has been investigated as a biocontrol agent for both *R. obtusifolius* and *R. crispus* (Bentley et al. 1980). Larvae of the leaf-mining fly *Pegomya nigratarsis* cause blotch mines on leaves of *R. obtusifolius* (Whittaker 1994). *R. obtusifolius* is the preferred host plant of *Coreus marginatus* and has been shown to moderately reduce its seed viability (Hruskova et al. 2005).

In its native range in China, the leaf beetle *Galerucella birmanica* has significant negative impacts on *Trapa natans* populations (Ding et al. 2006). However, this species has many other host species in the United States, making it unsuitable for use as a biocontrol agent (Maryland Sea Grant 2012).

The species *Aphis fabae*, *Impatiens glandulifera*, and *Deilephila elpenor* are known to feed on Ornamental Jewelweed (*Impatiens glandulifera*), but their capacity to act as biological control agents is still unknown (Beerling and Perrins 1993). Although an initial experiment by Tanner (2011) indicated that *D. elpenor* exhibited lower biomass and survivorship when raised on *I. glandulifera*.

Aphids may occasionally feed on *Juncus* spp., but most rushes are fairly resilient to extensive damage from insect or diseases (Stevens and Hoag 2003). Gypsy moths can defoliate Purple Willow and willow midges can cause significant (though rarely fatal) damage. The native boring-moth larvae (*Arzama* spp.) have been reported to cause damage to *Typha* stands, but their use as a species-specific biological control is unknown (Miklovic 2000). *Chrysolina herbacea* feeds on *Mentha aquatica*, despite the deterrents this species produces to minimize damage caused by herbivores (Atsbaha Zebelo et al. 2011). The fruit fly *Acinia picturata* has been known to use *Pluchea odorata* as a host, but it is unknown if this species could be used a biological control agent (Stegmaier 1967).

Pathogens are also being explored as potential biocontrol agents (biopesticides regulated by EPA as pesticides). *Conium maculatum* is susceptible to multiple viruses, including ring spot virus, carrot thin leaf thin virus (CTLV), alfalfa mosaic virus (AMV), and celery mosaic virus (CeMV) (Howell and Mink 1981). However, viral infections appear to stunt growth rather than cause mortality, diminishing their potential for biocontrol (Howell and Mink 1981, Pitcher 2004). Another disadvantage to using these types of biological control agents is the potential for them to escape into neighboring habitats, especially agricultural fields (J. McHenry pers. comm. in Pitcher 1985). The fungal pathogen *Exserohilum monoceras* has shown some success in controlling Barnyard Grass, *Echinochloa crus-galli* (Catindig et al. 2009). In its native range, *Impatiens glandulifera* has been known to harbor *Puccinia komarovii* (a rust pathogen), which is

currently undergoing research as a control agent (Tanner 2011). Laboratory research has shown that the fungus *Mycoleptodiscus terrestris* reduces the biomass of *Myriophyllum spicatum* significantly and may be a possible biocontrol agent (IL DNR 2009a). Although *Polygonum persicaria* plants are susceptible to *Arabidopsis* mosaic virus, no research has been undertaken on the development of biological control agents, whether viral or fungal.

The Leaf Spot Fungus *Ramularia rubella* causes red spots to develop on dock leaves but has no major effect on plant survival. The Rust Fungus *Uromyces rumicis* is also non-systemic but has been shown to have some potential as a biological control agent (Inman 1971, Schubiger et al. 1986). Dock species are also an alternate host for number of viruses, fungus (Dal Bello and Carranza 1995), and nematodes (Townshend and Davidson 1962, Edwards and Taylor 1963). Willow blight is a serious pest to *Salix* plantings, and has been documented to kill plants damaged by storms, indicating a potential for use in combination with mechanical control (USDA NRCS 2002).

Manipulation of competitors can also be an important element of biological control. Control of many species has the best long-term efficacy when followed by replanting with native species, which can outcompete seedlings of the invasive. For example, revegetation of disturbed riparian sites can be used to prevent Purple Loosestrife establishment and to reduce re-establishment after control procedures are applied. Fowl Mannagrass (*Glyceria striata*), Foxtail Sedge (*Carex alopecoidea*), and Reed Canarygrass (*Phalaris arundinacea*) have achieved dominance and prevented re-invasion in plots where Purple Loosestrife was experimentally removed (Lui et al. 2005). Smartweed (*Polygonum lapathifolium*) is reported to out-compete Purple Loosestrife during its first year of growth. Seeding Japanese Millet (*Echinochloa frumentacea*, also called billion-dollar grass) after drawdown and before Purple Loosestrife seedlings began to grow provided control (Jacobs 2008). Combining herbicides with overseeding an alternative desired grass will help discourage the regrowth of surviving *Poa trivialis* and improve overall success of control (Morton and Reicher 2007).

4.3 Fish

Harvesting of invasive fish is generally only effective if the species is of importance to fisheries and anglers. Even when Bighead and Silver Carp were considered for harvest, harvest was found to be one of the least effective methods available (Linfield 1980, Vacha 1998, Koehn et al. 2000, Wedekind et al. 2001, Arlinghaus and Mehner 2003).

Physical removal of fish can be an effective control technique in small ponds and other confined/constrained systems. Fish have been captured by drawdown, netting, gillnetting, trapping, and electroshocking. Goldfish and carp have frequently been managed in this way (Ritz 1987, Morgan et al. 2005, Pinto et al. 2005). Fine-mesh monofilament gill nets have been used to control Rudd in three shallow lakes in Waikato, New Zealand, but elimination was not achievable (Neilson et al. 2004). Small, potentially fecund fish in dense littoral vegetation proved

challenging to net, presenting a problem for total eradication, but removal of larger Rudd likely affected breeding success and netting is seen as a highly cost effective control method with low environmental impact (Neilson et al. 2004). Common Carp display jumping behavior when trying to escape entrapment. The Williams cage exploits this behavior by selectively removing the jumping carp from other fish (Stuart et al. 2011). Tests of the Williams cage in Australia proved to be extremely successful. Over a three-year test period, the Williams cage successfully separated 88% of adult Common Carp and allowed 99.9% of native species to pass through. The Williams cage is useful in controlling dispersal and abundance of Common Carp. The United States National Park Service uses physical removal through electrofishing to manage Rainbow Trout and Brown Trout populations that threaten native Brook Trout in Shenandoah National Park, Virginia (NPS 2011).

Barriers of several types have been used to control the spread of fish species and to prevent upstream migration or otherwise limit their access to spawning grounds. When using physical deterrents as barriers, combining methods can increase effectiveness. For example, Patrick et al. (1985) found that pelagic estuarine and freshwater fishes were successfully deterred by a barrier combining air bubbles and strobe lights.

Barriers and traps have been effective controls of Sea Lamprey since the 1950s. Barrier options include mechanical weirs, electrical barriers, low-head barriers, adjustable crest barriers, and velocity barriers (Scott and Crossman 1973, Smith and Tibbles 1980, GLFHC 2000ab). Traps are often used in association with barriers to capture Sea Lamprey while allowing desired species to continue upstream (GLFHC 2000a, FOC 2009).

Barriers including electric, bubble curtain, and sonic have been used to exclude Common Carp from industrial cooling intake structures (Koehn et al. 2000). When possible, Common Carp can be excluded from an area and then kept out through sorting of fish (allowing desirable species to pass a barrier), which has been done since 1997 at the Cootes Paradise Marsh in Hamilton, Ontario (Lougheed et al. 2004). Although labor intensive, this method has proven effective at keeping Common Carp from returning to the marsh.

Electrical barriers and phonotaxis traps (cages with conspecific acoustic calls which allure gravid females) may be successful at limiting the movement of Round Goby. In tank studies, Round Gobies did not move through a barrier (Savino et al. 2001). Rollo et al. (2007) reported Round Gobies will approach a speaker emitting conspecific male calls in the field, and female round gobies showed significant attractions to speakers emitting conspecific male calls in the laboratory. Therefore phonotaxis could be used to lure gravid females to traps. As Round Gobies will spawn multiple times throughout late spring and summer (Jude et al. 1992), they should remain receptive to male calls and bioacoustic capture for the entire breeding season.

Homing species such as salmonids typically return to their native stream for reproduction. Barriers can be constructed and/or natural barriers augmented to prevent upstream migration and

aid management and eradication efforts. Lintermans and Raadik (2003) noted three key aspects of successful barriers to Rainbow Trout migration: there is a 1.5 m or greater vertical drop from the barrier to the pool below; in higher flows, the direction of water flow is towards the middle of the barrier with no slower overland flow passing down the banks; and no deep pool below the barrier from which fish could jump.

The United States Army Corps of Engineers Great Lakes and Mississippi River Interbasin Study (USACE GLMRIS) noted the potential effectiveness of sensory deterrent systems in providing barriers to fish migration or eliciting fish movements (USACE 2012b). Specifically, Maiolie et al. (2001) reported success using underwater strobe lights to deter wild, free-ranging *Oncorhynchus nerka* in two large clear Idaho lakes an average distance of 30-136 m away, with an 80% reduction in fish density within 30 meters of the strobe lights. Models demonstrated strobe lights could be a successful deterrent to entrainment of Rainbow Smelt through Oahe Dam, Lake Oahe, South Dakota (Hamel et al. 2008). Patrick et al. (1985) found that combining strobe lights with air bubbles in a barrier would increase effectiveness of a barrier to Rainbow Smelt and other pelagic fishes.

The GLMRIS study also noted the potential effectiveness of acoustic fish deterrents in controlling or deterring *Proterorhinus semilunaris* populations. Acoustic deterrents include continuous wave and pulsed wave technology, which use sound/pressure waves to influence the behavior of aquatic organisms. Similarly, sensory deterrent systems such as acoustic air bubble curtains, electric barriers, and underwater strobe lights may prove useful in controlling populations in waterways and small bodies of water, but there are no studies of their effects on *P. semilunaris* at the present time (USACE 2012b).

Water level manipulation has been used to to disrupt spawning of Common Carp (Summerfelt 1999) and to exclude them from spawning habitat (Lougheed and Chow-Fraser 2001). Yamamoto et al. (2006) noted that physical drawdown of water levels has significant negative effects on cyprinid spawning abilities in Lake Biwa, Japan. *Carassius* spp. and *Cyprinus carpio* eggs were notably reduced after collection when water levels were lowered by 30 cm, and as little as a 10 cm reduction significantly reduced available shallow, litter-accumulated spawning areas (Yamamoto et al. 2006).

Chemical piscicides are commonly used to control or eradicate invasive fishes, especially in rapid response scenarios. It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fish, invertebrates, and other fauna or

flora, and their potential harmful effects should therefore be evaluated thoroughly (Finlayson et al. 2002).

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered “general” piscicides. Antimycin and rotenone are non-selective, and toxicity to other fishes and aquatic invertebrates will vary (USACE 2013). TFM (3-trifluoromethyl-4-nitrophenol) and niclosamide (2',5-dichloro-4'-nitrosalicylanilide) are registered as lampricides, but show some promise for control of other invasive fishes as well (USACE 2013).

Rotenone is arguably the best known and most commonly used of the registered piscicides. The International Joint Commission (2011) recommends rotenone for control of White Perch and Round Goby in rapid response scenarios. Rotenone is also effective against Rainbow Trout, but at much higher concentrations than antimycin (Gilderhus 1972, Finlayson et al. 2002).

Antimycin A is a registered piscicide in the United States that is documented as highly effective against scaled fishes. Antimycin is most effective in small streams, shallow ponds, and alpine lakes where there is ample mixing and an adequate contact time can be achieved (Gilderhus 1972, Finlayson et al. 2002). Antimycin does not seem to repel fish like rotenone, and rapidly breaks down by hydrolysis in natural waters (Finlayson et al. 2002). Disadvantages of antimycin include increasing ineffectiveness in waters with higher pH (>8), streams with significant gradients (80-150 m elevation drop), and large lakes where good mixing and contact time cannot be established (Finlayson et al. 2002). In a study on removal of toxic chemicals from water using activated carbon, Dawson et al. (1976) found that granular activated carbon is saturated by rotenone. Antimycin was efficiently absorbed and did not saturate carbon at similar effective doses (Dawson et al. 1976). Antimycin-impregnated baits have been used to target Common Carp (Rach et al. 1994).

TFM has been used primarily to kill larval lampreys in their stream habitats (Smith and Tibbles 1980). Beginning in the late 1950s, Sea Lampreys have been successfully controlled using TFM. The lampricide has reduced the population by over 90% of the 1961 peak (Scott and Crossman 1973). However, continued use of TFM is required to keep populations under control (Scott and Crossman 1973, Becker 1983). TFM is sometimes harmful to other fish (e.g., Walleye) as well as to the larvae of nonparasitic native lamprey species (Becker 1983).

Bayluscide (niclosamide) treatments in deltas are also a widely used and an effective control of Sea Lamprey larvae (NYSDEC 2012a). Exposure to niclosamide is known to be toxic to all fish species at 0.5 mg/L after a 48-h exposure (Clearwater et al. 2008). Niclosamide has been used for control of aquatic snails, Zebra Mussels, and oligochaetes, and is also toxic to many crayfish, frogs, clams, algae, and other amphibian and fish species (Clearwater et al. 2008).

Boogaard et al. (2003) found that the lampricides TFM and niclosamide demonstrate additive toxicity when combined. Evaluation of the effects of common piscicides on Ruffe revealed that the lampricide 3-trifluoromethyl-4-nitrophenol (TFM) has potential for selective control of the

species (Boogaard et al. 1996). Boogaard et al. (2003) found that Brown Trout are among the least sensitive fishes to the lampricides. Ruffe was three to six times more sensitive to TFM than both Yellow Perch (*Perca flavescens*) and Brown Trout (Boogaard et al. 1996). A cost benefit analysis of a United States Ruffe control program supported TFM as a promising chemical control (Leigh 1998). However, Dawson et al. (1998) suggest that TFM may have more application for treating entire bodies of water rather than localized areas because it tended to repel Ruffe in preference tests, allowing them to move to untreated areas. Bottom-release formulations of bayluscide and antimycin showed promise for effectiveness in treating localized concentrations of Ruffe (Dawson et al. 1998).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

Changes in pH are known to affect fish behavior. Ikuta et al. (2003) documented the effects of low pH on Sockeye Salmon, noting that salmon would not swim upstream into areas of pH lower than 6.0. Acute exposure to low pH levels can directly kill fish by discharge of sodium and chloride ions from body fluid, and sub-lethal levels can affect reproduction. In the case of Sockeye Salmon, weak acidic conditions of <pH 6 were enough to depress the prespawning behavior of swimming upstream. Increases in pH through liming are listed as a potential control method for Goldfish (Clearwater et al. 2008).

Biological controls have also been commonly employed to control invasive fish species. Techniques range from the general – e.g., ‘top-down’ control to influence predator-prey balance – to exploitation of species-specific interactions. Because the trade-off between fish species as agents of biological control is not directly correlated with consumption, management decisions involving shifts between species should not take consumption solely into account (Stewart et al. 1981).

Interactions among Sea Lamprey, salmonids (including several nonindigenous species), and the prey base (especially Alewife and Rainbow Smelt) have been key to biological controls targeting these species. Managing each of these species significantly impacts the others. Pacific salmon have significant environmental, socio-economic, and beneficial effects in the Great Lakes and therefore integrated management is essential. Pacific salmon prey heavily upon Alewife and Rainbow Smelt; Alewives remain a key food source and crucial to the survival of Pacific salmon. Over the past several decades, Pacific salmon populations have fluctuated with fluctuating Alewife populations. The management response to Great Lakes Alewife overabundance and recurring die-offs was to invest in Sea Lamprey (*Petromyzon marinus*) control and planting of

hatchery-reared Pacific salmonids (*Oncorhynchus* spp.) to re-establish top open-water predators (Kocik and Jones 1999, Hansen and Holey 2002). Older and larger fish tend to be most heavily affected by piscivores, while smaller and younger fish remain abundant (Hewett and Stewart 1989). Rainbow Smelt are also a major component of Pacific salmon diet. The presence or absence of this species significantly alters predator-prey relationships and competition between native species. Several species of non-native salmonids have been introduced to the Great Lakes, beginning in the 1960s, to control invasive Rainbow Smelt (USACE 2012b). Rainbow Smelt is heavily consumed by Atlantic Salmon (*Salmo salar*), Lake Trout (*Salvelinus namaycush*), Brook Trout (*Salvelinus fontinalis*), Coho Salmon (*Oncorhynchus kisutch*), Chinook Salmon (*Oncorhynchus tshawytscha*), Rainbow Trout (*Oncorhynchus mykiss*), Brown Trout (*Salmo trutta*), Splake (Brook Trout x Lake Trout), Burbot (*Lota lota*), Walleye (*Sander vitreus*), Northern Pike (*Esox lucius*), and many other freshwater piscivores (Stewart et al. 1981, Brandt and Madon 1986, Crossman 1991, He and LaBar 1994, Kirn and LaBar 1996, USACE 2012b). Observed Atlantic Salmon predation on smaller Rainbow Smelt, as well as bioenergetics modeling suggesting that by age 4, cumulative piscivory by Atlantic Salmon was nearly 10-fold greater than that of Lake Trout of the same age, implies its greater usefulness for management of Rainbow Smelt (Kirn and LaBar 1996). While Lake Trout consume large amounts of Rainbow Smelt, almost exclusively so in some studies, the species is believed to provide little potential for responsive management manipulation outside of stabilizing fluctuating prey populations, due to the long cycle of its predatory effect (peaking 3-5 years after stocking, lasting 7-8 years) (Stewart et al. 1981, He and LaBar 1994, Kirn and LaBar 1996). Chinook Salmon have been successfully used to eradicate Rainbow Smelt from small lakes in New Hampshire in 1936 (Stewart et al. 1981). Of the 23 nonindigenous diseases and parasites in the Great Lakes, *Aeromonas salmonicida*, *Renibacterium salmoninarum*, *Myxobolus cerebralis*, and *Novirhabdovirus* sp. infections have been realized in Great Lakes Pacific salmon, while *Heterosporosis* sp. and *Piscirickettsia* cf. *salmonis* infections have been realized clinically or outside the Great Lakes. *Glugea hertwigi*, a microsporidian, is known to cause mortality in rainbow smelt. Therefore, Pacific salmon management must include the management of these pathogens and parasites.

Minnesota and Wisconsin, with advice from the U.S. Fish and Wildlife Service, implemented a top-down control program for Ruffe in the St. Louis River, western Lake Superior, in 1989, using Northern Pike (*Esox lucius*), Walleye (*Sander vitreus*), Smallmouth Bass (*Micropterus dolomieu*), Brown Bullhead (*Ameiurus nebulosus*), and Yellow Perch (Mayo et al. 1998). A bioenergetics modeling evaluation of the top-down control program revealed that although predators ate as much as 47% of Ruffe biomass in one year, they avoided Ruffe and were selective for native prey, and were thus unable to halt the increase in Ruffe abundance. However, the authors noted that Northern Pike and Walleye appeared to have potential for top-down control of ruffe due to a combination of their diets and population sizes, and due to indications that they may learn to prey more selectively on Ruffe. As Mayo et al. noted, caution is advised when considering top-down biological control as a management tool because the stability

properties of a system do not just depend on predation, but also on the life histories of component species and their interactions.

Bottom-up control (reduction in food supply) of White Perch usually results in stunting accompanied by an increase in population so that the population consists of many small fish (Smith et al 2002).

Northern pike, *Esox lucius*, have been used as a biological tool to control Common Carp recruitment in the Sandhill lakes in Nebraska (Paukert et al. 2003). Great Lakes native Burbot were reported to depress abundance of Round Goby in eastern Lake Erie (Madenjian et al 2011).

An effective bio-control of Sea Lamprey is the implementation of the sterile-male-release program. Male Sea Lamprey are captured during spawning runs, sterilized using bisazir, and released to compete with fertile males for mating; thus reducing egg fertilization. Released males are sterilized past their parasitic phase and do not return to the lake. (FOC 2009, GLFHC 2000a).

Application of different pheromones such as migratory, alarm, and sex may be useful in the integrated management of carp (Sorensen & Stacey 2004) as well as Sea Lamprey.

Inducible Fatality Genes (IFG) involve breeding fishes with a fatal genetic weakness to a trigger substance, such as zinc. The fatal gene technology appears to be a potentially viable and long-term strategy for the environmentally benign control of carp (Koehn et al 2000).

4.4 Mollusks

Eradication of mollusks from infested open waters is unlikely – emphasis is generally on preventing further spread, limiting population impacts (especially relating to infrastructure such as water intakes), and controlling new infestations in small water bodies (e.g., ponds, water gardens).

Physical controls can be an important component in the control of invasive mollusks. With the possible exception of the large ‘escargot’ snail species (*Cipangopaludina* and *Viviparus* spp.), there is not a significant harvest of any of the Great Lakes nonindigenous mollusk species and harvest is not considered to impact control. Physical removal has little effect on control in ecosystems, but can be employed to protect infrastructure (e.g., water intakes, swimming beaches), usually in conjunction with physical barriers. Screens and traps are commonly employed to prevent *Corbicula* colonization of water intakes (GISD 2013). Diver assisted suction removal and bottom barriers are being researched as potential methods for physical control of *Corbicula* populations in Lake Tahoe (UC Davis TERC 2004). Benthic barriers have been effective for short-term control of *Corbicula fluminea*, but non-target mortality to other benthic invertebrates may be high (Wittmann et al 2012). Effective physical controls of *Dreissena* include infiltration intakes or sandfilter intakes (to filter out veligers), thermal treatments, carbon dioxide pellet blasting, high-pressure water jet cleaning, mechanical cleaning, freezing, scraping, scrubbing, pigging, and desiccation. Placement of water intakes in areas too deep or otherwise unsuitable for zebra mussel colonization has been used as a form of physical

control, but this is less successful in areas that also have Quagga Mussels. Other potential dreissenid controls include the use of electrical fields, pulse acoustics, low-frequency electromagnetism, ultraviolet light (UV light), and reduced pressure (USACE 2002). New Zealand Mudsnaill control in hatcheries has included using a flame thrower against the walls of raceways, since mudsnails cannot withstand warm temperatures (Dwyer et al. 2003, Richards et al. 2004) or low humidity situations (Dwyer and Kerans, unpublished, Richards et al. 2004). New Zealand Mudsnaills are also sensitive to freezing and can be eradicated if an infested area is drained in the winter, and the substrate is frozen to the depth containing the mudsnails. There is preliminary evidence that hydrocyclonic separators may be a useful tool to decontaminate fish hatchery water supplies and prevent the spread of New Zealand Mudsnaills within a hatchery.

Physical removal of visible vegetation (which may harbor invasive mollusks) from boats, trailers, and other equipment being moved from one water body to another is an important method in controlling spread. Asian clam, *Corbicula fluminea*, may be controlled at intake pipes by heating influent water to 99°F (GISD 2013). Flushing engines, cooling systems, live wells, and bilge with water over 110°F will kill dreissenid veligers and 140°F will kill adults. The New Zealand Mudsnaills can survive at 43.3°C, so the water temperature needs to exceed 120°F to eliminate that species (Medhurst 2003). Drying fishing gear at 28.9-30°C for at least 24 hours or at 40°C for at least two hours will eliminate New Zealand Mudsnaills (Richards et al. 2004). Air drying equipment for 5 days will kill most dreissenid larvae and smaller dreissenid mussels, but large mussels may survive up to two weeks out of water. Dessication (drying) is not an effective control method for *Cipangopaludina chinensis*. Field experiments under mesic conditions indicated that this snail can survive exposure to air for at least 4 weeks (Havel 2011). Putting fishing gear in a freezer for 6-8 hours will kill all attached NZ mudsnails (Medhurst 2003, Richards 2004).

Preliminary research demonstrates that *Cipangopaludina chinensis* will not migrate upstream against a small current (Rivera 2008). Authors suggest that acceleration of current may be an important management tool for preventing upstream spread.

Many chemical molluscicides are available, but are not species-specific and may harm native species to a greater extent than non-natives. Many of the nonindigenous mollusk species can close their shells tightly, reducing the effectiveness of chemical controls. In short-term experiments, *Sphaerium corneum* can reduce the bioaccumulation of 2,4,5-trichlorophenol (TCP) by closing their shell valves (Heinonen et al 1997) – this reaction to chemical stimuli generally may limit the usefulness of chemical molluscicides against this species. Likewise, *Cipangopaludina* spp. can close their operculum tightly and have been shown to be more resistant to chemical treatment than native snails without that ability.

Molluscicides are typically classified as either oxidizing or non-oxidizing compounds. Oxidizing chemicals include chlorine, chlorine dioxide, chloramines, ozone, bromine, hydrogen peroxide, and potassium permanganate. Non-oxidizing chemicals (including organic film-forming antifouling compounds, gill membrane toxins, and nonorganics) can be classified into several distinct groups: quaternary and polyquaternary ammonium compounds; aromatic hydrocarbons; endothal as the mono (N,N-dimethylalkyl amine) salt; metals and their salts (e.g., copper sulfate formulations); and niclosamide (including some formulations of Bayluscide). Bayluscide was

initially developed as a Sea Lamprey larvicide, but has molluscicidal activity. While some of these products are biodegradable, many require detoxification or deactivation to meet state and Federal discharge requirements (USACE 2012).

Chlorination has been the most common treatment for control of *Dreissena* mussels, but chlorine concentrations needed for effective control of quagga mussels may reach hazardous levels (Grime 1995). Oxidizing chemical control treatments effective against *D. polymorpha* include hypochlorite, chloramine, chlorine dioxide, bromine, ozone, potassium permanganate, and sodium chlorite.

Corbicula is not tolerant of fluctuating environmental conditions (particularly temperature and oxygen) and is prone to massive die-offs (Menninger 2013), this suggests that short-term chemical manipulation may be useful in controlling *Corbicula* populations. Low concentration of chlorine or bromine will kill juvenile Asian Clams (GISD 2013).

Chemical methods used to eradicate New Zealand Mudsnaills include: Bayer 73, copper sulfate, and 4-nitro-3-trifluoromethylphenol sodium salt (TFM). The only molluscicide known to have been tested against New Zealand Mudsnaills is Bayluscide (niclosamide). Hyamine and hydrogen peroxide have also been used to control New Zealand mud snails (IJC 2011). Preliminary investigations also suggest that copper and carbon dioxide under pressure may prove useful in both decontaminating fish hatchery water supplies and preventing spread into uncontaminated areas of a hatchery. Ozone has not been shown to be effective in killing New Zealand Mudsnaills in a hatchery environment.

Various chemical coatings – including copper-based, tributyltin-based, copolymer, vinyl/epoxy, resin and other films - can be applied to structures to prevent the attachment of Zebra Mussels. Tributyltin-based antifoulants are extremely toxic and restricted by federal law (Ohio Sea Grant 1992).

Biological control so far has proven to be ineffective in controlling *Dreissena* species. Predation by migrating diving ducks, fish species, and crayfish may reduce mussel abundance, though the effects are short-lived (Bially and MacIsaac 2000). Other biological controls (biopesticides) being researched are selectively toxic microbes and parasites that may play a role in management of *Dreissena* populations (Molloy 1998). Laboratory testing shows strain CL145A of *Pseudomonas fluorescens* (a bacterium) to be highly lethal to Zebra Mussels; capable of eliminating over 90% of adults and 100% of larvae (Molloy 2002, Abdel-Fattah 2011). Commercially, this product is known as Zequanox® and is developed by Marrone Bio Innovations (Abdel-Fattah 2011) – this product has been registered by EPA for use as a pesticide. Interfering with the synchronization of spawning by adults in their release of gametes could also offer control of *Dreissena* populations (Snyder et al. 1997). Another approach would be to inhibit the planktonic veliger (larvae) from settling and attaching to a surface to begin development (Kennedy 2002).

Competition with dreissenid mussels will likely limit expansion of *Lasmigona subviridis* and other large nonindigenous bivalves in the Great Lakes.

Manipulation of predator fishes and turtles that eat snails may be useful in the control of snail populations. However, as a relatively large snail species, *Cipangopaludina chinensis*, may escape predation by smaller fishes. Parasites of New Zealand Mudsnails from New Zealand may also become useful to control population size by inhibiting reproduction. Studies of the efficacy and specificity of a trematode parasite from the native range of New Zealand Mudsnails as a biological control agent have shown positive results so far (Dybdahl et al. 2005).

4.5 Insects

Acentria ephemerella is used for biological control of Eurasian Watermilfoil (*Myriophyllum spicatum*). Its population is best controlled by elimination of its host plants – which are predominantly Eurasian Watermilfoil but also may, to a lesser extent, include a variety of other native and nonindigenous plants (Cornell 2004). *Tanysphyrus lemnae* is an herbivore of duckweeds and other closely-related aquatic plants. It is likely also best controlled through control of its host plants.

4.6 Free-Living Crustaceans, including Waterfleas, Copepods, Mysids, and Gammarids

Microcrustaceans are easily transported overland by recreational boaters and on fishing gear. Species with large tail spines, such as *Bythotrephes* and *Cercopagis* are especially likely to be spread in this way, as are resting eggs that are adapted to promote attachment (as in the hooks on *Daphnia lumholtzi* ephippia). Fishing lines designed specifically to prevent the spread of waterfleas, such as the Flea Flicker brand, have been proven effective in significantly reducing fouling on lines, indicating their importance as a management tool (Jacobs and MacIsaac 2007). Cleaning all aquatic equipment with high-pressure water (>250 psi) or hot water (>50°C) after each use has proven effective for controlling spread via this pathway. *Echinogammarus ischnus* can tolerate a maximum water temperature between 31.0°C and 32.2°C and *Gammarus tigrinus* between 32.2°C and 34.2°C before irreversible physiological damage and mortality occur (Wijnhoven et al. 2003). *Hemimysis anomala* also exhibits mortality at temperatures below 0° C (Borcherding et al. 2006), making freezing of gear an alternative.

Electron beam irradiation has been used to control microorganisms in aquatic pathways, including invasive waterfleas and copepods (USACE 2012b). Electron beam irradiation is a non-selective control method which exposes water to low doses of radiation using gamma-sterilizers or electron accelerators, breaking down DNA in living organisms while leaving behind no by-products (USACE 2012b). Ultraviolet (UV) light can also effectively control microorganisms in water treatment facilities and narrow channels, where UV filters can be used to emit UV light into passing water, penetrating cell walls and rearranging DNA of microorganisms (USACE 2012b). Many ballast water treatment systems already available and/or under development use UV as a primary biocide, usually preceded by filtration (Lloyd's Register Marine 2014). Another possible non-selective pathway control is high water turbidity, which may decrease zooplankton (especially cladoceran) abundances due to the negative effects of suspended clay particles on filtering and assimilation rates (Suchy and Hann 2007).

Waterfleas are consumed by planktivorous fishes and predatory invertebrates; manipulation of predator populations (top-down control) has been used to control waterflea populations. Young-of-year Bloaters (*Coregonus hoyi*) have been shown to surface feed on *Eubosmina coregoni* in Lake Michigan, but their effectiveness as a control is unknown, especially because they may move to the benthos earlier to avoid competition from the invasive Alewife (*Alosa pseudoharengus*) (Crowder and Crawford 1984). *Bythotrephes* spp. are known to consume *E. coregoni* in Russia and the United States (Grigorovich et al. 1998), and *E. coregoni* populations declined significantly immediately following the invasion of the waterflea *Bythotrephes longimanus* in the Laurentian Great Lakes Michigan, Huron, and Erie, with direct predation by *B. longimanus* the most likely explanation (Barbiero and Tuchman 2004). *E. coregoni* levels in *B. longimanus*-invaded areas have remained at low levels, indicating a significant population-wide impact, but the long-term effectiveness of *B. longimanus* as a biological control method is unknown (Barbiero and Tuchman 2004). *B. longimanus* is consumed by Rainbow Smelt, Lake Herring, Lake Whitefish, Yellow Perch, White Perch, White Bass, Walleye, Alewife, Bloater Chub, Emerald Shiner, Spottail Shiner, Deepwater Sculpin, and Chinook Salmon in the Great Lakes (Bur et al. 1986, Makarewicz and Jones 1990, Branstrator and Lehman 1996). The defensive tailspine on *Bythotrephes longimanus* has been observed to increase in size throughout the summer in response to predation pressure (Straile and Halbach 2000). Consequently, larger fish are more likely to be successful predators (Branstrator and Lehman 1996). The opossum shrimp (*Mysis relicta*) has been observed eating *B. longimanus* in Ontario lakes, but the frequency of consumption appeared related to abundance of the invader and alternate prey (Nordin et al. 2008). Pothoven et al. (2007) found that adult large Alewives (*Alosa pseudoharengus*) (>100 mm) consume *Cercopagis pengoi* in Lake Michigan, but not significantly enough to control the species, concluding that the Alewife prefers *Bythotrephes longimanus* due to its larger size and conspicuousness. In contrast, a study of *C. pengoi* as a prey item in Lake Ontario found that at least 70% of Alewives larger than 70 mm contained *C. pengoi* spines (Bushnoe et al. 2003). The same study also found spines in Rainbow Smelt (*Osmerus mordax*) stomachs. Rainbow Smelt historically consume cladocerans in the Great Lakes, but prefer larger prey and may select *B. longimanus* over *C. pengoi* where both occur (Pothoven et al. 2009). Gorokhova et al. (2004) found that in the northern Baltic proper, Herring (*Clupea harengus* L.) and Sprat (*Sprattus sprattus* L.) are the dominant predators of *C. pengoi*, and a possible source of biological control through fisheries management, though it is possible that fully mature resting eggs may survive passage through fish digestive systems as has been observed with *B. longimanus* eggs in Yellow Perch (*Perca flavescens*). *B. longimanus* is also known to consume *C. pengoi*, but not as a main prey item (Cavaletto et al. 2010). *Daphnia lumholtzi* is preyed upon by a variety of zooplanktivorous fishes, including Inland Silversides, *Menidia beryllina*, in Lake Texoma, Bluegill (*Lepomis macrochirus*), White Bass (*Morone chrysops*), White Crappie (*Pomoxis annularis*), and Black Crappie (*Pomoxis nigromaculatus*) in Lake Chautauqua, Illinois (Lienesch and Gophen 2001, Lemke et al. 2003). The degree to which these fishes may be able to control *Daphnia lumholtzi* populations is not certain. The mysid

shrimps *Mysis mixta* and *Mysis relicta* consume *Eubosmina maritima* diapausing eggs (ephippia) selectively in the northern Baltic Sea, but only at a rate which can affect local abundances of *E. maritima* (Viitasalo and Viitasalo 2004). Research on *D. galeata galeata* is lacking, but many invertebrates are likely predators of *Daphnia* spp. where they occur in Europe and North America, including Great Lakes species such as the predacious phantom midge *Chaoborus flavicans*, the waterfleas *Leptodora* spp. and *B. longimanus*, (Lysebo 1995). Many small fish of species such as Yellow Perch (*Perca flavescens*), Three-spined Stickleback (*Gasterosteus aculeatus*), Alewife, Bluegill, and Ciscos have been documented consuming daphnids and other zooplankton in Canada and the United States (Mills and Forney 1983, Post and McQueen 1987, Hulsmann and Mehner 1997).

Free-living copepods are also preyed upon by fish and invertebrate predators and top-down control may be a useful tool. Treasurer (1992) found that Eurasian Perch, *Perca fluviatilis*, larvae selectively prey on *Cyclops strenuus abyssorum* in the Scottish lochs Kinord and Davan. The total zooplankton reduction observed was minimal, but Treasurer suggested that grazing by larvae is likely to impact copepod populations. Maes et al. (2005) found that juvenile Herring (*Clupea harengus*) and Sprat (*Sprattus sprattus*) exhibit top down control of *Eurytemora affinis* through predation pressure in the Scheldt estuary in Belgium. Yellow Perch (*Perca flavescens*) and Trout Perch (*Percopsis omiscomaycus*) are potential predators of *Megacyclops viridis* (Ogle et al. 1995). Hansen and Jeppesen (1992) found that a 50% reduction of planktivorous fish biomass (Roach, *Rutilus rutilus*, and Bream, *Abramis brama*) affected cyclopoid copepod population directly through reduction in fish predation pressure and changes in biological structure of Lake Væng, Denmark. *Megacyclops viridis* is also an important prey item for introduced Ruffe (*Gymnocephalus cernuus*), but the Ruffe feeds on a wide variety of benthic organisms, so its feasibility as a biocontrol is unknown (Ogle et al. 1995). *Nitokra hibernica* has been found in the stomach of Slimy Sculpin (*Cottus cognatus*) at Yankee Reef, Lake Huron, and it is known to be consumed by Rainbow Smelt (*Osmerus mordax*) in Lake Huron, but the significance of this predation on biological control is unknown (Lesko et al. 2003).

Benthic invertebrates including *Echinogammarus ischnus* are a major part of the native Yellow Perch (*Perca flavescens*) diet (Gonzalez and Burkart 2004). *Echinogammarus ischnus* has also become prey of the invasive Round Goby, *Neogobius melanostomus* (Gonzalez and Burkart 2004). The spread of dreissenid-covered substrate across the Great Lakes region has created an ideal habitat for *Echinogammarus ischnus*, where it is less susceptible to predation, indicating that efforts to control *Dreissena* spp. could also aid control of *Echinogammarus ischnus* (Gonzalez and Burkart 2004). A parasitic water mold (oomycete) detected in the upper St. Lawrence River is likely responsible for reducing *Echinogammarus ischnus* abundance despite favorable physical and chemical conditions (Kestrup et al. 2011). The oomycete also infects the native amphipod *Gammarus fasciatus*, but its effects are significantly less severe than in *Echinogammarus ischnus* (Kestrup et al. 2011). In a study of the effects of tide gate operation on hypoxic conditions in the Back River and Savannah River estuaries in Savannah, Georgia, it was

found that oxygen saturation levels below 30% are lethal to experimental populations of amphipods (Winn and Knott 1992). Electron beam irradiation can be used to control microorganisms in aquatic pathways, including *Echinogammarus ischnus* (USACE 2012b). Electron beam irradiation is a non-selective control method which exposes water to low doses of radiation using gamma-sterilizers or electron accelerators, breaking down DNA in living organisms while leaving behind no by-products (USACE 2012b). Ultraviolet (UV) light can also effectively control microorganisms including *E. ischnus* in water treatment facilities and narrow channels, where UV filters can be used to emit UV light into passing water, penetrating cell walls and rearranging DNA of microorganisms (USACE 2012b).

Various parasites have been shown to reduce host density and population survival in experimental *Daphnia* populations in Europe (Ebert 2005). However, it is unclear whether parasites regulate natural *Daphnia* populations, as all experiments have been completed under lab conditions.

Zooplankton biomass generally increases with increasing eutrophication, so reduction of excessive nutrient pollution causing abnormal eutrophication could help control invasive waterfleas. Gemza (1995) documented a shift from copepods to cladocerans as dominant zooplankton at increasingly eutrophic sites in Severn Sound, Lake Huron. Many of the non-native freshwater cladocerans that have invaded the Great Lakes are susceptible to salinities >3‰ (Nauwerck 1991). Santagata et al. (2008) found that ballast water exchange methods which flush freshwater organisms into euhaline seawater are effective against *Eubosmina maritima* at a minimum of 24 PSU (practical salinity units) for two hours in a laboratory simulation. However, *E. maritima* ephippia may remain in residual unpumpable sediment in ballast tanks, and Gray et al. (2005) found that exposing zooplankton ephippia to open ocean saline water of 32 ppt (parts per thousand) did not reduce egg abundances or consistently affect richness of invertebrates hatched from exposed eggs. *Hemimysis anomala* tolerates salinity of 0-19 ppt (parts per thousand) (Bij de Vaate et al. 2002). Ellis and MacIsaac (2009) documented 100% mortality for *H. anomala* after five hours in a simultaneous BWE treatment, in which salinity was gradually increased from 4-30 ppt, and 100% mortality after three hours in a sequential BWE treatment, in which species are immediately exposed to 30 ppt salinity.

A study of the effects of cadmium and zinc on Lake Michigan zooplankton found that *E. coregoni* was significantly reduced by separate and combined treatments, with negative effects primarily due to zinc (Marshall et al. 1981). A more recent study on the effects of copper sulfate (used to control algal biomass in eutrophic water bodies) and Carbaryl (used to control aquatic pests) on zooplankton found that levels of 50 µg/L Cu and 20 µg/L Carbaryl individually reduced *E. coregoni* biomass by >50% (Havens 1994).

Lindley et al. (1999) found that exposure to the organochlorine compounds pentachlorophenol (PCP) and 1, 2-dichlorobenzene (DCB), (both common industrial pollutants) accumulated in sediment significantly reduced hatching success and nauplii viability of *E. affinis* eggs. In a study

of the effect of salinity on toxicity of cadmium to Chesapeake Bay organisms, Hall et al. (1995) found that *E. affinis* is very sensitive to cadmium compared to other estuarine aquatic biota. The negative effects of the insecticide diflubenzuron on *E. affinis* nauplii were documented by Savitz and Wright. The insecticide is approved for use against the Gypsy Moth (*Lymantria dispar*) and other insect pests by the United States Environmental Protection Agency, and enters *E. affinis* habitat primarily through runoff (Savitz and Wright 1994). Diflubenzuron specifically targets the arthropod molting process, so the most explicit effects are expected in sub-adult crustaceans.

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of crustaceans.

4.7 Free-Living Worms including Annelids and Platyhelminthes

Little research has been pursued concerning control of aquatic annelids. The effects of industrial toxicants on Tubificidae species and explorations of their value as an indicator of environmental quality have been explored, but chemicals and heavy metals are not viable methods of control because of unknown and adverse effects on the surrounding environment (Das and Das 2005, Saha et al. 2006). However, there has been investigation into the control of *Branchiura sowerbyi* as a host of haemorrhagic thelohanellosis, which negatively impacts fish in aquaculture (Liyanage et al. 2003).

Brown Trout, *Salmo trutta* L., has been shown to prey on oligochaetes; its removal from an experimental environment led to rapid multiplication of benthic fauna (Wahab et al. 1989). However, Brown Trout is itself an invasive species in the Great Lakes region and across nearly all of the United States.

Research on benthic macroinvertebrate communities in southwestern Lake Ontario before and after the invasion of *Dreissena polymorpha* (Zebra Mussels) and *Dreissena bugensis* (Quagga Mussels) suggests that the presence of *Dreissena* helps to improve benthic habitat, facilitating increases in macroinvertebrates, including the tubificids *Potamothrix vej dovskyi* and *Spirosperma ferox* (Stewart and Haynes 1994). This indicates that control of invasive Quagga and Zebra Mussels could facilitate improved control of benthic macroinvertebrates such as the tubificids.

Researchers found that *Branchiura sowerbyi*, which is a vector in transmission of *Thelohanellus hovorkai* (myxozoa) to fish, prefers muddy substrate, while other benthic oligochaetes that are not susceptible to myxozoa prefer sandy substrate, and suggested that replacing bottom substrate from mud to sand would lead to a shift in oligochaete communities from *Branchiura sowerbyi* to non-susceptible oligochaetes such as *Limnodrilus socialus*, therefore reducing disease in aquaculture fauna (Liyanage et al. 2003).

Potamothrix bedoti has been shown to be more likely to occur in substrate with a high clay and silt content, (Sauter and Gude 1996) while *Potamothrix moldaviensis* has been shown to be more likely to occur in sandy substrate with a clay and silt content of less than 10%. This indicates that substrate type is a possible physical control method to be further explored.

Ripistes parasita has been found to occur in greater numbers where water quality is impaired by industrial pollution, therefore greater measures to control pollutants such as heavy metals and particulate matter might help control this oligochaete (Simpson and Abele 1984). Furthermore, declines in Oligochaeta in southern Lake Michigan were recorded between 1980 and 1993 in correlation with reductions in phosphorus loads (Nalepa et al. 1998), suggesting that reduction of excess nutrients would help to reduce oligochaete populations.

Only one free-living nonindigenous platyhelminth is found in the Great Lakes, *Dugesia polychroa*. No research is available supporting control of this species.

4.8 Bryozoans, Hydrozoans, and Testate Amoebae

Lophopodella carteri colonies grow on solid substrata (Lauer et al. 1999), therefore, physical removal methods such as scraping may be viable for localized areas. Chemical biocides have been used as anti-fouling agents to remove sessile macroinvertebrates from shipping equipment and industrial intakes, but none have been approved for use on bryozoans as of yet (USACE 2012b). The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of bryozoans. Freshwater Bryozoans are generally sensitive to heavy metals, particularly copper (Bushnell 1974). Pardue and Wood (1980) determined baseline toxicity of four heavy metals to three species of phylactolaemate bryozoa (*L. carteri*, *Pectinatella magnifica* Leidy, and *Plumatella emarginata* Oka), observing greatest sensitivity to cadmium, followed by copper, chromium, and zinc. It should be noted that the toxicity of these metals were not tested as control measures, but as a demonstration of the usefulness of some bryozoans as biomonitors of water quality. However, comparison of the 96-hr LC50 data to toxicity data from other studies indicates that the bryozoa are more sensitive to the tested metals than many other invertebrates and fish, indicating potential as chemical controls with further research (Pardue and Wood 1980). It should be noted that at least one invasive Marine Bryozoan (*Bugula neritina*) has demonstrated heavy metal-resistant genotypes, suggesting that metal-intensive antifouling agents may have diminished effectiveness on their populations (Piola and Johnston 2006).

Control of *Cordylophora caspia* will likely focus on its potential role as a biofouling agent. *Cordylophora* spp. has been documented colonizing the inner walls of power plants in Europe and the United States (Folino 2000), primarily causing blockages in nozzles and tailpipes of rapid gravity filter beds (RGFs) (Mant et al. 2011). The menont life-stage of *C. caspia* often found in hydroelectric intakes, is both drought and temperature resistant which may prove an

obstacle to control (Gutierre and Gutierre 2012). Thermal treatments greater than 37°C have been proven effective in eradicating colonies of *Cordylophora* spp. sampled from the walls of power plant intakes (Folino 2000), but are not efficient in water treatment facilities where there is no residual heat energy available (Mant et al. 2011). Gutierre and Gutierre (2012) found that *Cordylophora caspia* is completely eradicated at pH levels of 4.0 and 10.0, with increasing survival rates in between, and suggested maintaining pH levels at 10.0 for six hours or more by injection of NaOH to reduce and eliminate colonies. Chlorine treatments negatively affect *Cordylophora* growth, but treatments as high as 5 mg/L for periods of 105 minutes have been unsuccessful in completely eradicating colonies (Mant et al. 2011). Furthermore, chlorine use is highly regulated at water treatment facilities where *Cordylophora* most frequently cause problems. Hydroids are sensitive to vanadium leeching from slag stones used in riverbank reinforcement, and are sensitive to heavy metals in general, especially mercury, copper, cadmium, and arsenic, though it is unlikely that these will be useful in control (Ringelband and Karbe 1996).

Craspedacusta sowerbyi has spread across temperate climates for more than a century, but despite experimental observation of its possible contribution to trophic cascade effects (Jankowski et al. 2005), and studies on predation habits (Dendy 1978, Dodson and Cooper 1983, Spadinger and Maier 1999), little research on control is available.

Three nonindigenous members of Genus *Psammonobiotus* have been documented as established in the Great Lakes. No research is available supporting control of these species.

4.9 Parasites and Diseases

Most control research conducted for nonindigenous fish parasite and disease species has been in the context of aquaculture and recreational fisheries pond management. Minimizing fish stress can reduce the risk of disease outbreak (FTS 2012). No treatments exist to control these diseases and parasites in open systems. Establishment of quarantines may prevent transmission and be useful in controlling spread.

4.9.1 Parasitic Platyhelminthes

Implementation of Eurasian Ruffe management may potentially decrease *Dactylogyrus* spp. prevalence due to host specificity. Lampricide TFM may effectively eliminate up to 97% of Ruffe, potential carriers of *Dactylogyrus amphibothrium*, with minimal non-target mortality (Crosier et al. 2012). However, Ruffe management is considered by some (e.g., Ogle 1998) to be difficult and impractical given that the species has developed several adaptations to compensate for high mortality rates (Lind 1977) and populations rebound quickly (Lelek 1987). *Bothriocephalus acheilognathi* populations in aquaculture and ponds can be controlled by managing the intermediate host (i.e., copepods) population densities. Effective ectoparasiticides include Neguvon®, Masoten®, Dipterex®, Bromex®, and Naled® (Paperna 1996).

Bath treatments are effective control methods for *Bothriocephalus acheilognathi* infections. Baths should contain Droncit® (praziquantel), isopropyl alcohol, and water yielding a final mixture concentration ≥ 0.67 ppm praziquantel. Fish densities during treatment should be no greater than 60 mg fish/L and exposure should last 24 hours. After 24 hours, the treatment should be drained, worm parts discarded, and clean water added. Fish should then be transferred to a decontaminated container (Mitchell and Darwish 2009).

Dactylogyrus-specific treatments are unknown. However, multiple chemicals are effective at treating monogenean fluke infections in aquaculture systems. Effective benzimidazoles include levamisole (Buchmann 1997) and praziquantel, which has high efficacy against *Dactylogyrus* spp. (Schmahl and Mehlhorn 1985, Buchmann 1997). Effective bath treatments include formaldehyde (30-100 ppm), sodium chloride, copper sulphate, hydrogen peroxide, sodium percarbonate (Buchmann and Kristensen 2003), formalin (25 mg/L for prolonged exposure or 150-250 mg/L for 30 minutes), and potassium permanganate (2 mg/L for prolonged exposure or 10 mg/L for 30 minutes) (Reed et al. 1996). Effective organophosphate bath treatments include metrifonate (0.25-0.5 ppm) and dichlorvos (0.25-0.5 ppm) (Sarig et al. 1965). Pond infestations can be controlled with formalin (30 mg/L) or trichlorfon (Lepidex®; 0.5 mg/L) (Reed et al. 1996). However, monogenean eggs display chemical resilience and therefore the above chemical treatments are ineffective at destroying eggs (Reed et al. 1996, Rowland et al. 2007). Chemical toxicity varies considerably between monogeneans and fish species. Toxicology and tolerance tests are suggested prior to using anthelmintics (“dewormers”). Managers are encouraged to consider specific host drug tolerance, temperature, salinity, organic material content, and drug retention time prior to treatment (Buchmann and Bresciani 2006). Freshwater fish species can also be dipped in saltwater to minimize external parasite numbers prior to stocking (Reed et al. 1996).

Bothriocephalus acheilognathi infections can be treated with chemically enhanced feed. Drugs should be mixed in oil and sprayed on feed at a rate of 1 L/70 kg dry weight. Effective chemicals and doses include dibutyltin oxide or dibutyltin dilurate (250 mg/kg fish) fed over three days (Mitchell and Hoffman 1980), Yomesan® (500 g/500 kg dry pellets) fed at 1.5% of body weight 2–3 times weekly, and Yomesan® (28 g/40 kg) fed for three days (Korting 1974, Mitchell and Hoffman 1980, Brandt et al. 1981).

No effective treatments have been identified for *Ichthyocotylurus pileatus*, *Neascus brevicaudatus*, *Scolex pleuronectis*, or *Timoniella* sp.

4.9.2 Parasitic copepods

Electron beam irradiation can be used to control microorganisms in aquatic pathways, including *Neoergasilus japonicus* (USACE 2012b). Electron beam irradiation is a non-selective control method which exposes water to low doses of radiation using gamma-sterilizers or electron accelerators, breaking down DNA in living organisms while leaving behind no by-products

(USACE 2012b). Ultraviolet (UV) light can also effectively control microorganisms including *N. japonicus* in water treatment facilities and narrow channels, where UV filters can be used to emit UV light into passing water, penetrating cell walls and rearranging DNA of microorganisms (USACE 2012b).

Gault et al. (2002) documented the effectiveness of “egg-laying boards” in control of the confamilial *Argulus foliacus* in a Rainbow Trout (*Oncorhynchus mykiss*) fishery. Lightweight, corrugated polypropylene boards were anchored and submerged within six mm of the water surface in a horizontal plane or floated vertically at varying depths to provide ideal egg-depositing surfaces for *A. foliacus*. Replacement of the boards at two week intervals and constant deployment throughout the *A. foliacus* breeding season significantly reduced the prevalence and intensity of parasite infection in trout, but the boards do not stop all egg hatching due to clutches laid on other pond surfaces. The most effective method of removing *Argulus* spp. from recreational fisheries and aquaculture ponds is complete drainage and removal/restocking of fish, possibly combined with a treatment of lime, but this is not economically or physically possible in many cases (Gault et al. 2002, Taylor et al. 2006).

The organophosphate compound Dipterex (organophosphate trichlorphon) has been used to control argulids in ponds and aquaculture (Tonguthai 1997). Argulid adults and larvae fall off parasitized fish and die within 12-24 hours after treatments of 0.2-0.3 ppm, sprinkled or sprayed uniformly on the pond surface (Tonguthai 1997). However, argulid resting stages are not affected by Dipterex—multiple treatments must be applied over the hatching period for effective reduction. In a study on chemotherapeutic control of ectoparasites in aquaculture, Singhal et al. (1986) found gammexane (1-6-hexachlorocyclohexane) to be most effective in removing *Argulus indicus* from host fish Indian Carp (*Catla catla*), Rohu (*Labeo rohita*), Mrigal Carp (*Cirrhinus mrigala*), Grass Carp (*Ctenopharyngodon idella*), and Silver Carp (*Hypophthalmichthys molitrix*) after a treatment was sprayed over the pond surface at 0.2 mg L⁻¹ three times at weekly intervals. Placing *Catla catla*, *L. rohita*, and *Ctenopharyngodon idella* in 30 mg L⁻¹ sodium chloride solution for 2-5 minutes was effective in removing *A. indicus*, and 1-2 minutes sufficed for *H. molitrix* and *Cirrhinus mrigala* (Singhal et al. 1986). Placement of fish in 0.5 mg L⁻¹ potassium permanganate achieved 60% reduction of parasites within five minutes. For severe infestations, 0.2 g L⁻¹ of lime was used to treat drained, dried ponds, after which they were restocked. Combinations of organophosphate pesticides and salt are reported to have controlled *Argulus japonicus* after four treatments at one week intervals (Avenant-Oldewage 2001). As with Dipterex, it should be assumed that the above treatments are ineffective against argulid resting stages, as the effects have only been studied on hatched copepods.

4.9.3 Protozoan Parasites

Effective physical control methods for *Heterosporis* include complete desiccation of holding tanks and equipment for 24 hours, freezing at -20°C for 24 hours, and culling (Sutherland et al. 2006, GLFHC 2012b). Immersion of gear in a 2200 ppm bleach (0.7 L bleach per 20 L of water)

solution for five minutes will destroy the parasite (IDNR 2005, Sutherland et al. 2006, GLFHC 2012b).

Exposing myxospores (*Myxobolus cerebralis*) to 90°C water for 10 minutes is effective at destroying the spores (Hoffman and Markiw 1977). Electrical charges of 1-3 kV pulsed 1-25 times at 99 µsec/pulse is effective at killing large numbers of TAM spores (Wagner 2002). Experiments by Hoffman (1974) have demonstrated that filtration is not an effective method for removing TAM spores from water – due to the small spores size, the filter needed to remove them slows flow to rates unacceptable for most applications. Hatchery intake water treated with chlorine (0.5 ppm) administered at two-hour intervals once a week can reduce infection rates in Rainbow Trout by 63-73% without causing harm to the fish (Markiw 1992). Supply water treated with calcium cyanide (488 g/m²) mixed with chlorine gas (300 ppm) can be very effective at destroying *M. cerebralis* spores (Hoffman and Dunbar 1961). Water treated with chlorine (130-260 ppm) for 10 minutes may kill 100% of TAM spores present (Wagner 2002), and treating with chlorine (5,000 ppm) for 10 minutes is sufficient enough to destroy both triactinomyxon and myxospore (E. MacConnell, pers. comm. in Wagner 2002). Treating fry with chlorine (10 ppm) for 30 minutes may prevent whirling disease infection (Hoffman and O’Grodnick 1977). At the Roaring Judy Hatchery, a project is underway to install an ultraviolet system that kills *M. cerebralis* spores (CDW 2011). Treating water with 2537Å UV at doses of 35mWs/cm² can be 86-100% effective at preventing whirling disease in Rainbow Trout fry (Hoffman 1974) and administering 1,300 mWs/cm² of UV under a static collimated beam, can inactivate 100% of the TAM spores present (Hedrick et al. 2000). Earthen pond substrate treated with quicklime (CaO) at concentrations greater than 380 g/m² for two weeks prior to introducing fish can prevent whirling disease infection by destroying *M. cerebralis* spores (Hoffman and Hoffman 1972).

Myxobolus cerebralis is frequently controlled by managing the intermediate host, *Tubifex tubifex*. Managers have observed that using concrete in aquaculture facilities can reduce the abundance of *T. tubifex* and thus limit the ability of *M. cerebralis* to reproduce (Mills et al. 1993, Ricciardi 2001). Maintaining water quality, reducing favorable habitat by preventing sediment accumulation in aquaculture (Crosier et al. 2012), and desiccating holding tanks, equipment, and intake pipes may help control *T. tubifex* (Kaster and Bushnell 1981). Lampricide TFM (3-triflouromethyl-4-nitrophenol), administered at (4.2-14.0 mg/L) doses, is effective at destroying *T. tubifex* (Lieffers 1990). *T. tubifex* can also be treated in 30°C water for four days, causing triactinomyxon (TAM) spore production to stop, thus preventing the next stage of the parasites life cycle (El-Matbouli et al. 1999). *T. tubifex* ability to support *M. cerebralis*’ triactinomyxon (TAM) spore production may be due to genetic differences among *T. tubifex* populations. This variability may be an important factor in determining infection rates among fish (Baxa et al. 2006) and therefore might support certain management practices (Stromberg 2006). There is evidence that electricity (1,000 s exposure to low-level DC voltage for 48 hrs) can destroy *T. tubifex* in aquaculture (R. Ingraham and T. Claxton, pers. comm. in Wagner 2002).

It has been proposed that selective processes are yielding a surviving population of fish that is more resistant to *Myxobolus cerebralis* infection on the Madison River, Montana (Vincent 2006). The implications of this for management are still unclear. However, research is continuing to evaluate the possibility of a developing resistance within salmonid populations (Stromberg 2006).

It has been demonstrated that feeding Rainbow Trout with pellets containing (0.1%) Fumagillin is effective at reducing whirling disease infection. Two groups of Rainbow Trout were administered pellets from days 14-64 and 30-160 post infection. Approximately 10-20% of the medicated fish harbored spores, whereas 73-100% of non-medicated fish harbored spores (El-Matbouli and Hoffman 1991).

No effective treatments have been identified for *Acineta nitocrae*, *Glugea hertwigi*, *Sphaeromyxa sevastopoli*, or *Trypanosoma acerinae*.

4.9.4 Bacteria

Antimicrobial agents used to treat *A. salmonicida* infections include thiophenicol, furazolidone (Herman 1968), oxytetracycline (Herman 1968, Heo and Seo 1996, Wiklund and Dalsgaard 1998), sulphamerazine, tetracycline, and a combination of trimethoprim (Heo and Seo 1996, Wiklund and Dalsgaard 1998) and sulphonamide (McCarthy and Roberts 1980). Others include flumequine (Michel et al. 1980), oxolinic acid (Hastings and McKay 1987, Heo and Seo 1996, Wiklund and Dalsgaard 1998), florfenicol (Inglis et al. 1991b), amoxycillin (Inglis et al. 1992), enrofloxacin (Stoffregen et al. 1993), chloramphenicol, neomycin, nitrofurantoin, and ciprofloxacin (Heo and Seo 1996, Wiklund and Dalsgaard 1998). Feed containing terramycin and romet are effective in treating *A. salmonicida* (MIDNR 2012). However, an increase in antimicrobial resistance was recognized in the United States beginning in 1967 (Wood 1967). Antimicrobial resistance by *A. salmonicida* has been discovered with the following agents: sulphonamides (Herman 1968), oxytetracycline (Tsoumas et al. 1989, Inglis et al. 1991a, Grant and Laidler 1993), a combination of sulphonamide and trimethoprim (Tsoumas et al. 1989, Grant and Laidler 1993), oxolinic acid (Hastings and McKay 1987, Tsoumas et al. 1989, Inglis et al. 1991a, Höie 1992, Grant and Laidler 1993, Oppegaard and Sörum 1994), flumequin (Höie 1992), and amoxycillin (Grant and Laidler 1993). The United States FDA approved broad-spectrum, in-feed antibiotic AQUAFLO® is now available to control mortality in finfish due to furunculosis (MAA 2012). Antimicrobial agents used to treat *Renibacterium salmoninarum* include nitrofurans, bacitracin, chlortetracycline, oxytetracycline, novobiocin (Wolf and Dunbar 1959), cephalosporins, gentamicin, clindamycin, lincomycin, oleandomycin, kidasamycin, spiramycin, penicillin (Austin 1985), cefazolin, tiamulin (Bandín et al. 1991), cephradine, rifampicin, (Brown et al. 1990), tetracycline (Austin 1985, Bandín et al. 1991), chloramphenicol (Wolf and Dunbar 1959, Austin 1985), and erythromycin (Wolf and Dunbar 1959, Austin 1985, Bandín et al. 1991, Lee and Evelyn 1994). All commonly used aquaculture disinfectants are considered effective against *Piscirickettsia* spp. (Fryer et al. 1990, 1992, Corbeil and Crane

2009). Antimicrobial agents used to treat *P. salmonis* include oxolinic acid and flumequin (Guardabassi and Courvalin 2006, Todar 2008, Corbeil and Crane 2009).

Disinfecting eggs with 100 mg/L of iodine for 15 minutes may not eliminate vertical transmission of Bacterial Kidney Disease (BKD) but can reduce the severity of the disease (Bullock et al. 1978). Treating eggs with iodine at 250 or 500 mg/L for 15-120 minutes is effective at eliminating *Renibacterium salmoninarum*. However, after treatment, variable numbers of *R. salmoninarum* cells have survived. This is due to cells within the cell aggregates never coming in contact with the iodine (Evelyn et al. 1984, 1986). *salmoninarum* is inactivated by free chlorine (≤ 0.05 mg/L), which can be used as a disinfectant and to treat intake and effluent (Pascho et al. 1995).

The U.S. FDA approved vaccine Furogen® (Aqua Health, LTD) administered to brood stock prior to spawning has proven to be very effective at reducing *A. salmonicida* prevalence (GLFHC 2006). Very good to excellent results controlling and preventing out-breaks of furunculosis have also been obtained using an autogenous vaccine, produced by Microtechnologies; this treatment was accompanied by an immune-enhancing feed administered for three weeks prior and three weeks post vaccination, as well as stock thinning to reduce overall stress (GLFHC 2006). Vaccines derived from inactivated *Piscirickettsia* cf. *salmonis* bacteria are considered ineffective against the bacterium. However, vaccines being developed using recombinant DNA have promise in combating *P. salmonis* infections (Corbeil and Crane 2009). Further research is required to understand the practical application of this therapy.

Administration of bacteriophage to infected fish may help control outbreaks. In a study that administered the bacteriophage HER 110 to *A. salmonicida* HER 1107 infected Brook Trout (*Salvelinus fontinalis*), *A. salmonicida* populations declined by six log units (base 10) in 3 d. Further tests within fish populations are necessary to better understand the implementations of this therapy (Imbeault et al. 2006).

While supplements of lactic acid bacteria (*Carnobacterium* spp.) given with fish feed do not protect against *Aeromonas salmonicida* infection (Gildberg et al. 1995), such probiotic supplementation (also *A. hydrophila* and *Vibrio fluvialis*) can decrease mortality in Atlantic salmon (*Salmo salar*) and rainbow trout (*Oncorhynchus mykiss*) infected with *A. salmonicida* (Irianto and Austin 2002). *Carnobacterium* strain K1 colonizes the intestinal tract of rainbow trout and inhibits *A. salmonicida* growth (Jöborn et al. 1997). Short-term bathing of presmolt Atlantic Salmon infected with furunculosis with siderophore-producing *Pseudomonas fluorescens* is another successful biological control (Gram et al. 1999, Smith and Davey 1993). Bath treatments with *V. alginolyticus* (Austin et al. 1995) can also lead to a reduction in mortality (Verschuere et al. 2000). Renogen® can reduce mortality rates in *Piscirickettsia* cf. *salmonis* infected Pacific salmon (Torenzo et al. 2005).

Erythromycin-enhanced feed administered at 100 mg/kg/day for 21 days (Wolf & Dunbar 1959) or for 10 days (Austin 1985) is believed to be the most effective treatment for bacterial kidney disease (Hirvela-koski 2005). Erythromycin phosphate is an effective chemoprophylaxis in pre-spawning adult brood fish. Subcutaneous injections of 11 mg/kg (Hirvela-koski 2005) or 20 mg/kg (Gudmundsdóttir et al. 2000, Pascho et al. 1991) administered to fish entering trapping facilities and at 21-30 day intervals thereafter (Hirvela-Koski 2005) has reduced mortality in re-spawning Pacific salmon by 10-50% (Groman and Klonz 1983). However, this treatment does not effectively destroy the pathogen from all eggs (Brown et al. 1990). A study by Lee and Evelyn (1994) showed female Coho Salmon treated with 20 mg/kg erythromycin prior to spawning yielded no vertical transmission of BKD. Treatment success relies on careful timing of the injections in adult salmonids before spawning (Elliott et al. 1989).

Culling is effective at preventing horizontal transmission of *Piscirickettsia* cf. *salmonis* (Torenzo et al. 2005). Culling infected stock has reduced the prevalence of *Renibacterium salmoninarum* in aquaculture (Gudmundsdóttir et al. 2000, Maule et al. 1996, Pascho et al. 1991).

4.9.5 Viruses

Establishment of quarantines, culling, and stock density reduction during the winter and spring are beneficial management practices to prevent the spread of viruses (CFSPH 2007). VHS should be reported to Area Veterinarians in Charge (AVIC) or state veterinarians immediately upon diagnosis or recognition of the disease. Fish health surveillance programs and fallowing are also useful methods of control (CFSPH 2007). The U.S. Fish and Wildlife Service recommend implementation of the International Hazard Analysis and Critical Control Point (HACCP) planning standard to prevent the spread of VHS (Bakal 2012).

Rhabdovirus carpio is inactivated by UV irradiation (254 nm), gamma irradiation (103 krad), heating to 60°C (140°F) for 30 minutes, and exposure to pH 12 for 10 minutes, or pH 3 for 3 hours (CFSPH 2007, World Organization for Animal Health 2009). Exposure to VHS can be prevented through use of spring water, specific pathogen free (SPF) stock, and separate cultivation of salmonids and flatfish (CFSPH 2007). Multiple means of VHS control are available to fish hatchery managers, including treatment of water with UV light subtype C (280-200 nm wavelength) irradiation and heat (>15°C) (McAllister 1990), exposure to pH levels lower than 2.5 or higher than 12.2, desiccation of tanks and equipment (CFSPH 2007), minimization of stressors, cessation of water flow to adjacent waterways, and establishment of quarantines (Warren 1983, CFSPH 2007).

Disinfection of live wells and other equipment potentially contaminated with Largemouth Bass Virus (LMBV) or VHS can be accomplished with a 10% household bleach/water solution (i.e., 100 ml of household bleach to 900 mL of water). Waste water should be discarded away from any water body. The VHS virus is sensitive to ether, chloroform, glycerol, formalin, iodophor, sodium hydroxide, and sodium hypochlorite, which can be used as disinfectants (McAllister 1990, CFSPH 2007). *Rhabdovirus carpio* is susceptible to oxidizing agents like sodium dodecyl sulphate, non-ionic detergents, and lipid solvents. The virus is inactivated by formalin, chlorine, iodine, NaOH, benzalkonium chloride, alkyltoluene, chlorhexidine gluconate, and cresol (Ahne

and Held 1980, Ahne 1982, Fijan 1999, CFSPH 2007, Kiryu et al. 2007). Methisoprinol may be useful by inhibiting replication of Spring Viremia of Carp Virus (SVCV) in vitro; but further testing under culture conditions is necessary (Siwicki et al. 2003).

Single-stranded and double-stranded RNA injections can provide *Rhabdovirus carpio* protection for up to three weeks (Masychewa et al. 1995, Alikan et al. 1996). No effective anti-viral agents or commercial vaccines exist (CFSPH 2007) for VHS.

5. Discussion

Only a small fraction (28%) of the more than 180 nonindigenous species which have become established in the Great Lakes are regulated by name primarily as invasives under federal or state law; just 26 plants, 10 fishes, 5 fish pathogens, 6 mollusks, 3 waterfleas, and 1 mysid within this subset, laws are a patchwork with only four diseases plus Zebra Mussels regulated by all states.

In addition to the federal regulations applicable to the noxious weed *Sparganium glomeratum*, the Great Lakes states regulate 26 species of nonindigenous aquatic plants that are established in the basin. Seventy-three percent of the listed plants have documented moderate to high environmental and or socioeconomic impacts. An overlapping 35% have documented benefits. No individual plant species is regulated in all eight Great Lakes states. Some species (e.g., Black Alder) restricted in one state are recommended in another.

Many nonindigenous fishes are managed primarily as beneficial species (commercial and sport fisheries or prey fishes supporting those fisheries). Just 10 species of nonindigenous fish are regulated primarily as invasives.

Five species of fish pathogens, including two bacteria (Bacterial Kidney Disease (BKD) and furunculosis), a myxosporean (whirling disease), and two viruses (VHS and LMBV) are explicitly targeted by regulation by the Great Lakes states. Four of these species (all but furunculosis) are considered to have high environmental impact to the Great Lakes. Furunculosis is targeted primarily because it is considered a threat to hatchery operations for rearing salmonids.

Six species of mollusks are state-regulated. Of these, only Zebra Mussels are regulated in all eight Great Lakes states, though Quagga Mussels are listed in federal regulation so are also effectively regulated in all eight states as well. This list includes a mix of species with high (Dreissenid mussels), moderate (Asian Clam and New Zealand Mudsail) and unknown (mystery snails) impacts.

Three species of established nonindigenous waterfleas as well as one mysid are regulated by name by select Great Lakes states. These include two high impact raptorial waterfleas (*Bythotrephes* and *Cercopagis*) and two relatively recent invaders whose impact we are as yet unable to assess (*Daphni lumholtzi* and *Hemimysis anomala*).

Unregulated species are not necessarily innocuous. Our previous assessments identified only 35 of the unregulated species (30%) as being low impact for all three assessments (environmental, socioeconomic and beneficial impacts) and an additional 10 species (9%) as being beneficial (*Ulva flexuosa*, *Acentria epemerella*, *Lupinus polyphyllus*, *Mentha gracilis*, *Mentha spicata*, *Polygonum persicaria*, *Puccinellia distans*, *Salix alba*, *Salix fragilis*, and *Salix purpurea*) (Sturtevant et al. 2014). Most of the unregulated species (57%) are ‘understudied’ with little to no information on their impacts available in the literature. However, this list includes five species (4%) with known high impact: (*Nitellopsis obtusa*, *Bithynia tentaculata*, *Ichthyocotylurus pileatus*, *Polygonum persicaria* and *Heterosporis*) and 12 species (10%) with moderate impacts (*Actinocyclus normanii* fl. *Subsalsa*, *Cylindrospermopsis raciborskii*, *Stephanodiscus binderanus*, *Ulva flexuosa*, *Ulva intestinalis*, *Carex acutiformix*, *Juncus compressus*, *Juncus gerardii*, *Juncus inflexus*, *Salix fragilis*, *Echinogammarus ischnus*, and *Piscirickettsia cf. salmonis*).

A growing body of literature provides a toolbox for control of nonindigenous species. However, most control efforts are successful at eradication only when the population is confined (e.g., aquaculture or other facilities, small ponds, limited geographic areas, etc). Control options suitable for established populations in open waters are extremely limited. Most control efforts focus on (1) controlling populations in locations where they directly impact human activities (e.g., aquaculture facilities and water intakes), (2) reducing populations to a level that minimizes impact to human socioeconomic endeavors (e.g., limiting impact to fisheries, agriculture, beach use, etc), (3) eradicating small, new infestations in confined geographic areas (e.g., a new infestation of *Iris pseudacorus* at a small pond), or (4) controlling spread along a specific pathway (e.g., ballast water treatment; using rotenone to poison fish in a canal).

Control options for many species remain extremely limited, but new technologies are emerging, and the toolbox is growing. Options for control of established nonindigenous algae in open waters are limited. Only the largest two species (*Ulva prolifera* and *Nitellopsis obtusa*) can be effectively harvested mechanically, even then, they are often the first to recolonize (Pullman and Crawford 2010). Some species, particularly *Cylindrospermopsis raciborskii*, are associated with stratified water columns and mechanical destratification (mixing) may help prevent bloom development (Antenucci et al. 2005). Although in some cases, control options are extremely limited (e.g., to hand pulling) and large, established infestations may be difficult to control. Control options are at least available for all the established nonindigenous plants – with a broad array of herbicides and physical controls, as well as biological controls for some species, readily available. An array of options is also available for control of fishes – from harvesting and trapping to physical barriers, chemical piscicides, food web manipulations, and emerging genetic control technologies. Nonindigenous aquatic insects are currently controlled primarily via control of their host plants – a strategy also employed for a number of smaller invertebrates. Biocides exist for many additional species, though application in open systems remains unfeasible for most.

6. Literature Cited

- Abdel-Fattah, S. 2011. Aquatic invasive species early detection and rapid response: assessment of chemical response tools. International Joint Commission, Great Lakes Regional Office.
- Agami, M., and Y. Waisel. 1986. The role of mallard ducks (*Anas platyrhynchos*) in distribution and germination of seeds of the submersed hydrophyte *Najas marina*. *Oecologia* 68: 473-475.
- Agami, M., and Y. Waisel. 1988. The role of fish in distribution and germination of seeds of the submerged macrophytes *Najas marina* L. and *Ruppia maritima* L. *Oecologia* 76(1): 83-88.
- Ahmadi, M.S., L.C. Haderlie, and G.A. Wicks. 1980. Effect of growth stage and water stress on barnyardgrass (*Echinochloa crus-galli*) control and glyphosate absorption and translocation. *Weed Science* 28(3): 277-282.
- Adamus, P., T.J. Danielson, and A. Gonyaw. 2001. Indicators for monitoring biological integrity of inland, freshwater wetlands: a survey of North American technical literature (1990-2000). United States Environmental Protection Agency, Office of Water and Office of Wetlands, Oceans, and Watersheds. Washington, D.C.
- Ahne, W. 1982. Vergleichende Untersuchungen über die Stabilität von vier fischpathogenen Viren (VHSV, PFR, SVCV, IPNV). *Journal of Veterinary Medicine Series B* 29: 457-476.
- Ahne, W., and C. Held. 1980. Untersuchungen über die viruzide Wirkung von Actomar® K30 auf fischpathogene Viren. *Tierärztlich Umschau* 35: 308-318.
- Aida, M., K. Itoh, H. Ikeda, N. Harada, Y. Ishi, and K. Usui. 2004. Susceptibilities of some aquatic ferns to paddy herbicide bensulfuron methyl. *Weed Biology and Management* 4: 127-135.
- Aliken, Y.S., I.S. Shchelkunovis, T.I. Shchelkunovis, O.A. Kupinskaya, V.I. Masysheva, V.P. Klimenko, and V.A. Fadina. 1996. Prophylactic treatment of viral diseases in fish using native RNA linked to soluble and corpuscular carriers. *Journal of Fish Biology* 49: 195-205.
- Alminaite, A., D. Valinunas, and R. Jomantiene. 2002. First report of a Group 16SrI, Subgroup B, phytoplasma in diseased *Epilobium hirsutum* in the region of Tallin, Estonia. *Plant Disease* 86(10): 1177.
- Altop, E.K., and H. Mennan. 2011. Genetic and morphologic diversity of *Echinochloa crus-galli* populations from different origins. *Phytoparasitica* 39: 93-102.
- Amphlett, J., and C. Rea. 1909. The botany of Worcestershire. Cornish Brothers Ltd., Birmingham.

- Anderson, R.C. 1995. Nudibranchs: butterflies of the sea. *International Zoo Yearbook*. 43(1):65-70.
- Annen, C.A. 2007. Impact assessment: *Conium maculatum*. Wisconsin Department of Natural Resources.
- Antenucci, J.P., A. Ghadouani, M.A. Burford, and J.R. Romero. 2005. The long-term effect of artificial destratification on phytoplankton species composition in a subtropical reservoir. *Freshwater Biology* 50(6): 1081-1093.
- Aquatic Ecosystem Restoration Foundation (AERF). 2012. Aquatic herbicide use guide: herbicides. Available <http://www.aquatics.org/herbicides.htm>. Accessed 19 September 2012.
- Aquatic Ecosystem Restoration Foundation (AERF). 2013. Aquatic herbicides/agents. Available <http://www.aquatics.org/herbicides.html>. Accessed 16 April 2013.
- Arlinghaus, R., and T. Mehner. 2004. Management and ecological note: testing the reliability and construct validity of a simple inexpensive procedure to measure the use of recreational fishing. *Fisheries Management and Ecology* 11: 61-64.
- Armstrong, J. 2012. Herbicide How-to: Understanding Herbicide Mode of Action. Oklahoma Cooperative Extension Service PSS2778.
https://www.pioneer.com/CMRoot/Pioneer/US/products/stewardship/Oklahoma_Herbicide_MOA.pdf
- Atsbaha Zebelo, S., C.M. Berteau, S. Bossi, A. Occhipinti, G. Gnani, and M.E. Maffei. 2011. *Chrysolina herbacea* modulates terpenoid biosynthesis of *Mentha aquatica* L. *PLoS ONE* 6(3).
- Austin, B. 1985. Evaluation of antimicrobial compounds for the control of Bacterial Kidney Disease in Rainbow Trout, *Salmo gairdneri* Richardson. *Journal of Fish Diseases* 8(2): 209-220.
- Austin, B., L.F. Stuckey, P.W.A. Robertson, I. Effendi, and D.R.W. Griffith. 1995. A probiotic strain of *Vibrio alginolyticus* effective in reducing diseases caused by *Aeromonas salmonicida*, *Vibrio anguillarum* and *Vibrio ordalii*. *Journal of Fish Diseases* 18: 93-96.
- Australian Department of the Environment and Heritage (ADEH). 2003. Weed management guide: Cabomba- *Cabomba caroliniana*. CRC for Australian Weed Management; Commonwealth Department of the Environment and Heritage. 6 pp.
- Avenant-Oldewage, A. 2001. *Argulus japonicus* in the Olifants River system: possible conservation threat? *South African Journal of Wildlife Research* 31(1-2): 59-63.
- Baird, C. 2005. Great Lakes and surrounding states have varied and conflicting salmonid importation requirements. *Michigan Aquaculture Association Newsletter* 14(1).

- Bakal, R.S. 2012. National Fish Hatchery System Addressing VHS. United States Fish and Wildlife Service. Available <http://www.fws.gov/fisheries/nfhs/VHS.htm>. Accessed 12 May 2012.
- Balogh, G.R. 1986. Distribution and seedling ecology of Purple Loosestrife in Ohio's Lake Erie marshes USA. *Ohio Journal of Science* 86(2): 51.
- Balyan, R.S., and R.K. Malik. 1989. Control of horse purslane (*Trianthema portulacastrum*) and barnyardgrass (*Echinochloa crus-galli*) in mung bean (*Vigna radiata*). *Weed Science* 37(5): 695-699.
- Bandín, I., I. Santos, A.E. Toranzo, and J.L. Barja. 1991. MICs and MBCs of chemotherapeutic agents against *Renibacterium salmoninarum*. *Antimicrobial Agents and Chemotherapy* 35: 1011-1013.
- Banks, P.A., B. Branham, K. Harrison, T. Whitson, and I. Heap. 2004. Determination of the potential impact from the release of glyphosate- and glufosinate-resistant *Agrostis stolonifera* L. in various crop and non-crop ecosystems. *Weed Science Society of America*. 65 pp.
- Barbiero, R.P., and M.L. Tuchman. 2004. Changes in the crustacean communities of Lakes Michigan, Huron, and Erie following the invasion of the predatory cladoceran *Bythotrephes longimanus*. *Canadian Journal of Fisheries and Aquatic Sciences* 61: 2111-2125.
- Barger, C.T., D.J. Moorehead, G.K. Douce, R.C. Reardon, and A.E. Miller. 2003. Invasive plants of the eastern U.S.: identification and control. USDA Forest Service - Forest Health Technology Enterprise Team. Morgantown, WV.
Available <http://www.dnr.state.il.us/stewardship/cd/eppc/watermilfoil.html>. Accessed 25 April 2013.
- Barker, W.L., J.B. Beam, and S.D. Askew. 2005. Effects of rimsulfuron lateral relocation on Creeping Bentgrass (*Agrostis stolonifera*). *Weed Technology* 19: 647-652.
- BASF Corporation. 2011. Plateau® herbicide. Research Triangle Park, NC.
- Bates, A.L., and C.S. Smith. 1994. Submersed plant invasions and declines in the Southeastern United States. *Lake and Reservoir Management* 10(1): 53-55.
- Baxa, D.V., K.S. Mukkatira, K.A. Beauchamp, G.O. Kelley, and R.P. Hedrick. 2006. Susceptible and resistant strains of *Tubifex tubifex* in lineage III: Preliminary research findings on the effect of substratum on the development and release of the triactinomyxon stage of *Myxobolus cerebralis* and impact of interactions. Available <http://whirlingdisease.montana.edu/biblio/pdfs/Symposia/BaxaWDS2006.pdf>. Accessed 14 May 2012.

- Beam, J.B., W.L. Barker, and S.D. Askew. 2006. Selective Creeping Bentgrass (*Agrostis stolonifera*) control in cool-season turfgrass. *Weed Technology* 20: 340-344.
- Becker, G.C. 1983. *Fishes of Wisconsin*. University of Wisconsin Press, Madison, WI. 1052 pp.
<http://digital.library.wisc.edu/1711.dl/EcoNatRes.FishesWI>.
- Beerling, D.J., and J.M. Perrins. 1993. Biological Flora of the British Isles: *Impatiens glandulifera* Royle (*Impatiens roylei* Walp.). *Journal of Ecology* 81 (2): 367-382.
- Bentley, S., J.B. Whittaker, and A.J.C. Malloch. 1980. Field experiments on the effects of grazing by a chrysomelid beetle (*Gastrophysa viridula*) on seed production and quality in *Rumex obtusifolius* and *Rumex crispus*. *Journal of Ecology* 68: 671-674.
- Bially, A., and H.J. MacIsaac. 2000. Fouling mussels (*Dreissena* spp.) colonize soft sediments in Lake Erie and facilitate benthic invertebrates. *Freshwater Biology* 43: 85-97.
- Bij de Vaate, A., K. Jazdzewski, H.A.M. Ketelaars, S. Gollasch, and G. Van der Velde. 2002. Geographical patterns in range extension of Ponto-Caspian macroinvertebrate species in Europe. *Canadian Journal of Fisheries and Aquatic Sciences* 59: 1159-1174.
- Blossey, B. 2002. Purple Loosestrife. in Van Driesche, R., et al., eds. *Biological control of invasive plants in the Eastern United States*. United States Department of Agriculture Forest Service, Publication FHTET-2002-04. 413 pp.
- Blossey, B., and D. Schroeder. 1995. Host specificity of three potential biological weed control agents attacking flowers and seeds of *Lythrum salicaria*. *Biological Control* 5: 47-53.
- Blossey, B., D. Schroeder, S.D. Hight, and R.A. Malecki. 1994a. Host specificity and environmental impact of the weevil *Hylobius transversovittatus*, a biological control agent of Purple Loosestrife (*Lythrum salicaria*). *Weed Science* 42: 128-133.
- Blossey, B., D. Schroeder, S.D. Hight, and R.A. Malecki. 1994b. Host specificity and environmental impact of two leaf beetles (*Galerucella californiensis* and *G. pusilla*), for the biological control of Purple Loosestrife (*Lythrum salicaria*). *Weed Science* 42: 134-140.
- Boelman, S.F., F.M. Neilson, E.A. Dardeau, and T. Cross, Jr. 1997. Zebra Mussel (*Dreissena polymorpha*) Control Handbook for Facility Operators, First Edition. Miscellaneous Paper EL-97-1, U.S. Army Engineer Waterways Experiment Station, Vicksburg, MS.
- Boogaard, M.A., T.D. Bills, and D.A. Johnson. 2003. Acute toxicity of TFM and a TFM/niclosamide mixture to selected species of fish, including Lake Sturgeon (*Acipenser fulvescens*) and Mudpuppies (*Necturus maculosus*), in laboratory and field exposures. *Journal of Great Lakes Research* 29(Supplement 1): 529-541.

- Boogaard, M.A., T.D. Bills, J.H. Selgeby, and D.A. Johnson. 1996. Evaluation of piscicides for control of Ruffe. *North American Journal of Fisheries Management* 16(3): 600-607.
- Boos, T.K. Kearns, C. LeClair, B. Panke, B. Sriver, and B. Williams. 2010. A field guide to terrestrial invasive plants in Wisconsin. Wisconsin Department of Natural Resources, Madison, Wisconsin.
- Borcherding, J., S. Murawski, and H. Arndt. 2006. Population ecology, vertical migration and feeding of the Ponto-Caspian invader *Hemimysis anomala* in a gravel-pit lake connected to the River Rhine. *Freshwater Biology* 51: 2376-2387.
- Borland, K., S. Campbell, R. Schillo, and P. Higman. 2009. A field identification guide to invasive plants in Michigan's natural communities. Michigan Department of Natural Resources, Michigan State University Extension, Michigan Natural Features Inventory. 119 pp.
- Bosnic, A.C., and C.J. Swanton. 1997. Economic decision rules for postemergence herbicide control of barnyardgrass (*Echinochloa crus-galli*). *Weed Science* 45(4): 557-563.
- Bouzillé, J.B., A. Bonis, B. Clément, and M. Godeau. 1997. Growth patterns of *Juncus gerardi* clonal populations in a coastal habitat. *Plant Ecology* 132(1): 39-48.
- Brandt, S.B., and S.P. Madon. 1986. Rainbow Smelt (*Osmerus mordax*) predation on slimy sculpin (*Cottus sognatus*) in Lake Ontario. *Journal of Great Lakes Research* 12(4): 322-325.
- Brandt, F.W., J.G. Van As, H.J. Schoombee, and V.L. Hamilton-Attwell. 1981. The occurrence and treatment of bothriocephalosis in the Common Carp, *Cyprinus carpio* in fish ponds with notes on its presence in the largemouth yellowfish *Barbus kimberleyensis* from the Vaal Dam, Transvaal. *Water SA* 7: 35-42.
- Branstrator, D.K., and J.T. Lehman. 1996. Evidence for predation by young-of-the-year Alewife and Bloater Chub on *Bythotrephes cederstroemi* in Lake Michigan. *Journal of Great Lakes Research* 22: 917-924.
- Braverman, M.P. 1996. Control of mannagrass (*Glyceria declineata*) and southern watergrass (*Luziola fluitans*) in water-seeded rice (*Oryza sativa*). *Weed Technology* 10(1): 68-71.
- Brock, T. 2012. Herbicides for Weed Brush Control in Natural Areas. Invasive Plant Association of Wisconsin (IPAW), Madison, Wisconsin.
- Brown, L.L., L.J. Albright, and T.P.T. Evelyn. 1990. Control of vertical transmission of *Renibacterium salmoninarum* by injection of antibiotics into maturing female Coho Salmon *Oncorhynchus kisutch*. *Diseases of Aquatic Organisms* 9: 127-131.
- Buchmann, K. 1997. Population increase of *Gyrodactylus derjavini* on Rainbow Trout induced by testosterone treatment of the host. *Diseases of Aquatic Organisms* 30: 145-150.

- Buchmann, K., and J. Bresciani. 2006. Monogenea (Phylum Platyhelminthes). *in* Woo, P.T.K., ed. Fish diseases and disorders, 2nd edition. CABI Publishing, Nosworthy Way, Wallingford Oxfordshire, United Kingdom.
- Buchmann, K., and R. Kristensen. 2003. Efficacy of sodium percarbonate and formaldehyde bath treatment against *Gyrodactylus derjavini* infestations of Rainbow Trout. North American Journal of Aquaculture 65: 25-27.
- Buenzow, M.A.K. 2010. Control of Invasive Plants. Wisconsin Department of Natural Resources. 9 pp.
- Bugbee, G.J. 2009. Control of *Potamogeton crispus* and *Myriophyllum spicatum* in Crystal Lake, Middletown, CT 2006 - 2008. The Connecticut Department of Environmental Protection, The Connecticut Agricultural Experiment Station, New Haven, CT. 26 pp.
- Bullock, G.L., H.M. Stuckey, and D. Mulcahy. 1978. Corynebacterial kidney disease: egg transmission following iodophore disinfection. Fish Health News 7: 51-52.
- Bultemeier, B.W., M.D. Netherland, J.A. Ferrell, and W.T. Haller. 2009. Differential herbicide response among three phenotypes of *Cabomba caroliniana*. Invasive Plant Science and Management 2(4): 352-359.
- Bur, M.T., D.M. Klarer, and K.A. Krieger. 1986. First records of a European cladoceran, *Bythotrephes cederstroemi*, in Lakes Erie and Huron. Journal of Great Lakes Research 12: 144-146.
- Bureau of Environmental Programs. 2009. Illinois: Summary of Plant Protection Regulations. Illinois Department of Agriculture. Springfield, IL.
Available <http://pest.ceris.purdue.edu/pest.php?code=PEJAGBE>.
- Bureau of Plant Industry. 2012. Summary of Plant Protection Regulations: Wisconsin Department of Agriculture, Trade & Consumer Protection. Madison, WI. 12 pp.
- Bushnell, J.H. 1974. Bryozoans. Pages 157-194 *in* Hart, C.W. and S.L.H. Fuller, eds. Pollution ecology of freshwater invertebrates. Academic Press. New York, NY.
- Bushnoe, T.M., D.M. Warner, L.G. Rudstam, and E.L. Mills. 2003. *Cercopagis pengoi* as a new prey item for Alewife (*Alosa pseudoharengus*) and Rainbow Smelt (*Osmerus mordax*) in Lake Ontario. Journal of Great Lakes Research 29(2): 205-212.
- Campbell, S., P. Higman, B. Slaughter, and E. Schools. 2010. A Field Guide to Invasive Plants of Aquatic and Wetland Habitats for Michigan. Michigan DNRE, Michigan State University Extension, Michigan Natural Features Inventory. 90 pp.

- Canadensys. 2012. *Carex disticha*. Université de Montréal Biodiversity Centre. Available <http://data.canadensys.net/vascan/name/Carex%20disticha>. Accessed 17 April 2012.
- Canadian Food Inspection Agency (CFIA). 2012. Whirling disease. Available <http://www.inspect.gc.ca/animals/aquaticanimals/diseases/reportable/whirlingdisease/factsheet/eng/1336686597267/1336686806593>. Accessed 19 June 2012.
- Carey, J.H. 1995. *Agrostis gigantea*. In: Fire Effects Information System. U.S. Department of Agriculture, Forest Service, Rocky Mountain Research Station, Fire Sciences Laboratory. Available <http://www.fs.fed.us/database/feis/>.
- Castells, E., and M.R. Berenbaum. 2006. Laboratory rearing of *Agonopterix alstroemeriana*, the defoliating Poison Hemlock (*Conium maculatum* L.) moth, and the effects of piperidine alkaloids on preference and performance. *Environmental Entomology* 35(3): 607- 615.
- Castells, E., and M.R. Berenbaum. 2008. Resistance of the generalist moth *Trichoplusia ni* (Noctuidae) to a novel chemical defense in the invasive plant *Conium maculatum*. *Chemoecology* 18: 11-18.
- Castells, E., M.A. Berhow, S.F. Vaughn, and M.R. Berenbaum. 2005. Geographic variation in alkaloid production in *Conium maculatum* populations experiencing differential herbivory by *Agonopterix alstroemeriana*. *Journal of Chemical Ecology* 31(8): 1693 -1709.
- Catindig, J.L.A., R.T. Lubigan, and D. Johnson. 2009. *Echinochloa crusgalli*. Rice Knowledge Bank, Crop Health, Weed Management. International Rice Research Institute Available <http://www.knowledgebank.irri.org/ipm/the-dirty-dozen/echinochloa-crus-galli-l-p-beauv.html>. Accessed 4 September 2012.
- Catling, P.M. 2005. New "top of the list" invasive plants of natural habitats in Canada. *Botanical Electronic News* 345: 1-5. Available <http://www.ou.edu/cas/botany-micro/ben/ben345.html>. Accessed 2011.
- Catling, P.M., and B. Kostiuk. 2003. *Carex acutiformis* dominance of a cryptic invasive sedge at Ottawa. *Botanical Electronic News* 315: 1-6.
- Cavaletto, J., H. Vanderploeg, R. Pichlová-Ptácníková, S. Pothoven, J. Liebig, and G.L. Fahnenstiel. 2010. Temporal and spatial separation allow coexistence of predatory cladocerans: *Leptodora kindtii*, *Bythotrephes longimanus*, and *Cercopagis pengoi*, in southeastern Lake Michigan. *Journal of Great Lakes Research* 36(SP3): 65-73.
- Cavers, P.B., and J.L. Harper. 1964. Biological Flora of the British Isles No. 98 *Rumex obtusifolius* L. and *R. crispus*. *The Journal of Ecology* 52: 737-766.
- Centre for Ecology and Hydrology (CEH). 2004a. Information Sheet 2: Reeds, Rushes, Grasses and Sedges. Natural Environment Research Council, Centre for Aquatic Plant Management.

- Centre for Ecology and Hydrology (CEH). 2004b. Information Sheet 3: Himalayan Balsam. Centre for Ecology & Hydrology, Natural Environment Research Council. Natural Environment Research Council, Centre for Aquatic Plant Management.
- Centre for Ecology & Hydrology (CEH). 2004c. Information Sheet 6: Fringed Waterlily. Natural Environment Research Council, Centre for Aquatic Plant Management. 2 pp.
- Centre for Ecology and Hydrology (CEH). 2004d. Information Sheet 10: Spiked Water Milfoil. Natural Environment Research Council, Centre for Aquatic Plant Management.
- Centre for Ecology and Hydrology (CEH). 2004e. Information Sheet 12: Curled Pondweed. Natural Environment Research Council, Centre for Aquatic Plant Management. 2 pp.
- Centre for Ecology and Hydrology (CEH). 2004f. Information Sheet 15: Poison-Hemlock. Centre for Ecology & Hydrology, Natural Environment Research Council. Natural Environment Research Council, Centre for Aquatic Plant Management.
- Center for Food Security and Public Health (CFSPH). 2007. Viral hemorrhagic septicemia. Available http://www.cfsph.iastate.edu/Factsheets/pdfs/viral_hemorrhagic_septicemia.pdf. Accessed 24 May 2012.
- Champion, P.D., T.K. James, and E.C. Carney. 2008. Evaluation of triclopyr triethylamine for the control of wetlands weeds. *New Zealand Plant Protection* 61: 374-377.
- Chandler, M.A., L.C. Skinner, and L.C. Van Riper. 2010. Biological control of invasive plants in Minnesota. Minnesota Department of Agriculture, Minnesota Department of Natural Resources. St. Paul, MN. 2 pp.
- Charpentier, A., F. Mesléard, and P. Grillas. 1998. The role of water level and salinity in the regulation of *Juncus gerardi* populations in former ricefields in southern France. *Journal of Vegetation Science* 9(3): 361-370.
- Clark, F.H., C. Mattrick, and S. Shonbrun. 1998. Rogues Gallery: New England's Notable Invasives. *New England Wildflower Society*. 2(3): 19-26.
- Clearwater, S.J., C.W. Hickey, and M.L. Martin. 2008. Overview of potential piscicides and molluscicides for controlling aquatic pest species in New Zealand. Science & Technical Publishing, New Zealand Department of Conservation, Wellington, New Zealand.
- Colorado Division of Wildlife (CDW). 2011. Whirling disease and Colorado's trout. Available <http://wildlife.state.co.us/Fishing/Management/Pages/WhirlingDisease.aspx>. Accessed 12 May 2012.
- Cooper, M.R., and A.W. Johnson. 1984. Poisonous plants in Britain and their effects on animals and man. Her Majesty's Stationery Office, London, England.

Corbeil, S., and M.S. Crane. 2009. *Piscirickettsia salmonis*. Sub-Committee in animal health and laboratory standards Australian and New Zealand standard diagnostic procedure. Available http://www.scahls.org.au/data/assets/pdf_file/0005/1516514/Piscirickettsia_salmonis.pdf. Accessed 14 June 2012.

Corn, M.L., and R. Johnson. 2013. Invasive Species: Major Laws and the Role of Selected Federal Agencies. Congressional Research Service (CRS).

Cornell University Experimental Ponds. 2004. *Acentria ephemerella*. Available: <http://www.eeb.cornell.edu/ponds/acentria.htm>. Accessed 23 October 2013.

Cornell University. 2012. Barnyard Grass *Echinochloa crus-galli* (L.) Beauv. Weed Ecology and Management Laboratory. Available: <http://weedecology.css.cornell.edu/weed/weed.php?id=11>.

Cosyns, E., S. Claerbout, I. Lamoot, and M. Hoffmann. 2005. Endozoochorous seed dispersal by cattle and horse in a spatially heterogeneous landscape. *Plant Ecology* 178(2): 149-162.

Craft, C.D., and C.A. Myrick. 2011. Evaluation of Quagga Mussel veliger thermal tolerance: final report- January 2011 research season. Colorado State University.

Creed Jr., R.P. 1998. A biogeographic perspective on Eurasian Watermilfoil declines: Additional evidence for the role of herbivorous weevils in promoting declines? *Journal of Aquatic Plant Management* 36: 16-22.

Creed Jr., R.P., and S.P. Sheldon. 1995. Weevils and Watermilfoil: Did a North American herbivore cause the decline of an exotic plant? *Ecological Applications* 5(4): 1113-1121.

Crosier, D.M., D.P. Molloy, and J.E. Marsden. 2012. Ruffe - *Gymnocephalus cernuus*. New York State Museum and The University of Vermont. Available http://el.ercd.usace.army.mil/ansrp/gymnocephalus_cernuus.pdf. Accessed 18 June 2012.

Crossman, E. J. 1991. Introduced Freshwater Fishes: A Review of the North American Perspective With Emphasis on Canada. *Canadian Journal of Fisheries and Aquatic Sciences* 48(Suppl. 1): 46-58.

Crowder, L.B., and H.L. Crawford. 1984. Ecological shifts in resource use by bloaters in Lake Michigan. *Transactions of the American Fisheries Society* 113: 694-700.

CSIRO Entomology. 2011. Biological control of *Cabomba*. Available <http://www.csiro.au/science/ps2ku#a2>. Accessed 16 April 2013.

Curran, W.S., D.D. Lingenfelter, and L. Garling. 2009. Conservation Tillage Series: An introduction to weed management for conservation tillage systems. Pennsylvania State University, College of Agricultural Sciences: Agricultural Research and Cooperative Extension University Park, PA. 8 pp.

- Dal Bello, G.M., and M.R. Carranza. 1995. Weed diseases in LaPlata area II: Identification of pathogens with potential for weed biocontrol programmes. *Revista de la Facultad de Agronomía* 71: 7-14.
- Das, B.P., and N. Das. 2005. Impacts of quicklime (CaO) on the toxicity of copper (CuSO₄, 5H₂O) to fish and fish food organisms. *Chemosphere* 61(2): 186-191.
- Dawson, V.K., and C.S. Kohlar. 2003. Integrated management techniques to control nonnative fishes. United States Geological Survey, Upper Midwest Environmental Sciences Center, La Crosse, Wisconsin, Completion Report 01-AA-32-0040. 146pp.
- Dawson, V.K., L.L. Marking, and T.D. Bills. 1976. Removal of toxic chemicals from water with activated carbon. *Transactions of the American Fisheries Society* 105(1): 119-123.
- Dawson, V.K., T.D. Bills, and M.A. Boogaard. 1998. Avoidance behavior of Ruffe exposed to selected formulations of piscicides. *Journal of Great Lakes Research* 24(2): 343-350.
- Dendy, J.S. 1978. Polyps of *Craspedacusta sowerbyi* as predators on young striped bass. *The Progressive Fish-Culturist* 40(1): 5-6.
- Ding, J., B. Blossey, Y. Du, and F. Zheng. 2006. *Galerucella birmanica* (Coleoptera: Chrysomelidae), a promising potential biological control agent of Water Chestnut, *Trapa natans*. *Biological Control* 36(1): 80-90.
- DiTomaso, J.M., G.B. Kyser, et al. 2013. Weed Control in Natural Areas in the Western United States. Weed Research and Information Center, University of California. 544 pp
- Dodson, S.I., and S.D. Cooper. 1983. Trophic relationships of the freshwater jellyfish *Craspedacusta sowebyi* Lankester 1880. *Limnology and Oceanography* 28(2): 345-351.
- Doll, J., P. Compton, and R. Gildersleeve. The Dirty Dozen and Beyond: 25 Pasture Weeds of Wisconsin. University of Wisconsin- Extension. Publication GWQ042.
- Donovan, C.J., J.C. Ku, M.A. Quilliam, and T.A. Gill. 2008. Bacterial degradation of paralytic shellfish toxins. *Toxicon* 52: 91-100.
- Dwyer, W.P., B.L. Kerans, and M.M. Gangloff. 2003. Effect of acute exposure to chlorine, copper sulfate, and heat on the survival of New Zealand mud snails. *Intermountain Journal of Sciences* 9: 53-58.
- Dwyer and Kerans, unpublished.
- Dybdahl, M.F., A. Emblidge, and D. Drown. 2005. Studies of a trematode parasite for the biological control of an invasive freshwater snail. Report to the Idaho Power Company.

- Easy Butterfly Garden. 2013. *Polygonum persicaria*. Available <http://www.easy-butterfly-garden.com/polygonum-persicaria.html>. Accessed 9 December 2013.
- Ebert, D. 2005. Chapter 9, Population Dynamics and Community Ecology in Ecology, Epidemiology, and Evolution of Parasitism in Daphnia. National Center for Biotechnology Information. Bethesda, Maryland.
- Edlund, M.B., C.M. Taylor, C.L. Schelske, and E.F. Stoermer. 2000. *Thalassiosira baltica* (Grunow) Ostensfeld (Bacillariophyta), a new exotic species in the Great Lakes. Canadian Journal of Fisheries and Aquatic Sciences 57: 610-615.
- Edwards, D.I., and D.P. Taylor. 1963. Host range of an Illinois population of the stem nematode (*Ditylenchus dipsaci*) isolated from onion. Nematologica 9: 305-312.
- Elliott, D.G., R.J. Pascho, and G.L. Bullock. 1989. Developments in the control of Bacterial Kidney Disease of salmonid fishes. Diseases of Aquatic Organisms 6: 201-215.
- Ellis, S., and H.J. MacIsaac. 2009. Salinity tolerance of Great Lakes invaders. Freshwater Biology 54(1): 77-89.
- El-Matbouli, M., and R.W. Hoffman. 1991. Prevention of experimentally induced whirling disease in Rainbow Trout *Oncorhynchus mykiss* by Fumagillin. Diseases of Aquatic Organisms 10: 109-113.
- El-Matbouli, M., T.S. McDowell, D.B. Antonio, K.B. Andree, and R.P. Hedrick. 1999. Effect of water temperature on the development, release and survival of the triactinomyxon stage of *Myxobolus cerebralis* in its oligochaete host. International Journal of Parasitology 29(4): 627-641.
- Elmore, C. 1998. New weed in California: creeping field cress. IPM Notes, 3(2): unpaginated.
- Elmore, C.L., L. Kuhns, and T. Harpster. 1996. *Rorippa sylvestris*, creeping fieldcress: A threat to production ornamentals and its control. Hort Science 31(4): 578.
- ENSR International. 2005. Rapid Response Plan for Curlyleaf Pondweed (*Potamogeton crispus*) in Massachusetts. Massachusetts Department of Conservation and Recreation. Westford, MA. 25 pp.
- Etherington, J.R. 1984. Comparative studies of plant growth and distribution in relation to waterlogging: X. Differential formation of adventitious roots and their experimental excision in *Epilobium hirsutum* and *Chamerion angustifolium*. Journal of Ecology 72(2): 389 -404.
- Evans, J.R., J.J. Nugent, and J.K. Meisel. 2003. Invasive Plant Species Inventory and Management Plan for the Hanford Reach National Monument. The Nature Conservancy of Washington, Seattle, WA. 198 pp.

Evelyn, T.P.T., J.E Ketcheson, and L. Prosperi-Porta. 1984. Further evidence for the presence of *Renibacterium salmoninarum* in salmonid eggs and for the failure of povidone-iodine to reduce the intra-ovum infection rate in water-hardened eggs. *Journal of Fish Diseases* 7: 173-182.

Evelyn, T.P.T., L. Prosperi-Porta, and J.E. Ketcheson. 1986. Persistence of the kidney-disease bacterium, *Renibacterium salmoninarum*, in Coho Salmon, *Oncorhynchus kisutch* (Walbaum), eggs treated during and after water-hardening with povidone-iodine. *Journal of Fish Diseases* 9: 461-464.

Faisal, M., and D. Garling. 2004. What is Whirling Disease? North Central Regional Aquaculture Center Fact Sheet Series #113. Available <http://www.ustfa.org/Trout%20production/What%20is%20Whirling%20Disease%20%20ncrac%202004.pdf>. Accessed 12 May 2012.

Falck, M., and S. Garske. 2003. Invasive Non-native Plant Management Plan During 2002. Administrative Report 02-12. Great Lakes Indian Fish & Wildlife Commission (GLIFWC), Odanah, Wisconsin.

Falck, M., D. Olson, and S. Garske. 2009. Invasive Species Program 2008. Administrative Report 09-11. Great Lakes Indian Fish & Wildlife Commission (GLIFWC), Odanah, Wisconsin.

Falck, M., D. Olson, and S. Garske. 2009. Invasive Species Program 2007. Administrative Report 08-06. Great Lakes Indian Fish & Wildlife Commission (GLIFWC), Odanah, Wisconsin.

Falck, M., L. Hill, and R. Parisien. 2011. Invasive Species Program 2010. Administrative Report 11-11. Great Lakes Indian Fish & Wildlife Commission (GLIFWC), Odanah, Wisconsin.

Falck, M., S. Garske, and D. Olson. 2006. Invasive Species Program 2003-2005. Administrative Report 06-03. Great Lakes Indian Fish & Wildlife Commission (GLIFWC), Odanah, Wisconsin.

Falck, M., S. Garske, and D. Olson. 2010. Invasive Species Program 2009. Administrative Report 10-11. Great Lakes Indian Fish & Wildlife Commission (GLIFWC), Odanah, Wisconsin.

Falck, M.W., Gilane, and R. Parisien. 2012. Invasive Species Program 2011. Administrative Report 12-02. Great Lakes Indian Fish & Wildlife Commission (GLIFWC), Odanah, Wisconsin.

Falinska, K. 1999. Seed bank dynamics in abandoned meadows during 20-year period in the Bialowieza National Park. *Journal of Ecology* 87(3): 461-475.

Feeny, P., W.S. Blau, and P.M. Kareiva. 1985. Larval growth and survivorship of the black swallowtail butterfly in central New York. *Ecological Monographs* 55(2): 167-187.

Fijan, N. 1999. Spring viraemia of carp and other viral diseases and agents of warm-water fish. Pages 177–244 in Woo, P.T.K., and D.W. Bruno, eds. *Fish Diseases and Disorders 3: Viral, bacterial and fungal infections*. CAB International, New York, United States of America.

Finlayson, B.J., R.A. Schnick, R.L. Cailteux, L. Demong, W.D. Horton, W. McClay, and C.W. Thompson. 2002. Assessment of antimycin A use in fisheries and its potential for reregistration. *Fisheries* 27(6): 10-18.

Fisheries and Oceans Canada (FOC). 2009. Sea Lamprey Control. <http://www.dfo-mpo.gc.ca/regions/central/pub/bayfield/06-eng.htm>. Accessed 26 July 2012.

Fisheries Technical Services (FTS). 2012. *Aeromonas salmonicida*. Environment Agency. Available http://www.environmentagency.gov.uk/static/documents/Research/Aeromonas_salmonicida_in_cyprinids.pdf. Accessed 14 May 2012.

Folino, N.C. 2000. The freshwater expansion and classification of the colonial hydroid Cordylophora (Phylum Cnidaria, Class Hydrozoa). Pages 139-144 in Pederson, J., ed. *Marine Bioinvasions: Proceedings of the First National Conference*, January 24-27, 1999. Massachusetts Institute of Technology Sea Grant College Program, Cambridge, Massachusetts.

Food and Agriculture Organization (FAO). 2012. *Echinochloa crus-galli* (L.) Beauv. United Nations. Available <http://www.fao.org/ag/AGP/AGPC/doc/Gbase/DATA/Pf000227.htm>.

Forest Health Staff. 2006a. Bittersweet Nightshade: *Solanum dulcamara* L. United States Department of Agriculture Forest Service.

Forest Health Staff. 2006b. Carolina Fanwort; *Cabomba caroliniana* Gray. United States Department of Agriculture Forest Service.

Forest Health Staff. 2006c. Narrow-Leaved Cattail, *Typha angustifolia* L. Weed of the Week. United States Department of Agriculture Forest Service. Newtown Square, PA.

Forest Health Staff. 2006d. Poison Hemlock; *Conium maculatum* L. Weed of the Week. United States Department of Agriculture Forest Service. Newton Square, PA.

Forest Health Staff. 2006e. Reed Mannagrass: *Glyceria maxima* (Hartman) Holmb. Weed of the Week. United States Department of Agriculture Forest Service. Newtown, PA.

Forest Health Staff. 2006f. Yellow Iris: *Iris pseudacorus* L. Weed of the Week. United States Department of Agriculture Forest Service. Newton Square, PA. Available http://na.fs.fed.us/fhp/invasive_plants/weeds/yellow-iris.pdf.

Fraser, N. 2000. *Cirsium palustre* (Marsh Thistle). Literature search and habitat potential risk analysis. Prepared for Ministry of Forests, Forest Practices Branch, Kamloops, British Columbia. 53 pp. Available http://www.for.gov.bc.ca/hra/Publications/invasive_plants/marshplumethistleassessment2000.pdf.

- Freese, G. 1995. Structural Refuges in two stem-boring weevils on *Rumex crispus*. *Ecological Entomology* 20: 351-358.
- Fryer, J.L., C.N. Lannan, L.H. Garces, J.J. Larenas, and P.A. Smith. 1990. Isolation of a rickettsia-like organism from diseased Coho Salmon *Oncorhynchus kisutch* in Chile. *Fish Pathology* 25: 107-114.
- Fryer, J.L., C.N. Lannan, S.J. Giovannoni, and N.D. Wood. 1992. *Piscirickettsia salmonis* new genus new species, the causative agent of an epizootic disease in salmonid fishes. *International Journal of Systematic Bacteriology* 42: 120-126.
- Gault, N.F.S., D.J. Kilpatrick, and M.T. Stewart. 2002. Biological control of the fish louse in a Rainbow Trout fishery. *Journal of Fish Biology* 60(1): 226-237.
- Gealy, D.R., E.J. Wailes Jr., L.E. Estorninos, and R.S.C. Chavez. 2003. Rice cultivar differences in suppression of barnyardgrass (*Echinochloa crus-galli*) and economics of reduced propanil rates. *Weed Science* 51(4): 601-609.
- Gemza, A.F. 1995. Zooplankton seasonal dynamics and community structure in Severn Sound, Lake Huron. *Water Quality Research Journal of Canada* 30(4): 673-691.
- Getsinger, K.D., J.D. Madsen, T.J. Koschnick, M.D. Netherland, R.M. Stewart, D.R. Honnell, A.G. Staddon, and C.S. Owens. 2001. Whole-Lake Applications of Sonar™ for Selective Control of Eurasian Watermilfoil. United States Army Corps of Engineers, Engineer Research and Development Center, Aquatic Plant Control Research Program. 58 pp.
- Gildberg, A., A. Johansen, and J. Bogwald. 1995. Growth and survival of Atlantic Salmon (*Salmo salar*) fry given diets supplemented with fish protein hydrolysate and lactic acid bacteria during a challenge trial with *Aeromonas salmonicida*. *Aquaculture* 138: 23-34.
- Gilderhus, P.A. 1972. Exposure times necessary for antimycin and rotenone to eliminate certain freshwater fish. *Journal of the Fisheries Research Board of Canada* 25(2): 199-202.
- Gilderhus, P.A., R.M. Burress, and C.R. Walker. 1981. Simulated field testing methods for pest control agents in lotic and lentic ecosystems. Pages 161-166. *in* Schafer Jr., E.W., and C.R. Walker, eds. American Society for Testing and Materials.
- Global Invasive Species Database (GISD). 2013. *Corbicula fluminea*. Available http://www.issg.org/database/species/management_info.asp?si=537&fr=1&sts=&lang=EN Accessed 28 October 2013.
- Gonzalez, R.J., and W.A. Dunson. 1989. Differences in low pH tolerance among closely related sunfish of the genus *Enneacanthus*. *Environmental Biology of Fishes* 26(4): 303-310.

Great Lakes Fish Health Committee (GLFHC). 2000a. Fact sheet 5: Sea Lamprey barriers. New technologies help solve an old problem. Ann Arbor, Michigan.

Great Lakes Fish Health Committee (GLFHC). 2000b. Fact sheet 6: Sterile-male-release technique, An innovative Sea Lamprey control method. Ann Arbor, Michigan.

Great Lakes Fish Health Committee (GLFHC). 2006. Annual Agency Reports. 58 pp.
<http://www.glfc.org/boardcomm/fhealth/2006agencyreports.pdf>. Accessed 15 May 2012.

Great Lakes Fish Health Committee (GLFHC). 2012b. *Heterosporis* sp: Yellow Perch parasite. Available <http://www.glfc.org/research/reports/Marcquenski.pdf>. Accessed 23 July 2012.

Great Lakes Indian Fish & Wildlife Commission (GLIFWC). 2006. Eurasian Marsh Thistle (*Cirsium palustre*). Available http://www.glifwc.org/invasives/Cirsium_palustre/control.html. Accessed 15 May 2012.

Great Lakes Indian Fish & Wildlife Commission (GLIFWC). 2013. Crack Willows (*Salix alba*). Available http://invasives.glifwc.org/Salix_spp/control.html. Accessed 9 December 2013.

Great Lakes Panel on Aquatic Nuisance Species (GLPANS). 2008. Prohibited species in the Great Lakes region. Research Coordination Committee, GLPANS. Available:
<http://www.glc.org/ans/pdf/08-11-26-Great%20Lakes%20Reg%20Species%20List-complete.pdf>.

Great Lakes Restoration Initiative (GLRI) Task Force. 2010. FY2010-FY2014 Great Lakes Restoration Initiative Action Plan.

Gonzalez, M.J., and G.A. Burkart. 2004. Effects of food type, habitat, and fish predation on the relative abundance of two amphipod species, *Gammarus fasciatus* and *Echinogammarus ischnus*. Journal of Great Lakes Research 30: 100-113.

Gorokhova, E., T. Fagerberg, and S. Hansson. 2004. Predation by Herring (*Clupea harengus*) and Sprat (*Sprattus sprattus*) on *Cercopagis pengoi* in a western Baltic Sea bay. ICES Journal of Marine Science 61(6): 959-965.

Graham, J.M., and L.E. Graham. 1987. Growth and reproduction of *Bangia atropurpurea* (Roth) C. Ag. (*Rhodophyta*) from the Laurentian Great Lakes. Aquatic Botany 38(3-4): 317-332.

Gram, L., J. Melchiorson, B. Spanggaard, I. Huber, and T. Nielsen. 1999. Inhibition of *Vibrio anguillarum* by *Pseudomonas fluorescens* strain AH2—a possible probiotic treatment of fish. Applied and Environmental Microbiology 65: 969–973.

Grant, A.N., and L.A. Laidler. 1993. Assessment of antimicrobial sensitivity of *Aeromonas salmonicida* isolates from farmed Atlantic Salmon in Scotland. Veterinary Record 133:389-391.

- Gray, D.K., S.A. Bailey, I.C. Duggan, and H.J. MacIsaac. 2005. Viability of invertebrate diapausing eggs exposed to saltwater: implications for Great Lakes' ship ballast management. *Biological Invasions* 7: 531-539.
- Green, R.J. 1961. Septoria leafspot disease of scotch Spearmint. *Plant Disease Report* 45: 696.
- Grigorovich, I.A., O.V. Pashkova, Y.F. Gromova, and C.D. van Overdijk. 1998. *Bythotrephes longimanus* in the Commonwealth of Independent States: variability, distribution and ecology. *Hydrobiologia* 379: 183-198.
- Grime, T., 1995. The Great Lakes' Other Mussel Menace! University of Guelph, Canada. http://www.uoguelph.ca/research/publications/Assets/HTML_MAGS/oasis/environ4.html
- Groman, D.B., and G.W. Klonz. 1983. Chemotherapy and prophylaxis of Bacterial Kidney Disease with erythromycin. *Journal of the World Mariculture Society* 14: 226-235.
- Guardabassi, L., and P. Courvalin. 2006. Modes of antimicrobial action and mechanisms of bacterial resistance. in Aarestrup, F.M, ed. *Antimicrobial Resistance in Bacteria of Animal Origin*. ASM Press, Washington, D.C.
- Gucker, C.L. 2009. *Cirsium plaustre*. in United States Department of Agriculture Forest Service, Rocky Mountain Research Station, Fire Sciences Lab. Fire Effects Information System. Available <http://www.fs.fed.us/database/feis/plants/forb/cirpal/all.html>.
- Gudmundsdóttir, S., S. Helgason, H. Sigurjónsdóttir, S. Matthíasdóttir, H. Jónsdóttir, B. Laxdal, and E. Benediksdóttir. 2000. Measures applied to control *Renibacterium salmoninarum* infection in Atlantic Salmon: a retrospective study of two sea ranches in Iceland. *Aquaculture* 186: 193-203.
- Gumbart, D. 2012. *Cirsium palustre*, European Swamp Thistle. The Nature Conservancy.
- Gutierre, S.M., and M. Gutierre. 2012. pH Tolerance of the Biofouling Invasive hydrozoan *Cordylophora Caspia*. *Hydrobiologia* 671: 91-95.
- Hall Jr., L.W., M.C. Ziegenfuss, R.D. Anderson, and B.L. Lewis. 1995. The effect of salinity on the acute toxicity of total and free cadmium to a Chesapeake Bay copepod and fish. *Marine Pollution Bulletin* 30(6): 376-384.
- Hamel, M.J., M.L. Brown, and S.R. Chipps. 2008. Behavioral responses of Rainbow Smelt to in situ strobe lights. *North American Journal of Fisheries Management* 28(2): 394-401.
- Hansen, M.J., and M.E. Holey. 2002. Ecological factors affecting the sustainability of chinook and Coho Salmon populations in the Great Lakes, especially Lake Michigan. Pages 155-180 in Taylor, W.W., K.D. Lynch, and M.L. Jones, eds. *Sustaining North American Salmon: Perspectives Across Regions and Disciplines*.

- Hansen, A.M., and E. Jeppesen. 1992. Changes in the abundance and composition of cyclopoid copepods following fish manipulation in eutrophic Lake Vaeng, Denmark. *Freshwater Biology* 28(2): 183-193.
- Hanson, D., K. Gareve, and T. Markeson. 2012. Minnesota noxious weeds. Minnesota Department of Transportation, Office of Environmental Stewardship, Roadside Vegetation Management Unit. St. Paul, Minnesota. 36 pp.
- Hart, S.E., F. Yelverton, E.K. Nelson, D.W. Lycan, and G.M. Henry. 2005. Response of glyphosate-resistant and glyphosate-susceptible bentgrass (*Agrostis spp.*) to postemergence herbicides. *Weed Technology* 19: 549-559.
- Hartmann, E., H. Schuldes, R. Kübler, and W. Konold. 1995. Neophyten: Biologie, Verbreitung und Kontrolle ausgewählter Arten. Ecomed, Landsberg, Germany.
- Hastings, T.S., and A. McKay. 1987. Resistance of *Aeromonas salmonicida* to oxolinic acid. *Aquaculture* 61: 165-171.
- Havel, J.E. 2011. Survival of the exotic Chinese Mysterysnail (*Cipangopaludina chinensis malleata*) during air exposure and implications for overland dispersal by boats. *Hydrobiologia* DOI: 10.1007/s10750-010-0566-3. 8pp.
- Havens, K.E. 1994. An experimental comparison of the effects of two chemical stressors on a freshwater zooplankton assemblage. *Environmental Pollution* 84(3): 245-251.
- He, X., and G.W. LaBar. 1994. Interactive effects of cannibalism, recruitment, and predation on Rainbow Smelt in Lake Champlain: a modeling synthesis. *Journal of Great Lakes Research* 20(1):289-298.
- Hedrick, R.P., T.S. McDowell, G.D. Marty, L. Mukkatira, D.B. Antonio, K.B. Andree, Z. Bukhari, and T. Clancy. 2000. Ultraviolet irradiation inactivates the waterborne infective stages of *Myxobolus cerebralis*: a treatment for hatchery water supplies. *Diseases of Aquatic Organisms* 42: 53-59.
- Heidorn, R. 2011. Vegetation Management Guideline: Exotic Buckthorns. Illinois Natural History Survey: Prairie Research Institute, Illinois Department of Conservation. Available <http://www.inhs.illinois.edu/research/VMG/buckthorn.html>. Accessed 14 September 2012.
- Heinonen, J., J. Kukkonen, O-P. Penttinen, and I.J. Holopainen. 1997. Effects of hypoxia on valve-closure time and bioaccumulation of 2,4,5-trichlorophenol by the freshwater clam *Sphaerium corneum* [L.]. *Ecotoxicology and Environmental Safety* 36: 49-56.
- Heo, G.J., and J.J. Seo. 1996. Efficacy of ciprofloxacin against selected fish pathogens in cultured fish. *Bulletin of the European Association of Fish Pathologists* 17: 8-12.

- Herman, R.L. 1968. Fish furunculosis. Transactions of the American Fisheries Society 97: 221-230.
- Hewett, S.W., and D.J. Stewart. 1989. Zooplanktivory by Alewives in Lake Michigan: Ontogenetic, seasonal, and historical patterns. Transaction of the American Fisheries Society 118: 581-596
- Hight, S.D., B. Blossey, J. Laing, and R. Declerck-Floate. 1995. Establishment of insect biological control agents from Europe against *Lythrum salicaria* in North America. Environmental Entomology 24: 967-977.
- Higman, P., and S. Campbell. 2009. Meeting the challenges of invasive plants: A framework for action. Michigan Department of Natural Resources Wildlife Division. Lansing, Michigan. 87 pp.
- Hirvela-Koski, V. 2005. Fish pathogens *Aeromonas salmonicida* and *Renibacterium salmoninarum*: Diagnostic and epidemiological aspects. National Veterinary and Food Research Institute and University of Helsinki: academic dissertation, 23 September 2005. Available <http://ethesis.helsinki.fi/julkaisut/ela/elint/vk/hirvela-koski/fishpath.pdf>. Accessed 19 May 2012.
- Ho, L., E. Sawade, and G. Newcombe. 2012. Biological treatment options for cyanobacteria metabolite removal: a review. Water Research 46: 1536-1548.
- Hoffman, G.L. 1974. Disinfection of contaminated water by ultraviolet irradiation, with emphasis on whirling disease (*Myxosoma cerebralis*) and its effect on fish. Transactions of the American Fisheries Society 103: 541-550.
- Hoffman, G.L., and C.E. Dunbar. 1961. Studies on *Myxosoma cerebralis* (Hofer) Phlen (Protozoa: Myxosporidea) the cause of whirling disease of trout. Annual meeting of the American Society of Parasitologists, Abstract 53, August 27-31, Lafayette, Indiana. Journal of Parasitology 47(4): 29.
- Hoffman, G.L., and G.L. Hoffman Jr. 1972. Studies on the control of whirling disease (*Myxosoma cerebralis*). Journal of Wildlife Diseases 8: 49-53.
- Hoffman, G.L., and J.J. O'Grodnick. 1977. Control of whirling disease (*Myxosoma cerebralis*): effects of drying, and disinfection with hydrated lime or chlorine. Journal of Fish Biology 10: 175-179
- Hoffman, G.L., and M.E. Markiw. 1977. Control of whirling disease (*Myxosoma cerebralis*): use of methylene blue staining as a possible indicator of effect of heat on spores. Journal of Fish Biology 10: 181-183.
- Hogan. M.C. *Cirsium palustre*. Encyclopedia of Life (EOL). Available: <http://eol.org/pages/468314/overview/>.

Höie, S. 1992. Resistensundersökelse av *Aeromonas salmonicida* subsp. *Salmonicida* stammar, Del II. Norsk Veterinær Tidsskr 104: 565-567.

Homoya, M.A. 2010. Invasive exotic plants in Indiana natural areas. Available <http://www.in.gov/dnr/naturepreserve/6346.htm> Accessed 2 February 2012.

Hopkins, M.J.G., and J.B. Whittaker. 1980. Interactions between *Apion* spp. (Coleoptera: Curculionidae) and Polygonaceae. II. *Apion violaceum* Kirby and *Rumex obtusifolius*. Ecological Entomology 5: 241-247.

Hoskins, A. 2012. Elephant Hawkmoth *Deilephila elpenor*. Moths of Britain and Europe. Available <http://www.learnaboutbutterflies.com/Britain%20-%20Deilephila%20elpenor.htm>. Accessed 23 May 2012.

Howell, W.E., and G.I. Mink. 1981. Viruses isolated from wild carrot and Poison Hemlock. Plant Disease 65: 277-279.

Hroudová, A., and P. Zákravský. 1999. Vegetation dynamics in a fishpond littoral related to human impact. Hydrobiologia 415: 139-145.

Hroudová, Z., A. Krahulcová, P. Zákravský, and V. Jarolímová. 1996. The biology of *Butomus umbellatus* in shallow waters with fluctuating water level. Hydrobiologia 340: 27-30.

Hruskova, M., A.L. Honek, and S. Pekar. 2005. *Coreus marginatus* (Heteroptera: Coreidae) as a natural enemy of *Rumex obtusifolius* (Polygonaceae). Acta Oecologica 28(3): 281-7.

Hulsmann, S., and T. Mehner. 1997. Predation by underyearling perch (*Perca fluviatilis*) on a *Daphnia galeata* population in a short-term enclosure experiment. Freshwater Biology 38(1): 209-219.

Idaho Invasive Species Council Technical Committee, The (IISCTC). 2007. Idaho Aquatic Nuisance Species Plan. 139 pp.

Ikuta, K., Y. Suzuki, and S. Kitamura. 2003. Effects of low pH on the reproductive behavior of salmonid fishes. Fish Physiology and Biochemistry 28(1-4): 407-410.

Illinois Department of Natural Resources (IL DNR). 2007. Vegetation Management Guideline: Purple Loosestrife (*Lythrum salicaria* L.). Illinois Department of Natural Resources 1(17): 11pp.

Illinois Department of Natural Resources (IL DNR). 2009a. Aquatic Invasive Species: European Frog-bit. Available http://www.in.gov/dnr/files/EUROPEAN_FROG-BIT.pdf. Accessed 16 April 2013.

Illinois Department of Natural Resources (IL DNR). 2009b. Aquatic Invasive Species: Curlyleaf Pondweed. Available: http://www.in.gov/dnr/files/CURLYLEAF_PONDWEED.pdf.

Imbeault, S., S. Parent, M. Lagacé, C.F. Uhland, and J.F. Blais. 2006. Using bacteriophages to prevent furunculosis caused by *Aeromonas salmonicida* in farmed Brook Trout. *Journal of Aquatic Animal Health* 18(3): 203-214.

Indiana Department of Natural Resources (IDNR). n.d. Tree Species Information. Available <http://www.in.gov/dnr/forestry/3624.htm>. Accessed 2 February 2012.

Indiana Department of Natural Resources (IDNR). 2005. AIS. *Heterosporis* sp. Available <http://www.in.gov/dnr/files/HETEROSPORIS.pdf>. Accessed 23 July 2012.

Inglis, V., G.N. Frerichs, S.D. Millar, and R.H. Richards. 1991a. Antibiotic resistance of *Aeromonas salmonicida* isolated from Atlantic Salmon, *Salmo salar*, in Scotland. *Journal of Fish Diseases* 14: 353-358.

Inglis, V., M.K. Soliman, I. Higuera Ciapara, and R.H. Richards. 1992. Amoxycillin in the control of furunculosis in Atlantic Salmon. *Veterinary Record* 18: 45-48.

Inglis, V., R.H. Richards, K.J. Varma, I.H. Sutherland, and E.S. Brokken. 1991b. Florfenicol in Atlantic Salmon, *Salmo salar*, tolerance and assessment of efficacy against furunculosis. *Journal of Fish Diseases* 14: 343-351.

Ingraham, R., and T. Claxton, pers. comm. in Wagner 2002 (below)

Inman, R.E. 1971. A preliminary evaluation of Rumex rust as a biological control agent for Curly Dock. *Phytopathology* 61: 102-107.

International Joint Commission (IJC). 2011. 2009-2011 Priority cycle report on binational aquatic invasive species rapid response. Prepared by the Binational Aquatic Invasive Species Rapid Response Work Group for the International Joint Commission. Canada and the United States.

Invasive Plants Association of Wisconsin (IPAW). 2012. About Invasive Plants: Buckthorn. Available <http://www.ipaw.org/invaders/buckthorn/index.aspx>. Accessed 14 September 2012.

Invasive Plant Council of British Columbia. 2008. T.I.P.S. Targeted Invasive Plant Solutions: Marsh Plume Thistle. Invasive Plant Council of British Columbia, Williams Lake, British Columbia.

Irianto, A., and B. Austin. 2002. Use of probiotics to control furunculosis in Rainbow Trout, *Oncorhynchus mykiss*. *Journal of Fish Diseases* 25(6): 333-342.

Jackson, M.B. 1988. The dominant attached filamentous algae of Georgian Bay, the North Channel, and eastern Lake Huron, Canada. Field ecology and biomonitoring potential during 1980. *Hydrobiologia* 163: 149-172.

- Jacobs, J. 2008. Ecology and Management of Purple Loosestrife (*Lythrum salicaria* L.). United States Department of Agriculture, Natural Resource Conservation Service. Invasive Species Technical Note No. MT-21, August 2008. 9 pp.
- Jacobs, M.J., and H.J. MacIsaac. 2007. Fouling of fishing line by the waterflea *Cercopagis pengoi*: a mechanism of human-mediated dispersal of zooplankton? *Hydrobiologia* 583(1): 119-126.
- Jacono, C.C. 2001. *Iris pseudacorus*. USGS Nonindigenous Aquatic Species Program. Available http://nas.er.usgs.gov/taxgroup/plants/docs/ir_pseud.html. Accessed 23 August 2012.
- Jankowski, T., T. Strauss, and H.T. Ratte. 2005. Trophic interactions of the freshwater jellyfish *Craspedacusta sowerbii*. *Journal of Plankton Research* 27(8): 811-823.
- Jeffery, L.S., and L.R. Robinson. 1990. Poison-hemlock (*Conium maculatum*) control in alfalfa (*Medicago sativa*). *Weed Technology* 4(3): 585-587.
- Jensen, D. 2011. Flowering Rush (*Butomus umbellatus*). Minnesota Sea Grant. Available <http://www.seagrant.umn.edu/ais/floweringrush>. Accessed 20 March 2012.
- Jöborn A., J.C. Olsson, A. Westerdahl, P.L. Conway, and S. Kjelleberg. 1997. Colonization in the fish intestinal tract and production of inhibitory substances in intestinal mucus and fecal extracts by *Carnobacterium* sp. strain K1. *Journal of Fish Diseases* 20: 383–392.
- Johnson, R.L., and B. Blossey. 2002. Eurasian Watermilfoil. Pages 79-90 in *Biological control of invasive plants in the eastern United States*. United States Department of Agriculture Forest Service Publication FHTET-2002-04.
- Johnson, D.A., T.A. Ball, and W.M. Hess. 1999. Image analysis of urediniospores that infect *Mentha*. *Mycologia*, 91(6): 1016-1020.
- Johnson, G.R., and K.M. Himanga. 2009. Recommended trees for southeast Minnesota: an ecosystem approach. University of Minnesota Extension Service. Available <http://www.extension.umn.edu/distribution/naturalresources/DD6574.html>. Accessed 2011.
- Jude, D.J., R.H. Reider, and G.R. Smith. 1992. Establishment of Gobiidae in the Great Lakes Basin. *Canadian Journal of Fisheries and Aquatic Sciences* 49: 416-421.
- Julien, M.H., and M.W. Griffiths. 1998. *Biological control of weeds: A world catalogue of agents and their target weeds*, 4th Edition. CABI Publishing, Wallingford, United Kingdom.
- Jutila, H.M. 1998. Seed banks of grazed and ungrazed Baltic seashore meadows. *Journal of Vegetation Science* 9: 395–408.

Jutila, H.M. 1999. Effect of grazing on the vegetation of shore meadows along the Bothnian Sea, Finland. *Plant Ecology* 140: 77–88.

Kamer, K., and P. Fong. 2000. A fluctuating salinity regime mitigates the negative effects of reduced salinity on the estuarine maroalga, *Enteromorpha intestinalis* (L.) link. *Journal of Experimental Marine Biology and Ecology* 254(1): 53-69.

Kamer, K., and P. Fong. 2001. Nitrogen enrichment ameliorates the negative effects of reduced salinity on the green macroalga *Enteromorpha intestinalis*. *Marine Ecology Progress Series* 218: 87-93.

Kaster, J.L., and J.H. Bushnell. 1981. Cyst formation by *Tubifex tubifex* (Tubificidae). *Transactions of the American Microscopy Society* 100: 34-41.

Kato-Noguchi, H., H.L. Thi, H. Sasaki, and K. Suenaga. 2012. A potent allelopathic substance in cucumber plants and allelopathy of cucumber. *Acta Physiologiae Plantarum* 34(5): 2045-2049.

Keely, P.E., and R.J. Thullen. 1991. Growth and interaction of barnyardgrass (*Echinochloa crus-galli*) with cotton (*Gossypium hirsutum*). *Weed Science* 39(2): 369-375.

Kells, J.J., W.F. Meggitt, and D. Penner. 1986. Activity of selective postemergence grass herbicide in soil. *Weed Science* 34(1): 62-65.

Kelly, A., and R.R. Southwood. 2006. Restoration of the littoral margin by removing trees from the lake edge at Cockshoot Broad, Norfolk, England. *Conservation Evidence* 3: 71-72.

Kelly, J., and C.M. Maguire. 2009. Fringed Water Lily (*Nymphaeodes peltata*) invasive species action plan. Northern Ireland Environmental Agency (NIEA) and National Parks and Wildlife Service (NPWS) as part of Invasive Species Ireland. 14 pp.

Kennay, J., and G. Fell. 2011. Vegetation Management Guideline: Moneywort (*Lysimachia nummularia*). Illinois Natural History Survey; Prairie Research Institute. Available <http://www.inhs.illinois.edu/research/VMG/moneywort.html>. Accessed 2 May 2013.

Kennedy, V.S. 2002. Swimming and settlement behavior in the Quagga Mussel. Maryland Sea Grant. Available http://www.mdsg.umd.edu/programs/research/projects/past/R_ZM-03/index.php

Kestrup, A.M., S.H. Thomas, K. van Rensburg, A. Ricciardi, and M.A. Duffy. 2011. Differential infection of exotic and native freshwater amphipods by a parasitic water mold in the St. Lawrence River. *Biological Invasions* 13: 769-779.

Khanh, T.D., I.M. Chung, S. Tawata, and T.D. Xuan. 2006. Weed suppression by *Passiflora edulis* and its potential allelochemicals. *Weed Research* 46(4): 296-303.

King County Noxious Weed Control Program. 2007a. Policeman's Helmet. Weed Alert. Department of Natural Resources and Parks, Water and Land Resources Division. King County, WA. 2 pp.

King County Noxious Weed Program. 2007b. Weed Alert: Bittersweet Nightshade. Department of Natural Resources and Parks, Water and Land Resources Division. King County, WA 2 pp.

King County Noxious Weed Control Program. 2008. *Epilobium hirsutum*. Department of Natural Resources and Parks, Water and Land Resources Division, King County.
<http://dnr.metrokc.gov/wlr/lands/weeds/pdf/Hairy-Willowherb-Fact-Sheet.pdf>.

King County Noxious Weed Control Program. 2009. Yellow-flag iris (*Iris pseudacorus*). Best Management Practices. Department of Natural Resources and Parks; Water and Land Resources Division. King County, WA. 7 pp.

King County Noxious Weed Control Program. 2010a. Policeman's Helmet: Best Management Practices. King County Department of Natural Resources and Parks, Water and Land Resources Division. King County, Washington.

King County Noxious Weed Program. 2010b. Garden Loosestrife, *Lysimachia vulgaris*. Best Management Practices. King County Department of Natural Resources and Parks, Water and Land Resources Division. King County, Washington.

King County Noxious Weed Program. 2010c. Best Management Practices: Bittersweet Nightshade. King County Department of Natural Resources and Parks, Water and Land Resources Division. King County, Washington.

King County Noxious Weed Control Program. 2011. Poison Hemlock. King County Department of Natural Resources and Parks, Water and Land Resources Division, King County, Washington.

King County Noxious Weed Control Program. 2012. Reed sweetgrass: *Glyceria maxima*. Noxious Weeds. Department of Natural Resources and Parks, Water and Land Resources Division, King County. Available
<http://www.kingcounty.gov/environment/animalsAndPlants/noxious-weeds/weed-identification/reed-sweetgrass.aspx>. Accessed on 6 August 2012.

Kirn, R.A., and G.W. LaBar. 1996. Growth and survival of Rainbow Smelt, and their role as prey for stocked almonids in Lake Champlain. Transactions of the American Fisheries Society 125(1): 87-96.

Kiryu, I., T. Sakai, J. Kurita, and T. Iida. 2007. Virucidal effect of disinfectants on Spring Viremia of Carp virus. Fish Pathology 42: 111–113.

Kleppel, G.S., and E. LaBarge. 2011. Using sheep to control purple loosestrife (*Lythrum salicaria*). Invasive Plant Science and Management 4: 50-57.

- Klein, H. 2011. Water Foxtail, *Alopecurus geniculatus* L. University of Alaska Anchorage, Alaska Natural Program. Available <http://aknhp.uaa.alaska.edu/botany/akepic/non-native-plants-alaska/>. Accessed 2011.
- Klinkenberg, B. 2010. *Alopecurus geniculatus* L. in E-Flora BC. Electronic atlas of plants of British Columbia. Available <http://www.geog.ubc.ca/biodiversity/eflora/index.shtml>. Accessed 6 February 2011.
- Kocik, J.F., and M.L. Jones. 1999. Pacific salmonines in the Great Lakes basin. Pages 455-488. in Taylor, W.W., and C.P. Ferreri, eds. Great Lakes Fisheries Policy and Management: A Binational Perspective. Michigan State University Press, East Lansing, Michigan.
- Koehn, J., A. Brumley, and P. Gehrke. 2000. Managing the Impacts of Carp. Bureau of Rural Sciences. Department of Agriculture, Fisheries and Forestry, Australia, Canberra.
- Koger, C.H., D.M. Dodds, and D.B. Reynolds. 2007. Effect of adjuvants and urea ammonium nitrate on bispyribas efficacy, absorption, and translocation in barnyardgrass (*Echinochloa crus-galli*). Weed Science 55(5): 399-405.
- Korting, W. 1974. Bothriocephalosis of the carp. Veterinar-Medizinische Nachrichten 2(74): 152-158.
- Krausz, R.F., B.G. Young, G. Kapusta, and J.L. Matthews. 2000. Application timing determines giant foxtail (*Setaria faberi*) and barnyardgrass (*Echinochloa crus-galli*) control in no-till corn (*Zea mays*). Weed Technology 14(1): 161-166.
- Larina, S.Y. 2008. *Chenopodium glaucum*. in Afonin, A.N., S.L. Greene, N.I. Dzyubenko, A.N. Frolov, eds. Interactive agricultural ecological atlas of Russia and neighboring countries: economic plants and their diseases, pests and weeds. Available http://www.agroatlas.ru/en/content/weeds/Chenopodium_glaucum/. Accessed 2011.
- Larson, J.V.S. 2009. Buckthorn: a threat to our native woodland ecosystems. 8 pp.
- Latimore, J.A., P. Filice, E. LaPorte, and S. Ariganello. 2011. Clean Boats, Clean Waters. Guidelines for Clean Boats, Clean Waters: Michigan's Aquatic Invasive Species Volunteer Program. Michigan Sea Grant, 35pp.
- Lauer, T.E., D.K. Barnes, A. Ricciardi, and A. Spacie. 1999. Evidence of recruitment inhibition of zebra mussel (*Dreissena polymorpha*) by a freshwater bryozoan (*Lophopodella carteri*). Journal of the North American Benthological Society 18(3): 406-413.
- Lawniczak, A.E., J. Zbierska, A. Choinski, and W. Szczepaniak. 2010. Response of emergent macrophytes to hydrological changes in a shallow lake, with special reference to nutrient cycling. Hydrobiologia 656: 243-254.

- Lee, E.G-H., and T.P.T. Evelyn. 1994. Prevention of vertical transmission of the Bacterial Kidney Disease agent *Renibacterium salmoninarum* by broodstock injection with erythromycin. *Diseases of Aquatic Organisms* 18: 1-4.
- Leigh, P. 1998. Benefits and costs of the Ruffe control program for the Great Lakes fishery. *Journal of Great Lakes Research* 24(2): 351-360.
- Lelek, A. 1987. *The Freshwater Fishes of Europe: Threatened Fishes of Europe*. Weisbaden: AULA Verlag.
- Lembi, C.A. 2003. Aquatic plant management. Purdue University Cooperative Extension Service. 20 pp.
- Lemke, A.M., J.A. Stoekel, and M.A. Pegg. 2003. Utilization of the exotic cladoceran *Daphnia lumholtzi* by juvenile fishes in an Illinois River floodplain lake. *Journal of Fish Biology* 62: 938-954.
- Lenssen, J.P.M., F.B.J. Menting, W.H. Van Der Putten, and C.W.P.M. Blom. 2000. Vegetative reproduction by species with different adaptation to shallow-flooded habitats. *New Phytologist* 145(1): 61-70.
- Lesko, L.T., P.L. Hudson, J.W. Reid, and M.A. Chriscinske. 2003. Harpacticoid copepods of the Laurentian Great Lakes. Great Lakes Science Center, Ann Arbor, Michigan.
- Li, J., G-Z. Zhao, H-Y. Huang, S. Qin, W-Y. Zhu, L-X. Zhao, L-H. Xu, S. Zhang, W-J. Li, and G. Strobel. 2012a. Isolation and characterization of culturable endophytic actinobacteria associated with *Artemisia annua* L. *Antonie van Leeuwenhoek* 101: 515-527.
- Lieffers, H.J. 1990. Effects of the lampricide 3-trifluoromethyl-4-nitrophenol on macroinvertebrate populations in a small stream. Great Lakes Fisheries Commission Technical Report 55, Ann Arbor, Michigan.
- Lienesch, P.W., and M. Gophen. 2001. Predation by Inland Silversides on an exotic cladoceran, *Daphnia lumholtzi*, in Lake Texoma, United States of America 59: 1249-1257.
- Lin, C.K., and J.L. Blum. 1976. *Bangia* a recent invader of the St Lawrence Great Lakes. *Journal of Phycology* 12: 19-20.
- Lind, E.A. 1977. A review of pikeperch (*Stizostedion lucioperca*), Eurasian perch (*Perca fluviatilis*), and Ruffe (*Gymnocephalus cernua*) in Finland. *Journal of Fisheries Research Board of Canada* 34: 1684-1695.
- Lindley, J.A., P. Donkin, S.V. Evans, C.L. George, and K.F. Uil. 1999. Effects of two organochlorine compounds on hatching and viability of calanoid copepod eggs. *Journal of Experimental Marine Biology and Ecology* 242(1): 59-74.

Lindsey, J.K. 2005. *Cirsium palustre* (Linnaeus 1753; Scopoli 1772). Ecological Flora of the British Isles. Available:

<http://www.commanster.eu/commanster/Plants/Flowers/SpFlowers/Cirsium.palustre.html>.

Linfield, R.S.J. 1980. Catchability and stock density of Common Carp, *Cyprinus carpio* L. in a lake fishery. Fisheries Management 11: 11–22.

Loeb, R., E. van Daalen, L.P.M. Lamers, and J.G.M. Roelofs. 2007. How soils characteristics and water quality influence the biogeochemical response to flooding in riverine wetlands. Hydrobiologia 656: 243-254.

Ling, N. 2003. Rotenone—a review of its toxicity and use for fisheries management. New Zealand Department of Conservation, Wellington, New Zealand.

Lintermans, M., and T. Raadik. 2003. Local eradication of trout from streams using rotenone: the Australian experience. Pages 95-111 in Managing invasive freshwater fish in New Zealand: Proceedings of a workshop hosted by the Department of Conservation. Hamilton, New Zealand.

Liu, M.S., and J.A. Hellebust. 1976. Effects of salinity changes on growth and metabolism of the marine centric diatom *Cyclotella cryptica*. Canadian Journal of Botany 54(9): 930-937.

Liyanage, Y.S., H. Yokoyama, and H. Wakabayashi. 2003. Evaluation of a vector-control strategy of haemorrhagic thelohanellosis in carp, caused by *Thelohanellus hovorkai* (Myxozoa). Diseases of Aquatic Organisms 55(1): 31-35.

Lloyd's Register Marine. 2014. Understanding ballast water management - Guidance for shipowners and operators. Report. 29 pp. Available http://www.lr.org/en/images/213-35824_Understanding_Ballast_Water_Management_0314_tcm155-248816.pdf . Accessed 12 February 2015.

Lougheed, V.L., T. Theÿsmeÿer, T. Smith, and P. Chow-Fraser. 2004. Carp exclusion, food-web interactions, and the restoration of Cootes Paradise Marsh. Journal of Great Lakes Research 30(1): 44-57.

Lougheed, V.L., and P. Chow-Fraser. 2001. Spatial variability in the response of lower trophic levels after carp exclusion from a freshwater marsh. Journal of Aquatic Ecosystem Stress and Recovery 9: 21-34.

Lougheed, V.L., and R.J. Stevenson. 2004. Exotic marine macroalga (*Enteromorpha flexuosa*) reaches bloom proportions in a coastal lake of Lake Michigan. Journal of Great Lakes Research 30(4): 538-544.

Lui, K., F.L. Thompson, and C.G. Eckert. 2005. Causes and consequences of extreme variation in reproductive strategy and vegetative growth among invasive populations of a clonal aquatic plant, *Butomus umbellatus* L. (Butomaceae). Biological Invasions 7: 427-444.

- Luo, X-Y., and H. Ikeda. 2007. Effects of four rice herbicides on the growth of an aquatic fern, *Marsilea quadrifolia* L. *Weed Biology and Management* 7: 237-241.
- Lysebo, E.M. 1995. Behavioural and morphological changes in polymorphic *Daphnia* related to different predation regimes. *Hydrobiologia*. 307: 185-191.
- Ma, X., and J.E. Purcell. 2005. Temperature, salinity, and prey effects on polyp versus medusa *Craspedacusta sowerbyi* in Nacogdoches Reservoir, Texas and associated physical-chemical conditions. *Texas Journal of Science* 33:17-23.
- MacConnell, E. pers. comm. in Wagner 2002 (below)
- MacDonald, G.E., D.G. Shilling, and T.A. Bewick. 1993. Effects of endothall and other aquatic herbicides on chlorophyll fluorescence, respiration and cellular integrity. *Journal of Aquatic Plant Management* 31: 50-55.
- Madenjian, C.P., M.A. Stapanian, L.D. Witzel, D.W. Einhouse, S.A. Pothoven, and H.L. Whitford. 2011. Evidence for predatory control of the invasive Round Goby. *Biological Invasions* 13: 987-1002
- Maes, J., M. Tackx, and K. Soetaert. 2005. The predation impact of juvenile Herring *Clupea harengus* and Sprat *Sprattus sprattus* on estuarine zooplankton. *Hydrobiologia* 540(1-3): 225-235.
- Maiolie, M.A., B. Harryman, and B. Ament. 2001. Response of free-ranging Kokanee to strobe lights. Pages 27-35. *in* Coutant, C., ed. *Behavioral Technologies for Fish Guidance*. American Fisheries Society, Symposium 26, Bethesda, Maryland.
- Makarewicz, J.C., and H.D. Jones. 1990. Occurrence of *Bythotrephes cederstroemi* in Lake Ontario offshore waters. *Journal of Great Lakes Research* 16: 143-147.
- Malik, M.A.B. 1997. Isolates of soil actinomycetes with potential for phytotoxin production. *Journal of Chemical Ecology* 23(12): 2683-2693.
- Mant, R.C., G. Moggridge, and D.C. Aldridge. 2011. Biofouling by bryozoans, cordylophora, and sponges in UK water treatment works. *Water Science and Technology* 63(9): 1815-822.
- Marking, L.L., and T.D. Bills. 1985. Effects of contaminants on toxicity of the lampricides TFM and Bayer 73 to three species of fish. *Journal of Great Lakes Research* 11(2): 171-178.
- Marking, L.L., T.D. Bills, J.J. Rach, and S.J. Grabowski. 1983. Chemical control of fish and fish eggs in the Garrison Diversion Unit, North Dakota. *North American Journal of Fisheries Management* 3(4): 410-418.

- Markiw, M.E. 1992. Salmonid Whirling Disease. United States Fish and Wildlife Service Leaflet 17: 1-11
- Marshall, J.S., D.L. Mellinger, and J.I. Parker. 1981. Combined effects of cadmium and zinc on a Lake Michigan zooplankton community. *Journal of Great Lakes Research* 7(3): 215-223.
- Maryland Sea Grant. 2012. Invasive Species in the Chesapeake Watershed: Water Chestnut, *Trapa natans* L. Available http://www.mdsg.umd.edu/issues/restoration/non-natives/workshop/water_chestnut.html. Accessed 2 May 2013.
- Masycheva, V.I., Y.S. Alikin, V.P. Klimenko, V.A. Fadina, I.S. Shchelkunovis, T.I. Shchelkunovis, and O.A. Kupinskaya. 1995. Comparative antiviral effects of dsRNA on lower and higher vertebrates. *Veterinary Research* 26: 536–538.
- Maule, A.G., D.W. Rondorf, J. Beeman, and P. Haner. 1996. Incidence of *Renibacterium salmoninarum* infections in juvenile hatchery spring Chinook Salmon in the Columbia and Snake Rivers. *Journal of Aquatic Animal Health* 8: 37-46.
- Maun, M.A., and S.C.H. Barrett. 1986. The biology of Canadian weeds. *Canadian Journal of Plant Science* 66: 739-759.
- Maurer, D. 2009. Stopping New Invasive Plants in Their Tracks. New Invaders Watch Program, Outdoor Illinois. 3 pp.
- Mayo, K.R., J.H. Selgeby, and M.E. McDonald. 1998. A bioengetics modeling evaluation of top-down control of Ruffe in the St. Louis River, western Lake Superior. *Journal of Great Lakes Research* 24 (2): 329-342.
- McAllister, P.E. 1990. Fish Disease Leaflet 83. Viral hemorrhagic septicemia of fishes. United States Fish and Wildlife Service, National Fisheries Research Center-Leetown, National Fish Health Research Laboratory, Kearneysville, West Virginia.
- McCarthy, D.H., and R.J. Roberts. 1980. Furunculosis of fish - the present state of our knowledge. Pages 293-341. *in* Droop, M.R., D.H. McCarthy, and H.W. Jannasch, eds. *Advances in aquatic microbiology*. Academic Press, London.
- McCullough, P.E., and S.E. Hart. 2006. Temperature influences Creeping Bentgrass (*Agrostis stolonifera*) and annual bluegrass (*Poa annua*) response to bispyribac-sodium. *Weed Technology* 20: 728-732.
- McHenry, J. pers. comm. in Pitcher 1985 (below)
- Medhurst, R.B. 2003. Presentation of results at the New Zealand Mudsail Stakeholder Meeting, November 17, 2003. Mammoth Lake, California

Menninger, H. 2013. The Asian Clam, *Corbicula fluminea*: A brief review of the scientific literature.

Messyasz, B., and A. Rybak. 2011. Abiotic factors affecting the development of *Ulva* sp. (Ulvophyceae; Chlorophyta) in freshwater ecosystems. *Aquatic Ecology* 45(1): 75-87.

Michel, C., J.P. Gerard, B. Fourbet, R. Collas, and R. Chevalier. 1980. Emploi de la flumequine contre la furunculose des Salmonides: essais therapeutiques et perspectives pratiques. *Bulletin Français de Pisciculture* 277: 154-162

Michigan Aquaculture Association (MAA). 2012. New label for AQUAFLO® offers more options, flexibility to U.S fish farmers. Available <http://michiganaquaculture.org/2012/04/10/new-label-for-aquaflor-offers-more-options-flexibility-to-us-fish-farmers/>. Accessed 31 May 2012.

Michigan Department of Natural Resources (MIDNR). 2012. Furunculosis. Available http://www.michigan.gov/dnr/0,4570,7-153-10364_52259_10950-27389--,00.html. Accessed 24 May 2012.

Michigan Department of Natural Resources (MIDNR) and Michigan Department of Environmental Quality (MIDEQ). 2009. Sustainable soil and water quality practices on forest land. 79 pp.

Michigan Natural Features Inventory (MNFI). 2012. Glossy Buckthorn- *Frangula alnus*. Invasive Species- Best Control Practices. Michigan Department of Natural Resources.

Michigan Sea Grant. 2012b. Eurasian Watermilfoil: *Myriophyllum spicatum*. Available <http://www.miseagrant.umich.edu/files/2012/12/06-710-EWM-milfoil-2012rev.pdf>. Accessed 23 April 2013.

Midwest Invasive Species Information Network (MISIN). 2013. Yellow floatingheart (*Nymphoides peltata*). Available <http://www.misin.msu.edu/facts/detail.php?id=182>. Accessed 29 April 2013.

Michigan Invasive Species Information Network (MISIN) and Michigan Natural Features Inventory (MNFI). 2013a. Curly pondweed (*Potamogeton crispus*). Michigan State University. Available <http://www.misin.msu.edu/facts/detail.php?id=43>. Accessed 29 April 2013.

Michigan Invasive Species Information Network (MISIN) and Michigan Natural Features Inventory (MNFI). 2013b. Eurasian Watermilfoil (*Myriophyllum spicatum*). Michigan State University. Available <http://www.misin.msu.edu/facts/detail.php?id=44>. Accessed 25 April 2013.

Michigan Invasive Species Information Network (MISIN) and Michigan Natural Features Inventory (MNFI). 2013c. Garden Yellow Loosestrife (*Lysimachia vulgaris*). Available <http://www.misin.msu.edu/facts/detail.php?id=92>. Accessed 3 May 2013.

Michigan Invasive Species Information Network (MISIN) and Michigan Natural Features Inventory (MNFI). 2013d. Gingermint (*Mentha x gracilis*). Available <http://www.misin.msu.edu/facts/detail.php?id=96>. Accessed 6 May 2013.

Michigan Invasive Species Information Network (MISIN) and Michigan Natural Features Inventory (MNFI). 2013e. Moneywort (*Lysimachia nummularia*). Available <http://www.misin.msu.edu/facts/detail.php?id=178>. Accessed 2 May 2013.

Miklovic, S. 2000. *Typha angustifolia* Management: Implications for Glacial Marsh Restoration. Restoration and Reclamation Review. Department of Horticultural Science, University of Minnesota St. Paul, Minnesota. 11 pp.

Mikulyuk, A., and M. Nault. 2011. *Hydrocharis morsus-ranae*. Available <http://www.cabi.org/isc/default.aspx?site=144&page=481&LoadModule=DataSheet&dsID=28140&CompID=5>. Accessed 16 April 2013.

Miller, M. 2013. Adirondack Park Agency Completed Applications. New York Department of Environmental Conservation. Available http://www.dec.ny.gov/enb/20130213_not5.html. Accessed 25 April 2013.

Millie, D.F., and R.L. Lowe. 1983. Studies on Lake Erie's littoral algae: host specificity and temporal periodicity of epiphytic diatoms. *Hydrobiologia* 99: 7-18.

Mills, C.E., and F. Sommer. 1995. Invertebrate introductions in marine habitats: two species of hydromedusae (Cnidaria) native to the Black Sea, *Maeotias inexpectata* and *Blackfordia virginica*, invade San Francisco Bay. *Marine Biology* 122: 279-288.

Mills, E.L., J.H. Leach, J.T. Carlton, and C.L. Secor. 1993. Exotic species in the Great Lakes: a history of biotic crises and anthropogenic introductions. *Journal of Great Lakes Research* 19(1): 1-54.

Mills, E.L., and J.L. Forney. 1983. Impact on *Daphnia pulex* of predation by young Yellow Perch in Oneida Lake, New York. *Transactions of the American Fisheries Society* 112: 151-161.

Minnesota Department of Natural Resources (MN DNR). 2012a. Yellow Iris (*Iris pseudacorus*). Available <http://www.dnr.state.mn.us/invasives/terrestrialplants/herbaceous/yellowiris.html>. Accessed 16 April 2013.

- Minnesota Department of Natural Resources (MNDNR). 2012b. Flowering Rush (*Butomus umbellatus*). Available <http://www.dnr.state.mn.us/invasives/terrestrialplants/herbaceous/floweringrush.html>. Accessed 20 March 2012.
- Minnesota Department of Natural Resources (MN DNR). 2013a. Grass carp (*Ctenopharyngodon idella*). Available <http://www.dnr.state.mn.us/invasives/aquaticanimals/grasscarp/index.html>. Accessed 16 April 2013.
- Minnesota Department of Natural Resources (MN DNR). 2013b. Minnesota invasive species laws. Available <http://www.dnr.state.mn.us/invasives/laws.html>. Accessed 16 April 2013.
- Minnesota Invasive Species Advisory Council. 2009. A Minnesota state management plan for invasive species. 76 pp.
- Missouri Botanical Garden. 2012. *Juncus inflexus*. Gardening Help. Available <http://www.missouribotanicalgarden.org/gardens-gardening/your-garden/plant-finder/plant-details/kc/u310/juncus-inflexus-afro.aspx>. Accessed on 26 July 2012.
- Missouri Botanical Garden. 2013. *Mentha spicata*. Available <http://www.missouribotanicalgarden.org/gardens-gardening/your-garden/plant-finder/plant-details/kc/a244/mentha-spicata-kentucky-colonel.aspx>. Accessed 15 November 2013.
- Mitchell, A., and A. Darwish. 2009. Efficacy of 6-, 12-, and 24-h praziquantel bath treatments against Asian Tapeworms *Bothriocephalus acheilognathi* in grass carp. North American Journal of Aquaculture 71: 30-34.
- Mitchell, A.J., and G.L. Hoffman. 1980. Important tapeworms of North American freshwater fishes. United States Department of the Interior, Fish & Wildlife Service. Fish Disease Leaflet 59, 17 pp.
- Molloy, D.P. 1998. The potential of using biological control technologies in the management of Dreissena. Abstracts from the Eighth International Zebra Mussel and Other Nuisance Species Conference. California Sea Grant, Sacramento, California: 69-70.
- Molloy, D.P. 2002. Biological control of Zebra Mussels. Proceedings of the Third California Conference on Biological Control. University of California, Davis.
- Monsanto Company. 2007. Roundup PRO Concentrate: Supplemental Labeling. Monsanto Company, St. Louis, Missouri.
- Morgan, D., S. Beatty, and H. McLetchie. 2005. Control of feral Goldfish (*Carassius auratus*) in the Vasse River. Murdoch University. Available http://researchrepository.murdoch.edu.au/5948/1/Feral_Goldfish.pdf.

- Morrison, W.E., and M.E. Hay. 2011. Induced chemical defenses in a freshwater macrophyte suppress herbivore fitness and the growth of associated microbes. *Oecologia* 165: 427-436.
- Morton, D., and Z. Reicher. 2007. Controlling *Poa trivialis* in cool-season fairways. *Golf Course Management* 99.
- Nalepa, T.F., D.J. Hartson, D.L. Fanslow, G.A. Lang, and S.J. Lozano. 1998. Declines in Benthic Macroinvertebrate Populations in Southern Lake Michigan, 1980-1993. *Canadian Journal of Fisheries and Aquatic Sciences* 55(11): 2402-13.
- National Invasive Species Act (NISA). 1996. Public Law 104-332 (Oct 26, 1996).
- Nauwerck, A. 1991. The history of the genus *Eubosmina* in Lake Mondsee (upper Austria). *Hydrobiologia* 225: 87-103.
- NatureServe. 2008. NatureServe Explorer: An online encyclopedia of life [web application]. Version 7.1. NatureServe, Arlington, Virginia. Available <http://www.natureserve.org/explorer>. Accessed 9 June 2011.
- Naylor, M. 2003. Water Chestnut (*Trapa natans*) in the Chesapeake Bay Watershed: A Regional Management Plan. Maryland Department of Natural Resources. 35 pp.
- Newroth, P.R. 1988. Review of current aquatic plant management activities in British Columbia. Pages 66-71. *in* Proceedings of 22nd Annual meeting, Aquatic Plant Control Program. Miscellaneous Paper A-88-5, Environmental Laboratory, Waterways Experiment Station, Vicksburg, Mississippi.
- Nonindigenous Aquatic Nuisance Prevention and Control Act of 1990 (NASPCA). 2000. Amended through 29 December 2000. Available <http://www.anstaskforce.gov/Documents/nanpca90.pdf>. Accessed 26 July 2012.
- North Carolina State (NC State). 2012. Yellow fieldcress. Available http://www.cals.ncsu.edu/plantbiology/ncsc/containerWeeds/Rorippa_sylvestris.htm. Accessed 9 December 2013.
- North Central Regional Aquaculture Center (NCRAC). 2010a. Michigan - Aquaculture Contacts, Transport Regulations, and Approved Aquatic Species. Available <http://www.ncrac.org/Info/StateImportRegs/michigan.htm#sales>. Accessed 18 June 2012.
- North Central Regional Aquaculture Center (NCRAC). 2010b. State import regulations. Available <http://www.ncrac.org/node/378>. Accessed 18 June 2012.

Neilson, K., R. Kelleher, G. Barnes, D. Speirs, and J. Kelly. 2004. Use of fine-mesh monofilament gill nets for the removal of Rudd (*Scardinius erythrophthalmus*) from a small lake complex in Waikato, New Zealand. *New Zealand Journal of Marine and Freshwater Research* 38(3): 525-539.

New York Invasive Species Council (NYISC). 2010. Final report: a regulatory system for non-native species. Department of Environmental Conservation, Albany, NY. 131 pp.

New York State Department of Environmental Conservation (NYSDEC). 2011. Fish stocking Lists (2011) by County. NYSDEC Web Site. Available <http://www.dec.ny.gov/outdoor/7739.html>. Accessed 23 May 2012.

New York State Department of Environmental Conservation (NYSDEC). 2012a. Fish health regulations in response to VHS: summary of fish health regulations. NYSDEC Web Site. Available <http://www.dec.ny.gov/outdoor/33072.html>. Accessed 23 May 2012.

New York State Department of Environmental Conservation (NYSDEC). 2012b. NY River Herring management. NYSDEC Web Site. Available <http://www.dec.ny.gov/animals/76525.html>. Accessed 29 July 2012.

New York State Department of Environmental Conservation (NYSDEC). 2013. Triploid Grass Carp in New York Ponds. NYSDEC Web Site. Available <http://www.dec.ny.gov/outdoor/7973.html>. Accessed 23 May 2012.

Nordin, L.J., M.T. Arts, O.E. Johannsson, and W.D. Taylor. 2008. An evaluation of the diet of *Mysis relicata* using gut contents and fatty acid profiles in lakes with and without the invader *Bythotrephes longimanus* (Onychopoda, Cercopagidae). *Aquatic Ecology* 42: 421-436.

Ntanos, D.A., S.F. Koutroubas, and C. Mavrotas. 2000. Barnyardgrass (*Echinochloa crus-galli*) control in water-seeded rice (*Orzya sativa*) with cyhalofop-butyl. *Weed Technology* 14(2): 383-388.

Office of Water Resources, Rhode Island Department of Environmental Management. 2010. Freshwater Aquatic Invasive Species in Rhode Island: Sping Naiad. 2 pp.

Ogle, D.H., J.H. Selgeby, R.M. Newman, and M.G. Henry. 1995. Diet and feeding periodicity of Ruffe in the St. Louis River estuary, Lake Superior. *Transactions of the American Fisheries Society* 124: 356-369.

Ogle, D.H. 1998. A synopsis of the biology and life history of Ruffe. *Journal of Great Lakes Research* 24(2): 170-185.

Ohio Division of Natural Areas and Preserves, and The Nature Conservancy. 2000. Ohio's Invasive Plant Species. Fact Sheet, Ohio Division of Natural Areas and Preserves, Columbus, Ohio, 2 pp.

- Ohio Environmental Protection Agency (Ohio EPA). 2001. Invasive Plants of Ohio: Glossy Buckthorn and Common Buckthorn. Fact Sheet, 2pp.
- Ontario Federation of Anglers and Hunters (OFAH). N.d. Ontario's Invasive Species Awareness Program. Available: <http://www.invadingspecies.com/>.
- Ohio Sea Grant. 1992. Boaters: Take action against Zebra Mussels. OHSU-FS-054.
- Ohio State University (OSU). 2005. Meadow Foxtail, *Alopecurus pratensis* L. Ohio State Rangeland Ecology and Management. Columbus, Ohio. 2 pp.
- Ontario Ministry of Natural Resources (OMNR). 2011. Summary of current control measures to slow the spread of VHS in Ontario. Available <http://www.mnr.gov.on.ca/en/Business/LetsFish/2ColumnSubPage/200524.html>. Accessed 23 May 2012.
- Ontario Ministry of Natural Resources (OMNR). 2012. Fish farmers can help slow the spread of fish disease viral hemorrhagic septicemia (VHS). Available http://www.mnr.gov.on.ca/stdprodconsume/groups/lr/@mnr/@letsfish/documents/document/mnr_e001347.pdf. Accessed 15 June 2012.
- Open Learning Agency (OLA) and British Columbia Ministry of Agriculture, Food and Fisheries (MAFF). 2002. Guide to weeds in British Columbia. Open Learning Agency, Burnaby, British Columbia, Canada. 195 pp. Available <http://www.al.gov.bc.ca/weedsbc/pdf/GuidetoWeeds.pdf>.
- Oppegaard, H., and H. Sörum. 1994. gyrA Mutations in quinolone-resistant isolates of the fish pathogen *Aeromonas salmonicida*. Antimicrobial Agents and Chemotherapy 38: 2460-2464.
- Oregon Department of Agriculture (ODA) Plant Division. 2011. *Cheilosia corydon*. Noxious Weed Control. Available http://www.oregon.gov/ODA/PLANT/WEEDS/bio_profile_chco.shtml. Accessed 21 May 2012.
- Pacific Northwest Extension. 2013. Dock (*Rumex* spp.). Available <http://pnwhandbooks.org/weed/other-items/control-problem-weeds/dock-rumex-spp>. Accessed 9 December 2013.
- Paperna, I. 1996. Parasites, infections and diseases of fishes in Africa: an update. Food and Agriculture Organization of the United Nations. CIFA Technical Paper, 31. 204 pp.
- Pardue, W.J., and T.S. Wood. 1980. Baseline toxicity data for freshwater bryozoa exposed to copper, cadmium, chromium, and zinc. Journal of the Tennessee Academy of Science 55(1): 27-31.
- Parkinson, H., J. Mangold, V. Dupuis, and P. Rice. 2010. Biology, Ecology and Management of Flowering Rush (*Butomus umbellatus*). The U.S. Department of Agriculture, Montana State University Extension. Bozeman, Montana. 12 pp.

- Parsons, W.T. 1973. Noxious weeds of Victoria. Inkata Press Ltd., Melbourne, Australia.
- Pascho, R.J., D.G. Elliott, and J.M. Streufert. 1991. Brood stock segregation of spring Chinook Salmon *Oncorhynchus tshawytscha* by use of the enzyme-linked immunosorbent assay (ELISA) and the fluorescent antibody technique (FAT) affects the prevalence and levels of *Renibacterium salmoninarum* infection in progeny. Diseases of Aquatic Organisms 12: 25-40.
- Pascho, R.J., M.L. Landolt, and J.E. Ongerth. 1995. Inactivation of *Renibacterium salmoninarum* by free chlorine. Aquaculture 131: 165-175.
- Patrick, P.H., A.E. Christie, D. Sager, C. Hocutt, and J. Stauffer Jr. 1985. Responses of fish to a strobe light/air-bubble barrier. Fisheries Research 3: 157-172.
- Paukert, C.P., W. Stancill, T.J. DeBates, and D.W. Willis. 2003. Predatory effects of Northern Pike and largemouth bass: bioenergetic modeling and ten years of fish community sampling. Journal of Freshwater Ecology 18: 13-24.
- Peard, T. 2002. Freshwater jellyfish! Indiana University of Pennsylvania, Indiana, PA. Available <http://www.jellyfish.iup.edu/>.
- Pennsylvania Department of Conservation and Natural Resources (PADCNR). n.d. Invasive Plants in Pennsylvania Factsheets. Harrisburg, PA.
- Peters, W.L., M. Hockenberry Meyer, and N.O. Anderson. 2006. Minnesota horticultural industry survey on invasive plants. Euphytica 148: 75-86.
- Pham, L., D. West, G.P. Closs. 2013. Reintroduction of a native galaxiid (*Galaxias fasciatus*) following piscicide treatment in two streams: response and recovery of the fish population. Ecology of Freshwater Fish 22(3): 361-373.
- Pinto, L., N. Chandrasena, J. Pera, P. Hawkins, D. Eccles, and R. Sim. 2005. Managing invasive carp (*Cyprinus carpio* L.) for habitat enhancement at Botany Wetlands, Australia. Aquatic Conservation: Marine and Freshwater Ecosystems 15: 447-462.
- Piola, R.F., and E.L. Johnston. 2006. Differential Tolerance to Metals among Populations of the Introduced Bryozoan *Bugula neritina*. Marine Biology 148(5): 997-1010.
- Piper, G.L. 1997. Purple Loosestrife Biological Control Agent Propagation and Release in Washington. Final Report WSDA Contract # 1A97-7-5 WSU Contract # 60631, October 21, 1997. 9 pp.
- Pitcher, D. 1985. *Conium maculatum*. Bugwood Wiki. Available http://wiki.bugwood.org/Conium_maculatum. Accessed 22 June 2012.

- Pitcher, D. 2004. Element Stewardship Abstract for *Conium maculatum* L. The Nature Conservancy, Arlington, VA. Available <http://www.imapinvasives.org/GIST/ESA/esapages/documnts/conimac.pdf>.
- Pittaway, A.R. 2012. Deilephila [Laspeyres], 1809. Sphingidae of the Western Palaearctic. Available <http://tpittaway.tripod.com/sphinx/list.htm>. Accessed 23 May 2012.
- Plantwise. 2013. Plantwise Knowledge Bank Technical Factsheet: Redshank. Available <http://www.plantwise.org/KnowledgeBank/Datasheet.aspx?dsid=42691>. Accessed 9 December 2013.
- Poovaliah, C.R., S.C. Weller, and M.A. Jenks. 2006. Adventitious shoot regeneration of scotch Spearmint (*Mentha x gracilis sole*). Vitro Cellular & Developmental Biology 42(4): 354-358.
- Popay, I., and R. Field. 1996. Grazing animals as weed control agents. Weed Technology 10(1): 217-231.
- Post, J.R., and D.J. McQueen. 1987. The impact of planktivorous fish on the structure of a plankton community. Freshwater Biology 17: 79-89.
- Pothoven, S.A., H.A. Vanderploeg, J.F. Cavaletto, D.M. Krueger, D.M. Mason, and S.B. Brandt. 2007. Alewife planktivory controls the abundance of two invasive predatory cladocerans in Lake Michigan. Freshwater Biology 52(3): 561-573.
- Pothoven, S.A., H.A. Vanderploeg, S.A. Ludsin, T.O. Hook, and S.B. Brandt. 2009. Feeding Ecology of Emerald Shiners and Rainbow Smelt in Central Lake Erie. Journal of Great Lakes Research 35(2): 190-198.
- Post, G. 1983. Textbook of Fish Health. T. F. H. Publications, Inc. Ltd., The British Crown Company of Hong Kong. 256 pp.
- Prather, T., T. Miller, and A. Hulting. 2014. Control of problem weeds: hemlock, poison (*Conium maculatum*). Pacific Northwest Weed Management Handbook. Pacific Northwest Extension, Oregon State University, Corvallis, Oregon. Available <http://pnwhandbooks.org/weed/sites/default/files/chapters/pdf/w-problemweeds.pdf>.
- Pullman, G., and G. Crawford. 2010. A Decade of Starry Stonewort in Michigan. Lakeline. pp. 36-42.
- Rach, J.J., J.A. Luoma, and L.L. Marking. 1994. Development of an antimycin-impregnated bait for controlling Common Carp. North American Journal of Fisheries Management 14: 442-446.
- Reed, P., R. Francis-Floyd, and R. Klinger. 1996. Monogenean parasites of fish. Document FA28 University of Florida Institute of Food and Agricultural Sciences, Gainesville, Florida. 4pp.

Rees, N.E., P.C. Quimby Jr., G.L. Piper, E.M. Coombs, C.E. Turner, N.R. Spencer, and L.V. Knutson. 1996. The Biological Control of Weeds in the West. Western Society of Weed Science, Bozeman, Montana.

Relyea, R.A. 2005. The impact of insecticides and herbicides on the biodiversity and productivity of aquatic communities. *Ecological Applications* 15(2): 618-627.

Rhode Island Coastal Resources Management Council (RICRMC). 2007. Rhode Island Aquatic Invasive Species Management Plan. University of Rhode Island; Rhode Island Natural History Survey. 139 pp.

Ricciardi, A. 2001. Facilitative interaction among aquatic invaders: is an “invasional meltdown” occurring in the Great Lakes? *Canadian Journal of Fisheries and Aquatic Sciences* 58(12): 2513-2525.

Ricciardi, A. 2006. Patterns of invasion in the Laurentian Great Lakes in relation to changes in vector activity. *Diversity and Distributions* 12: 425–433.

Richards, D.C., P. O'Connell, and D.C. Shinn. 2004. Simple control method to limit the spread of the New Zealand Mudsnail *Potamopyrgus antipodarum*. *North American Journal of Fisheries Management* 24: 114-117.

Ringelband, U., and L. Karbe. Effects of Vanadium on Population Growth and Na-K-ATPase Activity of the Brackish Water Hydroid *Cordylophora caspia*. *Bulletin of Environmental Contamination and Toxicology* 57: 118-24.

Ritz, A.W. 1987. Commercial fishing for carp. Pages 17-30 in Cooper, E.L., ed. *Carp in North America*. American Fisheries Society, Bethesda, Maryland.

Riutta, T., J. Laine, and E-S. Tuittila. 2007. Sensitivity of CO₂ exchange of fen ecosystem components to water level variation. *Ecosystems* 10: 718-733.

Rivera, C.J.R. 2008. Obstruction of the upstream migration of the invasive snail *Cipangopaludina chinensis* by high water currents. Unpublished practicum. Available <http://www3.nd.edu/~underc/east/education/documents/Rivera2008.pdf>. Accessed 28 October 2013.

Roberts, H. 1986. Persistence of seeds of some grass species in cultivated soil. *Grass and Forage Science* 41(3): 273-276.

Robert W. Freckmann Herbarium. 2012. Invasive/Eradication Information. University of Wisconsin- Stevens Point. Available <http://wisplants.uwsp.edu/stopit.html>. Accessed 22 June 2012.

- Robinson, M. 2004. European Naiad: An Invasive Aquatic Plant (*Najas minor*). Massachusetts Department of Conservation and Recreation; Office of Water Resources; Lakes and Ponds Program. 4 pp.
- Rollo, A., G. Andraso, J. Janssen, and D. Higgs. 2007. Attraction and localization of Round Goby (*Neogobius melanostomus*) to conspecific calls. *Behaviour* 144:1-21.
- Roman, K. 2007. Exotic Buckthorns: Common, Glossy, Dahurian, Japanese, and Chinese Buckthorn (*Rhamnus cathartica* L., *Frangula alnus* Mill. syn. *R. frangula* L., *R. davurica* Pall., *R. japonica* Maxim., and *R. utilis* Decne.). Illinois Nature Preserves Commission 1(5): 9 pp.
- Rowland, S.J., M. Landos, R.B. Callinan, G.L. Allan, P. Read, C. Mifsud, M. Nixon, P. Boyd, and P. Tully. 2007. Development of a Health Management Strategy for the Silver Perch Aquaculture Industry. Fisheries Final Report Series No. 93. NSW Department of Primary Industries, Grafton, New South Wales. 221pp.
- Saha, N., F. Bhunia, and A. Kaviraj. 2006. Comparative toxicity of three organic acids to freshwater organisms and their impact on aquatic ecosystems. *Human and Ecological Risk Assessment* 12(1): 192-202.
- Sager, L., and C. Clerc. 2006. Factors influencing the distribution of *Hydrocharis morsus-ranae* L. and *Rumex hydrolapathum* Huds. in a mowed low-lying marshland, Reserve de Cheyres, lac de Neuchatel, Switzerland. *Hydrobiologia* 570: 223-229.
- Santagata, S., Z.R. Gasiunaite, E. Verling, J.R. Cordell, K. Eason, J.S. Cohen, K. Bacela, G. Quilez-Badia, T.H. Johengen, D.F. Reid, and G.M. Ruiz. 2008. Effect of osmotic shock as a management strategy to reduce transfers of non-indigenous species among low-salinity ports by ships. *Aquatic Invasions* 3(1): 61-76.
- Sarig, S., M. Lahav, and M. Child. 1965. Control of *Dactylogyrus vastator* on carp fingerlings with dipterex. *The Israeli Journal of Aquaculture* 14: 47-52.
- Sauter, G., and H. Gude. 1996. Influence of grain size on the distribution of tubificid oligochaete species. *Hydrobiologia* 334: 97-101.
- Savino, J.F., D.J. Jude, and M.J. Kostich. 2001. Use of Electrical Barriers to Deter Movement of Round Goby. *Behavioral Technologies for Fish Guidance: American Fisheries Society Symposium*, pp 171-182.
- Savitz, J.D., and D.A. Wright. 1994. Toxic effects of the insecticide diflubenzuron (Dimilin®) on survival and development of nauplii of the estuarine copepod, *Eurytemora affinis*. *Marine Environmental Research* 37(3): 297-312.

Schmahl, G., and H. Mehlhorn. 1985. Treatment of fish parasites. 1 Praziquantel effective against Monogenea (*Dactylogyrus vastator*, *Dactylogyrus extensus*, *Diplozoorn paradoxum*). Zeitschrift für Parasitenkunde 7: 727-737.

Schubiger, F.X., G. Défago, H. Kern, and L. Sedlar. 1986. Damage to *Rumex crispus* L. and *Rumex obtusifolius* L. caused by the rust fungus *Uromyces rumicis* (Schum.). Winter Weed Research 26: 347-50.

Schütz, W. 1998. Seed dormancy cycles and germination phonologies in sedges (*Carex*) from various habitats. Wetlands 18(2): 288-297.

Scott, W.B., and E.J. Crossman. 1973. Freshwater Fishes of Canada. Fisheries Research Board of Canada Bulletin 184. 966 pp.

Setre Chemical Company. 1986. Setre Prowl® Herbicide + Propanil. Available http://www.epa.gov/pesticides/chem_search/ppls/038167-00010-19860306.pdf.

Shaner, D. 2006. An Overview of Glyphosate mode of action: Why is it such a great herbicide? North Central Weed Science Society Proceedings 61:94.

Sheath, R.G., and K.M. Cole. 1984. Systematics of *Bangia Rhodophyta* in North America. Biogeographic trends in morphology. Phycologia 23(3): 383-396.

Michigan Sea Grant. 1999. The Purple Loosestrife Project: Rearing and Releasing Natural Enemies to Control Purple Loosestrife. Michigan Sea Grant College Program.

Sheehan, M. 2007. Wisconsin Impact Assessment: *Cirsium palustre* (L.) Scop.

Sheldon, S.P. 1994. Invasions and declines of submersed macrophytes in New England, with particular reference to Vermont Lakes and herbivorous invertebrates in New England. Lake and Reservoir Management 10(1): 13-17.

Sheldon, S.P., and K.N. Jones. 2001. Restricted gene flow according to host plant in an herbivore feeding on native and exotic watermilfoils (*Myriophyllum* : *Haloragaceae*). International Journal of Plant Sciences 162(4): 793-799.

Sheppard, A.W., R.H. Shaw, and E.R. Sforza. 2006. Top 20 environmental weeds for classical biological control in Europe: a review of opportunities, regulations and other barriers to adoption. Weed Research 46: 93-117.

Available <http://onlinelibrary.wiley.com/doi/10.1111/j.1365-3180.2006.00497.x/pdf>

Shiels, A.L., and M.A. Hartle. 2012. Pond Owner's Guide to the Use of Triploid Grass Carp in Pennsylvania. Pennsylvania Fish & Boat Commission. Available http://fishandboat.com/images/pages/forms/pfbc_tgc002.pdf. Accessed 16 April 2013.

- Simpson, K.W., and L.E. Abele. 1984. *Ripistes parasita* (Schmidt) (Oligochaeta: Naididae), a distinctive oligochaete new to North America. *Freshwater Invertebrate Biology* 3(1): 36-41.
- Simon, B. 2008. Aquatic Weeds Management Fund Final Report: Yellow-flag Iris Control and Education. Washington State Department of Agriculture. Available <http://www.ecy.wa.gov/programs/wq/plants/weeds/YFI%20Final%20Report%20to%20DOE%2006-30-08.pdf>.
- Singhal, R.N., S. Jeet, and R.W. Davies. 1986. Chemotherapy of six ectoparasitic diseases of cultured fish. *Aquaculture* 54(3): 165-171.
- Siwicki, A.K., F. Pozet, M. Morand, B. Kazun, S. Trapkowska, and J. Malaczewska. 2003. Influence of methisoprinol on the replication of rhabdoviruses isolated from carp (*Cyprinus carpio*) and catfish (*Ictalurus melas*): in vitro study. *Polish Journal of Veterinary Science* 6: 47-50.
- Skotland, C.B. 1979. Rhizome rot of scotch Spearmint *Mentha cardica* caused by a sclerotium forming basidiomycete. *Phytopathology* 69: 920.
- Skotland, C.B., and J.A. Traquair. 1982. A dormant rhizome rot of scotch Spearmint *Mentha cardiac*, caused by *Rhizoctonia solani*. *Phytopathology* 72: 994.
- Smith, D.G., S.F. Werle, and E. Klekowski. 2002. The rapid colonization and emerging biology of *Cordylophora caspia* (Pallas, 1771) (Cnidaria: Clavidae) in the Connecticut River. *Journal of Freshwater Ecology* 17(3): 423-430.
- Smith, A.S., and J.E. Alexander. 2008. Potential effects of the freshwater jellyfish *Craspedacusta sowerbii* on zooplankton community abundance. *Journal of Plankton Research* 30(12): 1323-1327.
- Smith, P., and S. Davey. 1993. Evidence for the competitive exclusion of *Aeromonas salmonicida* from fish with stress-inducible furunculosis by a fluorescent pseudomonad. *Journal of Fish Diseases* 16: 521-524.
- Smith, B.R., and J.J. Tibbles. 1980. Sea Lamprey (*Petromyzon marinus*) in Lakes Huron, Michigan, and Superior: history of invasion and control, 1936-78. *Canadian Journal of Fisheries and Aquatic Sciences* 37(11): 1780-1801.
- Snipes, C.E., and J.E. Street. 1987. Fenoxaprop for postemergence barnyardgrass (*Echinochloa crus-galli*) control in rice (*Oryza sativa*). *Weed Science* 35(2): 244-227.
- Snyder, F.L., M.B. Hilgendorf, and D.W. Garton. 1997. Zebra Mussels in North America: The invasion and its implications. Ohio Sea Grant, Ohio State University, Columbus, OH. Available <http://ohioseagrant.osu.edu/documents/publications/FS/FS-045%20Zebra%20mussels%20in%20North%20America.pdf>.

- Solarz, S.L., and R.M. Newman. 1996. Oviposition specificity and behavior of the watermilfoil specialist *Euhrychiopsis lecontei*. *Oecologia* 106: 337-344.
- Sorensen, P.W., and N.E. Stacey. 2004. Brief review of fish pheromones and discussion of their possible uses in the control of non-indigenous teleost fishes. *New Zealand Journal of Marine and Freshwater Research* 38: 399-417.
- Spadinger, R., and G. Maier. 1999. Prey selection and diel feeding of the Freshwater Jellyfish, *Craspedacusta sowerbyi*. *Freshwater Biology* 41: 567-573.
- Spencer, D.R., and P.L. Hudson. 2003. The Oligochaeta (Annelida, Clitellata) of the St. Lawrence Great Lakes Region: an Update. *Journal of Great Lakes Research* 29(1): 89-104.
- Sprecher, S.L., and K.D. Getsinger. 2000. Zebra Mussel chemical control guide. United States Army Corps of Engineers, Engineer Research Development Center.
- Minnesota Department of Natural Resources (MNDNR). 2009. Buckthorn: What You Should Know. What You Can Do. St. Paul, MN. 4 pp.
- Stegmaier Jr., C.E. 1967. *Pluchea odorata*, a New Host Record for *Acinia picturata* (Diptera, Tephritidae). *The Florida Entomologist* 50(1): 53-55.
- Stewart, T.W. 2008. Benthic algae of Lake Erie (1865-2006): A review of assemblage, composition, ecology, and causes and consequences of changing abundance. *Ohio Journal of Science* 108(5): 92-94.
- Stevens, M., and S. Hoag. 2003. Baltic Rush *Juncus balticus* Wild. Plant Guide. United States Department of Agriculture (USDA) Natural Resources Conservation Service (NRCS). 5 pp.
- Stevens, M., and C. Hoag. 2006. Narrowleaf Cattail, *Typha angustifolia* L. Plant Guide. United States Department of Agriculture (USDA) Natural Resources Conservation Service (NRCS). 4 pp.
- Stewart, D.J., J.F. Kitchell, and L.B. Crowder. 1981. Forage fishes and their salmonid predators in Lake Michigan. *Transactions of the American Fisheries Society* 110(6): 751-763.
- Stewart, T.W., and J.M. Haynes. 1994. Benthic Macroinvertebrate Communities of Southwestern Lake Ontario Following Invasion of Dreissena. *Journal of Great Lakes Research* 20(2): 479-493.
- Stoermer, E.F. 1978. Phytoplankton assemblages as indicators of water quality in the Laurentian Great Lakes. *Transactions of the American Microscopical Society* 97: 2-16.
- Stoermer, E.F., and J.J. Yang. 1969. Plankton diatom assemblages in Lake Michigan. Great Lakes Research Division, University of Michigan, Special Report No. 47.

Stoffregen, D.A., A.J. Chako, S. Backman, and J.G. Babish. 1993. Successful therapy of furunculosis in Atlantic salmon, *Salmo salar* L., using the fluoroquinolone antimicrobial agent enrofloxacin. *Journal of Fish Diseases* 16: 219-228.

Street, J., and P. Shettatt. 2013. *Poa* factsheet. OSU Buckeye Turf Program. Available http://buckeyeturf.osu.edu/index.php?option=com_content&id=1192&Itemid=170. Accessed 6 December 2013.

Stromberg, K. 2006. Developments in Whirling Disease Research through the National Symposium. *Aquaculture Health International*, May 2006, pp31-33. Available http://whirlingdisease.montana.edu/biblio/pdfs/WDSymposiumArticle_AHI-May06.pdf. Accessed 11 May 2012.

Straile, D., and A. Halbach. 2000. Life history and multiple antipredator defenses of an invertebrate pelagic predator, *Bythotrephes longimanus*. *Ecology* 81: 150-163.

Stuart, I.G., A. Williams, J. McKenzie, and T. Holt. 2011. Managing a migratory pest species: a selective trap for Common Carp. *North American Journal of Fisheries Management*. Available <http://www.tandfonline.com/doi/pdf/10.1577/M05-205.1>. Accessed 1 November 2013.

Sturtevant, R., J. Larson, L. Berent, M. McCarthy, A. Bogdanoff, A. Fusaro, and E. Rutherford. 2014. An impact assessment of Great Lakes aquatic nonindigenous species. NOAA Technical Memorandum GLERL- 161. NOAA Great Lakes Environmental Research Laboratory, Ann Arbor, MI, 1015 pp. Available http://www.glerl.noaa.gov/ftp/publications/tech_reports/glerl-161/tm-161.pdf.

Suchy, K.D., and B.J. Hann. 2007. Using microfossil remains in lake sediments to examine the invasion of *Eubosmina coregoni* (Cladocera, Bosminidae) in Lake of the Woods, Ontario, Canada. *Journal of Great Lakes Research* 33(4): 867-874.

Summerfelt, R.C. 1999. Lake and reservoir management. Pages 285-320 in C.C. Kolhler and W. A. Hubert, editors. *Inland Fisheries Management in North America*. American Fisheries Society, Bethesda, Maryland.

Sunblad, K., and K. Robertson. 1988. Harvesting reed sweetgrass (*Glyceria maxima*, Poacea): Effects on growth and rhizome storage of carbohydrates. *Economic Botany* 42(2): 495-502.

Sutherland, D., S. Marcquenski, J. Marcino, P. Stelzig, J. Lom, I. Dykova, F. Nilsen, S. Cooper, H.M. Hsu, W. Jahns, J. Hoyle, and R. Penny. 2006. *Heterosporis* sp. (Microspora: Pleistophoridae): a parasite from *Perca flavescens*, *Stizostedion vitreum* and *Esox lucius* in Minnesota, Wisconsin and Lake Ontario. Available <http://www.glfc.org/boardcomm/fhealth/GLFHC2006FebMinutes.pdf>. Accessed 23 July 2012.

Sutherland, W.J. 1990. *Iris pseudacorus* L. *Journal of Ecology* 78(3): 833-848.

- Swearingen, J., K. Reshetiloff, B. Slattery, and S. Zwicker. 2002. Plant Invaders of Mid-Atlantic Natural Areas 82. pp National Parks Service and U.S. Fish & Wildlife Service.
Available <http://www.invasive.org/eastern/midatlantic/mysp.html>. Accessed 25 April 2013.
- Syska, S., T. Crawford, and S. Ramsey-Westbrook. 2012. Largemouth bass fact sheet-LMBV. Missouri Department of Conservation. Available <http://mdc.mo.gov/landwater-care/animal-management/animal-diseases/largemouth-bass-fact-sheet-lmbv>. Accessed 15 June 2012.
- Tanner, R.A. 2011. An Ecological Assessment of *Impatiens glandulifera* in its Introduced and Native range and the Potential for its Classical Biological Control. Thesis for Doctorate of Philosophy. School of Biological Sciences, Royal Holloway, University of London. Egham, Surrey United Kingdom. 285 pp.
- Tarver, D.P., J.A. Rogers, M.J. Mahler, and R.L. Lazor. 1986. Aquatic and Wetland Plants of Florida. Third Edition. Florida Department of Natural Resources, Tallahassee, Florida.
- Taylor, A.H., S.R. Tracey, K. Hartmann, and J.G. Patil. 2012 Exploiting seasonal habitat use of the Common Carp, *Cyprinus carpio*, in a lacustrine system for management and eradication. Marine and Freshwater Research 63: 587-597.
- Taylor, N.G.H., C. Sommerville, and R. Wootten. 2006. The epidemiology of *Argulus* spp. (Crustacea: Branchiura) infections in stillwater trout fisheries. Journal of Fish Diseases 29(4): 193-200.
- Terrestrial Herbaceous Plants Species Assessment Group. 2007. Assessment Summary for European Marsh Thistle.
- The Nature Conservancy of Vermont (TNC Vermont). 1998. Vermont Invasive Exotic Plant Fact Sheet: Eurasian Watermilfoil.
- The Nature Conservancy. n.d. Yellow-Flag Iris. Invasive fact sheet. Montpelier, VT. 2 pp.
Available <http://www.nature.org/ourinitiatives/regions/northamerica/unitedstates/vermont/yellowiris2010-2.pdf>.
- Thomas, M.V., and M. Faisal. 2009. *Piscirickettsia* infection in the muskellunge population of Lake St. Clair. Michigan Department of Natural Resources, Fisheries Research Report, 2092, Ann Arbor.
- Thompson, A.L., and C.S. Luthin. Chapter 6: Invasive Species and Wetland Management. Pages 58-74. in Wisconsin Department of Natural Resources. Wetland Restoration Handbook for Wisconsin Landowners (2nd Edition).

Thorsness, K.B., C.G. Messersmith, and R.G. Lym. 1992. Evaluation of Rodeo™ (glyphosate) efficacy for cattail management. Cattail Management Symposium. U.S. Dept. of Agriculture, Animal and Plant Health Inspection Service, Animal Damage Control, Denver Wildlife Research Center, U.S. Dept. of Interior, Fish and Wildlife Service. Jamestown, ND.

Tilley, D., D. Ogle, and L. St. John. 2010. Plant guide for Redtop (*Agrostis gigantea*). USDA-Natural Resources Conservation Service, Idaho Plant Materials Center. Aberdeen, ID. Available <ftp://ftp-fc.sc.egov.usda.gov/ID/programs/plant/redtop.pdf>. Accessed 12 January 2012.

Todar, K. 2008. Antimicrobial agents used in treatment of infectious diseases. Todar's online textbook of bacteriology. Available <http://textbookofbacteriology.net/antimicrobial.html>. Accessed 30 January 2008.

Tody, W.H., and H.A. Tanner. 1966. Coho Salmon for the Great Lakes. Michigan Department of Natural Resources, Fisheries Division, Fish Management Report 1, Lansing.

Toogood, S.E., C.B. Joyce, and S. Waite. 2008. Response of floodplain grassland plant communities to altered water regimes. *Plant Ecology* 197(2): 285—298.

Tonguthai, K. 1997. Control of freshwater fish parasites: a Southeast Asian perspective. *International Journal for Parasitology* 27(10): 1185-1191.

Torezo, A.E., B. Magariños, and J.L. Romalde. 2005. A review of the main bacterial fish diseases in mariculture systems. *Aquaculture* 246(1-4): 37-61.

Townshend, J.L., and T.R. Davidson. 1962. Some weed hosts of northern root-knot nematode, *Meloidogyne hapla* Chitwood, 1949, in Ontario. *Canadian Journal of Botany* 40: 543-548.

Treasurer, J.W. 1992. The predator-prey relationship of perch *Perca fluviatilis* larvae and zooplankton in two Scottish lochs. *Environmental Biology of Fishes* 35(1): 63-74.

Tsoumas, A., D.J. Alderman, and C.J. Rodgers. 1989. *Aeromonas salmonicida*: development of resistance to 4-quinolone antimicrobials. *Journal of Fish Diseases* 12: 493-507.

Tu, M. 2003. Element Stewardship Abstract for *Iris pseudacorus* L. (yellow flag iris, water flag). The Nature Conservancy. Arlington, Virginia. 11 pp.

Tu, M., C. Hurd, J.M. Randall, and the Nature Conservancy. 2001. Weed Control Methods Handbook: Tools & Techniques for Use in Natural Areas. All Government Documents (Utah Regional Depository). Paper 533.

University of California Davis Tahoe Environmental Research Center (UC Davis TERC). 2004. Aquatic Invasive Species. Available <http://terc.ucdavis.edu/research/aquaticinvasives.html>. Accessed 28 October 2013.

United States Army Corps of Engineers (USACE). 2002. Zebra Mussel Information System. Available <http://el.erdc.usace.army.mil/zebra/zmis/>. Accessed 26 July 2012.

United States Army Corps of Engineers (USACE). 2012a. Molluscicides. Available <http://glmr.is.anl.gov/documents/docs/anscontrol/Molluscicides.pdf>. Accessed 29 October 2013.

United States Army Corps of Engineers (USACE). 2012b. Great Lakes and Mississippi River Interbasin Study: Inventory of Available Controls for Aquatic Nuisance Species of Concern, Chicago Area Waterway System. United States Army Corps of Engineers.

United States Army Corps of Engineers (USACE). 2013. Piscicides. Great Lakes and Mississippi River Interbasin Study, Argonne National Laboratory. January 2013.

United States Department of Agriculture (USDA) Animal and Plant Health Inspection Service (APHIS). 2008. Rules and Regulations: Viral Hemorrhagic Septicemia; Interstate Movement and Import Restrictions on Certain Live Fish. Federal Register 73(175): 52173-52189.

United States Department of Agriculture (USDA) Animal and Plant Health Inspection Service (APHIS). 2012. Import and export. Available: http://www.aphis.usda.gov/import_export/animals/animal_import/marine_import_fish.shtml. Accessed 5 July 2012.

United States Department of Agriculture (USDA) Forest Service. 2005a. Environmental Assessment: Chequamegon-Nicolet Invasive Plant Control Project, July 2005. Available: http://www.fs.fed.us/r9/cnnf/natres/eis/so/nnis_ea/index.html.

United States Department of Agriculture (USDA) Forest Service. 2005b. Weed of the Week: Marsh Thistle, *Cirsium palustre* (L.) Scop. Available http://www.na.fs.fed.us/fhp/invasive_plants/weeds/marsh-thistle.pdf. Accessed 14 May 2012.

United States Department of Agriculture (USDA) Forest Service. 2012. *Typha angustifolia*. Index of Species Information. Available <http://www.fs.fed.us/database/feis/plants/graminoid/typang/all.html>. Accessed 19 September 2012.

United States Department of Agriculture (USDA) Natural Resources Conservation Service (NRCS). 2002. Plant Fact Sheet: Redtop *Agrostis gigantea* Roth. Available http://plants.usda.gov/factsheet/pdf/fs_aggi2.pdf. Accessed 14 May 2012.

United States Department of Agriculture (USDA) Natural Resources Conservation Service (NRCS). 2002. Plant Fact Sheet: *Purpleosier willow*.

United States Department of Agriculture (USDA) Natural Resources Conservation Service (NRCS). 2006. Plant fact sheet: European Black Alder, *Alnus glutinosa* L. Available <http://plants.usda.gov/java/factSheet>. Accessed 7 August 2012.

United States Department of Agriculture (USDA) Natural Resources Conservation Service (NRCS). 2007. Pest Management- Invasive Plant Control: Buckthorn (Common and Glossy). Available <http://www.wayzata.org/DocumentCenter/Home/View/477>. Accessed 7 August 2012.

United States Department of Agriculture (USDA) Natural Resources Conservation Service (NRCS). 2012. Fact Sheet Series.

United States Department of Homeland Security (DHS), Coast Guard. 2012. Standards for Living Organisms in Ships' Ballast Water Discharged in United States Waters. Federal Register 77(114): 35268.

United States Department of the Interior, Fish and Wildlife Service Region 3 Fisheries Program, and Great Lakes Fishery Commission (USFWS/GLFC). 2010. Great Lakes Fish Stocking database. Available <http://www.glfc.org/fishstocking/index.htm>.

United States Department of the Interior, National Park Service (NPS). 2011. Natural Resource Fact Sheet- Exotic Fish. National Park Service.

United States Environmental Protection Agency (US EPA). 2000. Significant Ongoing and Emerging Issues. Available <http://www.epa.gov/lakeerie/lamp2000/Section11.pdf>. 20 pp.

United States Government Printing Office. Electronic Code of Federal Regulations (e-CFR). Available <http://www.ecfr.gov/cgi-bin/ECFR?page=browse>.

Vacha F. (1998) Information on Czech Republic fisheries. Pages 48-57. in Hickley, P., and H. Tompkins, eds. Recreational Fisheries: Social, Economic and Management Aspects. Fishing News Books, Oxford, UK:.

van Leeuwen, B.H. 1983. The consequences of predation in the population biology of the monocarpic species *Cirsium palustre* and *Cirsium vulgare*. Oecologia 58: 178-187.

Verschuere, L., G. Rombaut, P. Sorgeloos, and W. Verstraete. 2000. Probiotic Bacteria as Biological Control Agents in Aquaculture. Microbiology and Molecular Biology Reviews 64(4): 655-671.

Viitasalo, S., and M. Viitasalo. 2004. Predation by the mysid shrimps *Mysis mixta* and *M. relicta* on benthic eggs of *Bosmina longispina maritima* (Cladocera) in the northern Baltic Sea. Marine Ecology Progress Series 281: 155-163.

Vincent, E.R. 2006. Are wild Rainbow Trout in the Madison River developing resistance to severe infections by the whirling disease parasite *Myxobolus cerebralis*? Proceedings from the 12th annual Whirling Disease Symposium.

Volger, D., and J. Stressler. 2011. "Off with their heads!!" Guidelines for controlling European Marsh Thistle on your property. Catskill Regional Invasive Species Partnership (CRISP), United States Department of Agriculture: Natural Resources Conservation Service (NRCS), State University of New York State (SUNY), Oneonta, NY.

Wadsworth, R.A., Y.C. Collingham, S.G. Willis, B. Huntley, and P.E. Hulme. 2000. Simulating the spread and management of alien species riparian weeds: are they out of control? *Journal of Applied Ecology* 37: 28-38.

Waggy, M.A. 2009. *Solanum dulcamara*. In: Fire Effects Information System. U.S. Department of Agriculture, Forest Service, Rocky Mountain Research Station, Fire Sciences Laboratory. Available <http://www.fs.fed.us/database/feis/plants/shrub/soldul/all.html>. Accessed 12 April 2013.

Wagner, E.J. 2002. Whirling disease prevention, control, and management: a review. *American Fisheries Society Symposium* 29:217-225. Whirling Disease Initiative. 1997 – 1998. Research Findings 1997 – 1998, National Partnership on the management of wild and native cold water fisheries, Montana University System Water Center, MSU-Bozeman. 6 pp.

Wahab, M.A., H.P. Stirling, and D.A. Robertson. 1989, Influence of Brown Trout, *Salmo trutta* L., predation on the benthic fauna of earthen ponds. *Aquaculture Research* 20: 147–158.

Warren, J.W. 1983. Viral Hemorrhagic Septicemia. United States Department of the Interior: Fish and Wildlife Service. Pp. 175-179 in Meyer, F.P., J.W. Warren, and T.G. Carey (editors), *A Guide to Integrated Fish Health Management in the Great Lakes Basin*, Spec. Pub. 83-2. Great Lakes Fishery Commission, Ann Arbor, Michigan.

Washington State Department of Ecology. 2012. Non-native Invasive Freshwater Plants: Purple Loosestrife (*Lythrum salicaria*). Available <http://www.ecy.wa.gov/programs/wq/plants/weeds/aqua009.html>. Accessed 12 May 2012.

Washington State Department of Ecology. 2013. Non-native Invasive Freshwater Plants: *Myriophyllum spicatum* (Eurasian Watermilfoil). Available <http://www.ecy.wa.gov/programs/wq/plants/weeds/aqua004.html>. Accessed 25 May 2013.

Water Bureau. 2005. Aquatic Herbicides. Michigan Department of Environmental Quality. 2 pp.

Wedekind, H., V. Hilge, and W. Steffens. 2001. Present status, and social and economic significance of inland fisheries in Germany, and social and economic significance of inland fisheries in Germany. *Fisheries Management Ecology*. 8: 405–414.

- Welling, C. 2013. Eurasian Watermilfoil Management Program. Minnesota Department of Natural Resources.
Available <http://www.dnr.state.mn.us/invasives/aquaticplants/milfoil/program.html>. Accessed 25 April 2013.
- West, L.D., J.H. Dawson, and A.P. Appleby. 1980. Factors influencing barnyardgrass (*Echinochloa crus-galli*) control with diclofop. *Weed Science* 28(4): 366-371.
- Westrick, J.A., D.C. Szlang, B.J. Southwell, and J. Sinclair. 2010. A review of cyanobacteria and cyanotoxins removal/ inactivation in drinking water treatment. *Analytical and Bioanalytical Chemistry* 397(5): 11705-1714.
- Whittaker, J.B. 1994. Physiological responses of leaves of *Rumex obtusifolius* to damage by a leaf miner. *Functional Ecology* 8: 627-630.
- Wicks, G.A., and G.E. Hanson. 1995. Effect of rainfall on glyphosate plus 2,4-D performance on *Echinochloa crus-galli*. *Weed Science* 43(4): 666-670.
- Winjnhoven, S., M.C. van Riel, and G. van der Velde. 2003. Exotic and indigenous freshwater gammarid species: physiological tolerance to water temperature in relation to ionic content of the water. *Aquatic Ecology* 37: 151-158.
- Wiklund, T., and I. Dalsgaard. 1998. Occurrence and significance of atypical *Aeromonas salmonicida* in non-salmonid and salmonid fish species: a review. *Diseases of Aquatic Organisms* 32: 49-69.
- Williams, Perry. 2007. Faucet Snail. University of Minnesota. Available http://fwcb.cfans.umn.edu/courses/nresexotics3002/GradPages/Faucet_snail/. Accessed 28 October 2013.
- Willoughby, I., F.L. Dixon, D.V. Clay, and R.L. Jinks. 2007. Tolerance of broadleaved tree and shrubs seedlings to preemergence herbicides. *New Forests* 34: 1-12.
- Wilson, C.E., S.J. Darbyshire, R. Jones. 2007. The biology of invasive alien plants in Canada. 7. *Cabomba caroliniana* A. Gray. *Canadian Journal of Plant Science* 87: 615-638.
- Wilson, L.M., M. Schwarzlaander, B. Blossey, and C.B. Randall. 2004. Biology and Biological Control of Purple Loosestrife. USDA Forest Service, FHTET-04-12.
- Winn, R.N., and D.M. Knott. 1992. An evaluation of the survival of experimental populations exposed to hypoxia in the Savannah River estuary. *Marine Ecology Progress Series* 88(2-3): 161-179.

Winston, R., R. Hansen, M. Schwarzlander, E. Coombs, C.B. Randall, and R. Lym. 2008. Biology and Biological Control of Exotic True Thistles. United States Department of Agriculture (USDA) Forest Service. University of Nebraska- Lincoln Faculty Publications, Paper 114.

Wisconsin Department of Natural Resources (WIDNR). 2008. Poison Hemlock or Poison Parsley (*Conium maculatum*). Available http://dnr.wi.gov/invasives/fact/poison_hemlock.htm.

Wisconsin Department of Natural Resources (WIDNR). 2010a. NR 40 Invasive Species Lists. Available <http://dnr.wi.gov/topic/invasives/documents/nr40listssep292010.pdf>. Accessed 22 June 2012.

Wisconsin Department of Natural Resources (WIDNR). 2010b. Watercress: *Nasturtium officinale*. 5 pp. Available <http://dnr.wi.gov/topic/Invasives/documents/classification/Nasturtium%20officinale.pdf>. Accessed 29 April 2013.

Wisconsin Department of Natural Resources (WIDNR), Division of Forestry. 2011. Herbicide Sensitivity Table for Disturbed Area Herbaceous Weeds. Available http://dnr.wi.gov/topic/ForestHealth/documents/HerbicideTables_AgWeeds.pdf.

Wisconsin Department of Natural Resources (WIDNR). 2012a. Curly-leaf pondweed (*Potamogeton crispus*). Available <http://dnr.wi.gov/topic/invasives/fact/curlyleafpondweed.html>. Accessed 29 April 2013.

Wisconsin Department of Natural Resources (WIDNR). 2012b. Eurasian water-milfoil (*Myriophyllum spicatum*). Available <http://dnr.wi.gov/topic/invasives/fact/eurasianwatermilfoil.html>. Accessed 25 April 2013.

Wisconsin Department of Natural Resources (WIDNR). 2012c. European frog-bit (*Hydrocharis morsus-ranae*). Available <http://dnr.wi.gov/topic/Invasives/fact/EuropeanFrogbit.html>. Accessed 17 April 2013.

Wisconsin Department of Natural Resources (WIDNR). 2012d. Fanwort (*Cabomba caroliniana*). Available <http://dnr.wi.gov/topic/invasives/fact/fanwort.html>. Accessed 16 April 2013.

Wisconsin Department of Natural Resources (WIDNR) 2012e. Water Chestnut (*Trapa natans*). Available <http://dnr.wi.gov/topic/invasives/fact/waterchestnut.html>.

Wisconsin Department of Natural Resources (WIDNR). n.d. So, what should I plant? Trees, shrubs, and vines with wildlife. Wisconsin Department of Natural Resources. Available <http://dnr.wi.gov/org/land/wildlife/publ/whattoplant1.pdf>. Accessed 2 February 2012.

- Wisconsin Department of Natural Resources (WIDNR), Division of Forestry. 2011. Herbicide Sensitivity Table for Invasive Herbaceous Plants. Wisconsin Department of Natural Resources. Madison, WI. 2 pp.
- Wittmann, M.E., S. Chandra, J.E. Reuter, S.G. Schladow, B.C. Allen, and K.J. Webb. 2012. The control of an invasive bivalve, *Corbicula fluminea*, using gas impermeable benthic barriers in a large natural lake. *Environmental Management* 49(6): 1163-73.
- Wolf, K., and C.E. Dunbar. 1959. Methods of infecting trout with kidney disease and some effects temperature on experimental infections. United States Fish and Wildlife Service's Special Report 286: 1-8.
- Woodard, C.A. 2008. Poison Hemlock (*Conium maculatum* L.): biology, implications for pastures and response to herbicides. Unpublished Master's thesis. University of Missouri, Columbia, Missouri.
- Wood, J.W. 1967. Salmon disease report. Washington Department of Fish and Wildlife Annual Report 77: 111-112.
- World Organization for Animal Health (OIE). 2009. Manual of Diagnostic Tests for Aquatic Animals: Spring Viremia of Carp. Chapter 2.3.8.
http://www.oie.int/fileadmin/Home/eng/Health_standards/ahm/2010/2.3.08_SVC.pdf. Accessed 14 June 2012.
- World Organization for Animal Health (OIE). 2012. OIE Listed Diseases.
<http://www.oie.int/animal-health-in-the-world/oie-listed-diseases-2012>. Accessed 10 June 2012.
- Wu, M., and J. Wu. 2007. In-vitro investigations on ultrasonic control of Water Chestnut. *Journal of Aquatic Plant Management* 45: 76-83.
- Yamamoto, T., Y. Kohmatsu, and M. Yuma. 2006. Effects of summer drawdown on cyprinid fish larvae in Lake Biwa, Japan. *Limnology* 7(2): 75-82.
- Ye, N., X. Zhang, Y. Mao, C. Liang, D. Xu, J. Zou, Z. Zhuang, and Q. Wang. 2011 'Green tides' are overwhelming the coastline of our blue planet: taking the world's largest example. *Ecological Research* 26(3): 477-485.

7. Acknowledgements

Many thanks are given to the GLANSIS expert review panel for valuable input and feedback, including Anthony Ricciardi, Patricia Chow-Fraser, Hugh MacIsaac, Eugene Stoermer, Sarah Bailey, Hunter Carrick, Susan Galatowitsch, Jeff Gunderson, Rex Lowe, Nicholas Mandrak, and Robin Scribailo. We also appreciate the feedback received on individual species assessments from several external reviewers, including Steve Hensler, Tim Campbell, Titus Selheimer, Kevin Irons, Blake Ruebush, and Lisa Huberty. We thank the staff of the Cooperative Institute for

Limnology and Ecosystem Research (CILER) and the Great Lakes Environmental Research Laboratory (GLERL) for providing many forms of support throughout this project. We also recognize past GLANSIS team members, including Gabriela Núñez, Kyle Dettloff, Emily Baker, Julie Larson, Mary McCarthy, Alex Bogdanoff and Katie Thompson, for their contributions and dedication to the improvement of GLANSIS. This research was funded through Great Lakes Restoration Initiative interagency agreement #DW-13-92312201-0. This is GLERL Contribution No. 1795.

Appendix A. Species Management Profiles

Notes:

- Check federal, state/provincial, and local regulations for the most up-to-date information.
- Check state and local regulations for the most up-to-date information regarding permits for control methods.
- Follow all label instructions.

A.1 Algae

***Actinocyclus normanii f. subsalsa* (Juhlin-Dannfelt) Hustedt, 1957**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Actinocyclus normanii f. subsalsa is one of the most pollution tolerant algal species and thrives in warm, shallow, and eutrophic waters (Edlund et al. 2000). The reduction of pollution and nutrient run-off would decrease the viable habitat for *A. normanii f. subsalsa*.

***Bangia atropurpurea* (Roth) Agardh, 1824**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

The distribution of *B. atropurpurea* in the Great Lakes is associated with elevated salinity and eutrophic conditions (Lin and Blum 1976, Sheath and Cole 1984, Graham and Graham 1987, Jackson 1988, Stewart 2008). The reduction of pollution and nutrient run-off could decrease the viable habitat for *B. atropurpurea*.

***Chaetoceros muelleri subsalsum* J. R. Johansen and Rushforth, 1985**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

The reduction of nutrient pollution, specifically of NaCl, would eliminate viable water conditions for *C. muelleri* var. *subsalsum* in the Great Lakes.

***Chroodactylon ornatum* (C. Agardh) Basson, 1979**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

***Contricribra guillardii* (Hasle) K. Stachura-Suchoples & D.M. Williams**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species

Physical

There are no known physical control methods for this species

Chemical

Contricribra guillardii is a brackish water species. Therefore, the reduction of pollution and nutrient run-off could decrease the viable habitat for *C. guillardii*.

***Cyclotella atomus* Hustedt, 1937**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

The reduction of pollution and nutrient run-off would decrease the viable habitat for *C. atomus*.

Cyclotella cryptica Reimann, Lewin, and Guillard, 1963

Regulations (pertaining to the Great Lakes region)Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Cyclotella cryptica is a euryhaline species (Liu and Hellebust 1976). As a result, reduction in run-off from winter road salt could decrease the chloride levels in the rivermouth areas and reduce the viable habitat for *C. cryptica*.

Cylindrospermopsis raciborskii (Wolosz.) Seenayya and Subbaraju, 1972

Regulations

There are no known regulations for this species.

Control

Biological

There are no known organisms that can degrade cylindrospermopsin. However, several studies have found that a variety of unidentified bacteria degraded 100% of saxitoxin, another toxin produced by *C. raciborskii* (Donovan et al. 2008, Ho et al. 2012).

Physical

Cylindrospermopsis raciborskii blooms are often associated with a stratified water column, which is one of the reasons they are so prevalent in shallow water bodies with a long turnover period. A mechanical system can be used to create artificial destratification to increase vertical mixing, introduce oxygen, and reduce internal nutrient loading. The installation of a destratification mechanism was attempted in a reservoir in Australia. While there was a reduction

in internal nutrient loading, the increased turbidity of the water yielded a competitive advantage for *C. raciborskii*, which can move throughout the water column to compete for light (Atenucci et al. 2005).

Cylindrospermopsin can be absorbed by activated carbon with high mesopor capacity, and nanofiltration may be another viable option, but not enough research has been done to be conclusive about the efficacy of either technique. Saxitoxins can be absorbed by activated carbons that have a large fraction of the pores being smaller than 1 nm (Westrick 2010).

Chemical

Cylindrospermopsin can be inactivated by chlorine, ozone, and hydroxyl radical treatments. Saxitoxin can be inactivated by chlorine (Westrick 2010). However, the use of copper-based algicides may inhibit the degradation of cylindrospermopsin (Smith et al. 2008).

***Diatoma ehrenbergii* Kützing, 1844**

Regulations (pertaining to the Great Lakes region)

There are no known regulations of this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Diatoma ehrenbergii was recorded in eutrophic waters in the Great Lakes (Stoermer and Yang 1969). The reduction of pollution and nutrient run-off could decrease the viable habitat for *D. ehrenbergii*.

***Discostella pseudostelligera* (Hustedt) Houk and Klee, 1939**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species

Control

Biological

There are no known biological control methods for this species

Physical

There are no known physical control methods for this species.

Chemical

Reduction of pollution and nutrient run-off would decrease the viable habitat for *D. pseudostelligera*.

Discostella woltereckii Hustedt, 1942

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical methods of control for this species.

Hymenomonas roseola Stein, 1878

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical methods of control for this species.

Nitellopsis obtusa (Desvaux in Loiseleur) J. Groves, (1919)

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species

Physical

It is difficult to mechanically remove all of a *N. obtusa* population from an inland lake because of the large amounts of biomass. Additionally, even when entire plants are removed, *N. obtusa* is typically the first macrophyte to reestablish the disturbed area because it is such an aggressive and efficient recolonizer (Pullman and Crawford 2010).

Chemical

Nitellopsis obtusa is very sensitive to common algaecides containing copper and endothall based compounds. When *N. obtusa* is still low growing, algaecide treatment can treat the entire organism. However, in taller individuals, the algaecide is absorbed in the top of the plant, killing that portion but leaving the bottom of the plant alive. This type of treatment has been found to be somewhat successful and is called a “hair cut treatment” by managers. The timing of the algaecide treatment is also important. Treatment early in the spring could help open up spawning habitat for native fish species, but *N. obtusa* or other nonnative aquatic plants are likely to recolonize these areas in the early summer (Pullman and Crawford 2010). Some applicators report control of *Nitellopsis obtusa* with 150ppb flumioxazin.

Pleurosira laevis (Ehrenberg) Compère, (1843) 1982

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical methods of control for this species.

Skeletonema potamos (Weber) Hasle in Hasle & Evensen, (1970)

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical methods of control for this species.

Skeletonema subsalsum (Cleve-Euler) Bethge, (1912) 1928

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical methods of control for this species.

Sphacelaria fluviatilis Jao, 1943

Regulations (pertaining to the Great Lakes region)

There are no known methods of regulation for this species.

Control

There are no known biological, physical or chemical methods of control for this species.

Sphacelaria lacustris Schloesser and Blum, 1980

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical methods of control for this species.

Stephanodiscus binderanus Krieger, 1927

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Stephanodiscus binderanus thrives in eutrophic waters. The reduction of pollution and nutrient run-off could decrease the viable habitat for *S. binderanus*.

Stephanodiscus subtilis (Van Goor) A. Cleve, 1951

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Stephanodiscus subtilis occurs primarily in eutrophic waters with elevated chloride levels (Millie and Lowe 1983). The reduction of pollution and nutrient run-off could decrease the viable habitat for *S. subtilis*.

Thalassiosira baltica Ostenfeld, 1901

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical methods of control for this species.

Thalassiosira lacustris (Grunow) Hasle in Hasle and Fryxell, 1977

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

The ability of *T. lacustris* to reproduce and grow is highly dependent upon salinity. The reduction of pollution and run-off could decrease the viable habitat for this nonindigenous species.

Thalassiosira pseudonana (Hustedt) Hasle and Heimdal, (1957) 1970

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical methods of control for this species.

Thalassiosira weissflogii (Grunow) G. Fryxell & Hasle, (1896) 1977

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Thalassiosira weissflogii struggles to grow or reproduce at salinities below 5 ‰ (Stoermer 1978). The reduction of pollution from road salt run-off could decrease the viable habitat for *T. weissflogii*.

Ulva (Enteromorpha) flexuosa* subsp. *flexuosa* and *flexuosa* subsp. *paradoxa (Wolfen ex Roth) J. Agardh, 1883

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

The presence of *U. flexuosa* is associated with high nutrient availability and high levels of salinity (Shories et al. 1997, Lougheed and Stevenson 2004). The reduction of pollution and nutrient run-off could decrease the viable habitat for *U. flexuosa*.

***Ulva (Enteromorpha) intestinalis* Linnaeus, 1753**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

In locations outside of the Great Lakes, the distribution and abundance of *U. intestinalis* is dependent on salinity and nutrient levels (Kamer and Fong 2000, 2001, Messyasz and Rybak 2011). The reduction of pollution and nutrient run-off could decrease the viable habitat for *U. intestinalis*.

***Ulva (Enteromorpha) prolifera* O.F. Müller, 1778**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

Ulva prolifera can be physically harvested from the water and beaches. However, depending on the size of the bloom this may not be an economically viable option (Ye et al. 2011).

Chemical

Ulva prolifera blooms occur in eutrophic marine waters. As a result, the reduction of pollution and nutrient run-off could decrease the viable habitat for *U. prolifera*.

A.2 Annelids

Branchiura sowerbyi

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Branchiura sowerbyi has not received much attention regarding control studies. The effects of industrial toxicants on *Tubificidae* species and explorations of their value as an indicator of environmental quality have been explored, but chemicals and heavy metals are not viable

methods of control because of unknown and adverse effects on the surrounding environment (Das and Das 2005, Saha et al. 2006). However, there has been investigation into the control of *Branchiura sowerbyi* as a host of haemorrhagic thelohanellosis, which negatively impacts fishes in aquaculture (Liyanage et al. 2003).

Biological

Brown Trout, *Salmo trutta* L., has been shown to prey on oligochaetes; its removal from an experimental environment led to rapid multiplication of benthic fauna (Wahab et al. 1989). However, Brown Trout is itself an invasive species in the Great Lakes region and across nearly all of the United States.

Physical

Researchers found that *Branchiura sowerbyi*, which is a vector in transmission of *Thelohanellus hovorkai* (myxozoa) to fish, prefers muddy substrate, while other benthic oligochaetes that are not susceptible to myxozoa prefer sandy substrate, and suggested that replacing bottom substrate from mud to sand would lead to a shift in oligochaete communities from *Branchiura sowerbyi* to non-susceptible oligochaetes such as *Limnodrilus socialus*, therefore reducing disease in aquaculture fauna (Liyanage et al. 2003).

Chemical

While there are no known chemical controls specifically for *Branchiura sowerbyi*, declines in Oligochaeta in southern Lake Michigan were recorded between 1980 and 1993 in correlation with reductions in phosphorus loads (Nalepa et al. 1998), suggesting that reduction of excess nutrients would help to reduce oligochaete populations.

Gianius aquaedulcis

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Little research has been pursued concerning control of *Gianius aquaedulcis*, likely because of its limited known range to the Niagara River in North America (Spencer and Hudson 2003).

Biological

While there are no known biological controls specifically for *Gianius aquaedulcis*, Brown Trout, *Salmo trutta* L., has been shown to prey on oligochaetes, and its removal from an experimental environment led to rapid multiplication of benthic fauna (Wahab et al. 1989). However, Brown Trout is itself nonindigenous to the Great Lakes region and across nearly all of the United States.

Research on benthic macroinvertebrate communities in southwestern Lake Ontario before and after the invasion of *Dreissena polymorpha* (Zebra Mussels) and *Dreissena bugensis* (Quagga Mussels) suggests that the presence of *Dreissena* helps to improve benthic habitat, facilitating increases in macroinvertebrates, including the tubificids *Potamothrix vejdoskyi* and *Spirosperma ferox* (Stewart and Haynes 1994). This indicates that control of invasive quagga and

Zebra Mussels could facilitate improved control of benthic macroinvertebrates such as the tubificids.

Physical

There are no known physical control methods for this species.

Chemical

While there are no known chemical controls specifically for *Gianius aquaedulcis*, declines in Oligochaeta in southern Lake Michigan were recorded between 1980 and 1993 in correlation with reductions in phosphorus loads (Nalepa et al. 1998), suggesting that reduction of excess nutrients would help to reduce oligochaete populations.

***Potamothrix bedoti* (Piquet, 1913)**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

While there are no known biological controls specifically for *Potamothrix bedoti*, Brown Trout, *Salmo trutta* L., has been shown to prey on oligochaetes, and its removal from an experimental environment led to rapid multiplication of benthic fauna (Wahab et al. 1989). However, Brown Trout is itself nonindigenous to the Great Lakes region and across nearly all of the United States.

Research on benthic macroinvertebrate communities in southwestern Lake Ontario before and after the invasion of *Dreissena polymorpha* (Zebra Mussels) and *Dreissena bugensis* (Quagga Mussels) suggests that the presence of *Dreissena* helps to improve benthic habitat, facilitating increases in macroinvertebrates, including the tubificids *Potamothrix vejdoskyi* and *Spirosperma ferox* (Stewart and Haynes 1994). This indicates that control of invasive quagga and Zebra Mussels could facilitate improved control of benthic macroinvertebrates such as the tubificids.

Physical

Potamothrix bedoti has been shown to be more likely to occur in substrate with a high clay and silt content, but information on its ability to survive in other substrates is not available (Sauter and Gude 1996). However, this does indicate that substrate type is a possible physical control method to be further explored.

Chemical

While there are no known chemical controls specifically for *Potamothrix bedoti*, declines in Oligochaeta in southern Lake Michigan were recorded between 1980 and 1993 in correlation with reductions in phosphorus loads (Nalepa et al. 1998), suggesting that reduction of excess nutrients would help to reduce oligochaete populations.

***Potamothrix moldaviensis* Vejdovsky and Mrazek, 1902**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

While there are no known biological controls specifically for *Potamothrix moldaviensis*, Brown Trout, *Salmo trutta* L., has been shown to prey on oligochaetes, and its removal from an experimental environment led to rapid multiplication of benthic fauna (Wahab et al. 1989). However, Brown Trout is itself nonindigenous to the Great Lakes region and across nearly all of the United States.

Research on benthic macroinvertebrate communities in southwestern Lake Ontario before and after the invasion of *Dreissena polymorpha* (Zebra Mussels) and *Dreissena bugensis* (Quagga Mussels) suggests that the presence of *Dreissena* helps to improve benthic habitat, facilitating increases in macroinvertebrates, including the tubificids *Potamothrix vejnovskyi* and *Spirosperma ferox* (Stewart and Haynes 1994). This indicates that control of invasive quagga and Zebra Mussels could facilitate improved control of benthic macroinvertebrates such as the tubificids.

Physical

P. moldaviensis has been shown to be more likely to occur in sandy substrate with a clay and silt content of less than 10%, but information on its ability to survive in other substrates is not available (Sauter and Gude 1996). However, this does indicate that substrate type is a possible physical control method to be further explored.

Chemical

While there are no known chemical controls specifically for *P. moldaviensis*, declines in Oligochaeta in southern Lake Michigan were recorded between 1980 and 1993 in correlation with reductions in phosphorus loads (Nalepa et al. 1998), suggesting that reduction of excess nutrients would help to reduce oligochaete populations.

***Potamothrix vejnovskyi* Hrabě, 1941**

Regulations (pertaining to the Great Lakes region)

Regulations

There are no known regulations for this species.

Note: Check federal, state/provincial, and local regulations for the most up-to-date information.

Control

Biological

While there are no known biological controls specifically for *Potamothrix vejnovskyi*, Brown trout, *Salmo trutta* L., has been shown to prey on oligochaetes, and its removal from an experimental environment led to rapid multiplication of benthic fauna (Wahab et al. 1989). However, brown trout is itself nonindigenous to the Great Lakes region and across nearly all of the United States.

Research on benthic macroinvertebrate communities in southwestern Lake Ontario before and after the invasion of *Dreissena polymorpha* (zebra mussels) and *Dreissena bugensis* (quagga mussels) suggests that the presence of *Dreissena* helps to improve benthic habitat, facilitating increases in macroinvertebrates, including the tubificids *Potamothrix vejovskyi* and *Spirosperma ferox* (Stewart and Haynes 1994). This indicates that control of invasive quagga and zebra mussels could facilitate improved control of benthic macroinvertebrates such as the tubificids.

Physical

There are no known physical control methods for this species.

Chemical

While there are no known chemical controls specifically for *Potamothrix vejovskyi*, declines in Oligochaeta in southern Lake Michigan were recorded between 1980 and 1993 in correlation with reductions in phosphorus loads (Nalepa et al. 1998), suggesting that reduction of excess nutrients would help to reduce oligochaete populations.

Ripistes parasita

Regulations

There are no known regulations for this species.

Control

Biological

While there are no known biological controls specifically for *Ripistes parasita*, Brown trout, *Salmo trutta* L., has been shown to prey on oligochaetes, and its removal from an experimental environment led to rapid multiplication of benthic fauna (Wahab et al. 1989). However, brown trout is itself an invasive species in the Great Lakes region and across nearly all of the United States.

Physical

There are no known physical control methods for this species.

Chemical

Ripistes parasite has been found to occur in greater numbers where water quality is impaired by industrial pollution, therefore greater measures to control pollutants such as heavy metals and particulate matter might help control this oligochaete (Simpson and Abele 1984). Furthermore, declines in Oligochaeta in southern Lake Michigan were recorded between 1980 and 1993 in correlation with reductions in phosphorus loads (Nalepa et al. 1998), suggesting that reduction of excess nutrients would help to reduce oligochaete populations.

A.3 Bacteria

Aeromonas salmonicida Emmerich and Weible, 1890

Regulations (pertaining to the Great Lakes)

Ohio requires source facilities outside the Great Lakes basin to document annual health inspections showing no furunculosis occurrences for the previous five years prior to importing salmonids to the Lake Erie watershed (Baird 2005). Indiana requires source facilities outside the basin to document they are furunculosis free prior to importing salmonid stock. Salmonids found carrying the pathogen, but asymptomatic, can be sold in state if source facilities are within the Great Lakes basin (Baird 2005). Michigan, Illinois, Wisconsin, and Minnesota also require imported salmonid health inspections. However, *A. salmonicida* is not a targeted pathogen. Minnesota allows the importation of furunculosis infected eggs, if prior egg treatments are approved (Baird 2005).

New York, Pennsylvania, Michigan, Illinois, Wisconsin, and Minnesota have instated similar baitfish regulations to control the spread of furunculosis and other fish pathogens. Those of New York include that bait harvested from inland waters for personal use is only permitted to be used within the same body of water from which it was taken and cannot be transported overland (with the exception of smelt, suckers, alewives, and blueback herring). Once transported, baitfish cannot be replaced to its original body of water (NYSDEC 2012).

Live or frozen bait harvested from inland New York waters for commercial purposes is only permitted to be sold or possessed on the same body of water from which it was taken and cannot be transported over land unless under a permit and or accompanied by a fish health certification report. Bait that is preserved and packaged by any method other than freezing, such as salting, can be sold and used wherever the use of bait fish is legal as long as the package is labeled with the name of the packager-processor, the name of the fish species, the quantity of fish packaged, and the means of preservation (NYSDEC 2012).

Certified bait may be sold for retail and transported overland as long as the consumer maintains a copy of a sales receipt that contains the name of the selling vendor, date sold, species of fish sold, and quantity of fish sold. Bait that has not been certified may still be sold but the consumer must maintain a sales receipt containing the body of water where the bait fish was collected and a warning that the bait cannot be transported by motor vehicle. Bait sold for resale require a fish health certification along with a receipt that contains the name of the selling vendor, date sold, species of fish sold, and quantity of fish sold, which must be kept for 30 days or until all bait is sold (NYSDEC 2012).

In addition to baitfish protections, prior to placing fish in New York waters, a fish health certification report must document that the fish are furunculosis free.

Control

Biological

While supplements of lactic acid bacteria (*Carnobacterium* spp.) given with fish feed do not protect against *A. salmonicida* infection (Gildberg et al. 1995), such probiotic supplementation (also *A. hydrophila* and *Vibrio fluvialis*) can decrease mortality in Atlantic Salmon (*Salmo salar*) and rainbow trout (*Oncorhynchus mykiss*) infected with *A. salmonicida* (Irianto and Austin 2002). *Carnobacterium* strain K1 colonizes the intestinal tract of rainbow trout and inhibits *A. salmonicida* growth (Jöborn et al. 1997). Short-term bathing of presmolt Atlantic Salmon

infected with furunculosis with siderophore-producing *Pseudomonas fluorescens* is another successful biological control (Gram et al. 1999, Smith and Davey 1993). Bath treatments with *V. alginolyticus* (Austin et al. 1995) can also lead to a reduction in mortality (Verschuere et al. 2000).

Administration of bacteriophage to infected fish may also help control outbreaks. In a study that administered the bacteriophage HER 110 to *A. salmonicida* HER 1107 infected Brook Trout (*Salvelinus fontinalis*), *A. salmonicida* populations declined by six log units (base 10) in 3 d. Further tests within fish populations are necessary to better understand the implementations of this alternative therapy (Imbeault et al. 2006).

The US Food and Drug Administration approved vaccine Furogen® (Aqua Health, LTD) administered to brood stock prior to spawning has proven to be very effective at reducing *A. salmonicida* prevalence (GLFHC 2006). Very good to excellent results controlling and preventing out-breaks of furunculosis have also been obtained using an autogenous vaccine, produced by Microtechnologies; this treatment was accompanied by an immune-enhancing feed administered for three weeks prior and three weeks post vaccination, as well as stock thinning to reduce overall stress (GLFHC 2006).

Physical

There are no known physical control methods for this species.

Chemical

Antimicrobial agents used to treat *A. salmonicida* infections include thiophenicol, furazolidone (Herman 1968), oxytetracycline (Herman 1968, Heo and Seo 1996, Wiklund and Dalsgaard 1998), sulphamerazine, tetracycline and a combination of trimethoprim (Heo and Seo 1996, Wiklund and Dalsgaard 1998) and sulphonamide (McCarthy and Roberts 1980). Others include flumequine (Michel et al. 1980), oxolinic acid (Hastings and McKay 1987, Heo and Seo 1996, Wiklund and Dalsgaard 1998), florfenicol (Inglis et al. 1991b), amoxycillin (Inglis et al. 1992), enrofloxacin (Stoffregen et al. 1993), chloramphenicol, neomycin, nitrofurantoin and ciprofloxacin (Heo and Seo 1996, Wiklund and Dalsgaard 1998). Feed containing terramycin and romet are effective in treating *A. salmonicida* (MIDNR 2012).

However, an increase in antimicrobial resistance was recognized in the United States beginning in 1967 (Wood 1967). Antimicrobial resistance by *A. salmonicida* has been discovered with the following agents: sulphonamides (Herman 1968), oxytetracycline (Tsoumas et al. 1989, Inglis et al. 1991a, Grant and Laidler 1993), a combination of sulphonamide and trimethoprim (Tsoumas et al. 1989, Grant and Laidler 1993), oxolinic acid (Hastings and McKay 1987, Tsoumas et al. 1989, Inglis et al. 1991a, Höie 1992, Grant and Laidler 1993, Oppegaard and Sörum 1994), flumequin (Höie 1992), and amoxycillin (Grant and Laidler 1993). The United States Food and Drug Administration approved broad-spectrum, in-feed antibiotic AQUAFLO® is now available to control mortality in finfish due to furunculosis (MAA 2012).

Other

Minimizing fish stress can reduce the risk of disease outbreak (FTS 2012).

Piscirickettsia cf. salmonis

Regulations (pertaining to the Great Lakes)

There are no known regulations for this species.

Control

Biological

Vaccines derived from inactivated *P. cf. salmonis* bacterins are considered ineffective against the bacterium. However, vaccines being developed using recombinant DNA have promise in combating *P. salmonis* infections (Corbeil and Crane 2009). Further research is required to understand the practical application of this therapy.

Physical

Culling is effective at preventing horizontal transmission of *P. cf. salmonis* (Torenzo et al. 2005).

Chemical

Renogen® can reduce mortality rates in *P. cf. salmonis* infected Pacific salmon (Torenzo et al. 2005). All commonly used aquaculture disinfectants are considered effective against *Piscirickettsia* spp. (Fryer et al. 1990 and 1992, Corbeil and Crane 2009,). Antimicrobial agents used to treat *P. salmonis* include oxolinic acid and flumequin (Guardabassi and Courvalin 2006, Todar 2008, Corbeil and Crane 2009).

Other

Establishment of muskellunge fingerling index surveys may help monitor trends in spawning success and or fingerling survival (Thomas and Faisal 2009).

Renibacterium (Corynebacterium) salmoninarum Sanders and Fryer, 1980

Regulations (pertaining to the Great Lakes)

Ohio requires source facilities outside the Great Lakes basin to document annual health inspections showing no bacterial kidney disease (BKD) occurrences for the previous five years prior to importing salmonids to the Lake Erie watershed (Baird 2005). Indiana requires source facilities outside the basin to document they are BKD free prior to importing salmonid stock. Asymptomatic salmonids found carrying the pathogen can be sold in-state if source facilities are within the Great Lakes basin (Baird 2005). Michigan, Illinois, Wisconsin, and Minnesota also require imported salmonid health inspections (NCRAC 2010ab). Minnesota allows the importation of infected eggs, if prior egg treatments have been approved (Baird 2005).

New York, Pennsylvania, Ohio, Michigan, Indiana, Illinois, Wisconsin, and Minnesota have instated similar baitfish regulations to control the spread of BKD and other fish pathogens. Those of New York include that bait harvested from inland waters for personal use is only permitted to be used within the same body of water from which it was taken and cannot be transported overland (with the exception of smelt, suckers, Alewives, and Blueback Herring). Once transported, baitfish cannot be replaced to its original body of water (NYSDEC 2012a). Live or frozen bait harvested from inland New York waters for commercial purposes is only permitted to be sold or possessed on the same body of water from which it was taken and cannot

be transported over land unless under a permit and or accompanied by a fish health certification report. Bait that is preserved and packaged by any method other than freezing, such as salting, can be sold and used wherever the use of baitfish is legal as long as the package is labeled with the name of the packager-processor, the name of the fish species, the quantity of fish packaged, and the means of preservation (NYSDEC 2012a).

Certified bait may be sold for retail and transported overland as long as the consumer maintains a copy of a sales receipt that contains the name of the selling vendor, date sold, species of fish sold, and quantity of fish sold. Bait that has not been certified may still be sold but the consumer must maintain a sales receipt containing the body of water where the baitfish was collected and a warning that the bait cannot be transported by motor vehicle. Bait sold for resale require a fish health certification along with a receipt that contains the name of the selling vendor, date sold, species of fish sold, and quantity of fish sold, which must be kept for 30 days or until all bait is sold (NYSDEC 2012).

In addition to baitfish protections, prior to placing fish in New York waters, a fish health certification report must document that the fish are BKD free.

Control

Biological

There are no known biological control methods for this species.

Physical

Culling infected stock has reduced the prevalence of *R. salmoninarum* in aquaculture (Gudmundsdóttir et al. 2000, Maule et al. 1996, Pascho et al. 1991).

Chemical

Antimicrobial agents used to treat *R. salmoninarum* include nitrofurans, bacitracin, chlortetracycline, oxytetracycline, novobiocin (Wolf and Dunbar 1959), cephalosporins, gentamicin, clindamycin, lincomycin, oleandomycin, kitasamycin, spiramycin, penicillin (Austin 1985), cefazolin, tiamulin (Bandín et al. 1991), cephradine, rifampicin, (Brown et al. 1990), tetracycline (Austin 1985, Bandín et al. 1991), chloramphenicol (Austin 1985, Wolf and Dunbar 1959), and erythromycin (Austin 1985, Bandín et al. 1991, Lee and Evelyn 1994, Wolf and Dunbar 1959).

Erythromycin-enhanced feed administered at 100 mg/kg/day for 21 days (Wolf & Dunbar 1959) or for 10 days (Austin 1985) is believed to be the most effective treatment for bacterial kidney disease (Hirvela-koski 2005). Erythromycin phosphate is an effective chemoprophylaxis in pre-spawning adult brood fish. Subcutaneous injections of 11 mg/kg (Hirvela-koski 2005) or 20 mg/kg (Gudmundsdóttir et al. 2000, Pascho et al. 1991) administered to fish entering trapping facilities and at 21-30 day intervals thereafter (Hirvela-Koski 2005) has reduced mortality in re-spawning Pacific salmon by 10-50% (Groman and Klonz 1983). However, this treatment does not effectively destroy the pathogen from all eggs (Brown et al. 1990). A study by Lee and Evelyn (1994) showed female Coho Salmon treated with 20 mg/kg erythromycin prior to spawning yielded no vertical transmission of BKD. Treatment success relies on careful timing of the injections in adult salmonids before spawning (Elliott et al. 1989).

Disinfecting eggs with 100 mg/L of iodine for 15 minutes may not eliminate vertical transmission, but can reduce the severity of the disease (Bullock et al. 1978). Treating eggs with iodine at 250 or 500 mg/L for 15-120 minutes is effective at eliminating *R. salmoninarum*. However, after treatment, variable numbers of *R. salmoninarum* cells have survived. This is due to cells within the cell aggregates never coming in contact with the iodine (Evelyn et al. 1984, 1986). *Renibacterium salmoninarum* is inactivated by free chlorine (≤ 0.05 mg/L), which can be used as a disinfectant and to treat intake and effluent (Pascho et al. 1995).

Other

Prevention is the preferred control method of BKD in cultured stocks (World Organization for Animal Health 2003).

A.4 Bryozoa

Lophopodella carteri (Hyatt, 1865)

Regulations

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

Lophopodella carteri colonies grow on solid substrata (Lauer et al. 1999), therefore, physical removal methods such as scraping may be viable for localized areas.

Chemical

Chemical biocides have been used as anti-fouling agents to remove sessile macroinvertebrates from shipping equipment and industrial intakes, but none have been approved for use on bryozoans as of yet (United States 2011). The Great Lakes and Mississippi River Interbasin Study (GLMRIS 2012) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of bryozoans.

Pardue and Wood (1980) determined baseline toxicity of four heavy metals to three species of phylactolaemate bryozoa (*L. carteri*, *Pectinatella magnifica* Leidy, and *Plumatella emarginata* Oka). They recorded 96-hr LC₅₀ values (lethal concentration for 50 percent of organisms tested) of *L. carteri*, observing greatest sensitivity to cadmium (LC₅₀ 0.15 mg/L), followed by copper (LC₅₀ 0.51 mg/L), chromium (LC₅₀ 1.56 mg/L), and zinc (LC₅₀ 5.63 mg/L). It should be noted that the toxicity of these metals were not tested as control measures, but as a demonstration of the usefulness of some bryozoans as biomonitors of water quality. However, comparison of the 96-hr LC₅₀ data to toxicity data from other studies indicates that the bryozoa are more sensitive to the tested metals than many other invertebrates and fish, indicating potential as chemical controls with further research (Pardue and Wood 1980).

A.5 Coelenterates

Cordylophora caspia (Pallas, 1771)

Regulations

There are no known regulations for this species.

Control

Control of *Cordylophora caspia* will likely focus on its potential role as a biofouling agent. *Cordylophora* spp. has been documented colonizing the inner walls of power plants in Europe and the United States (Folino 2000), primarily causing blockages in nozzles and tailpipes of rapid gravity filter beds (RGFs) (Mant et al. 2011). The menont life-stage of *Cordylophora caspia* often found in hydroelectric intakes, is both drought and temperature resistant which may prove an obstacle to control (Gutierre 2012).

Biological

In estuarine, brackish habitats such as San Francisco Bay on the Pacific and the North American Atlantic coast, nudibranchs such as *Tenellia adspersa* feed on *Cordylophora caspia* and other hydroids (Mills and Sommer 1995). However, unlike hydroids, nudibranchs are exclusively sea-dwelling invertebrates (Anderson 1995), and thus are only a source of biological control in brackish areas where the macroinvertebrates' habitats intersect.

Physical

Thermal treatments of $\geq 37^{\circ}\text{C}$ have been proven effective in eradicating colonies of *Cordylophora* spp. sampled from the walls of power plant intakes (Folino 2000), but are not efficient in water treatment facilities where there is no residual heat energy available (Mant et al. 2011).

Chemical

Gutierre (2012) found that *Cordylophora caspia* is completely eradicated at pH levels of 4.0 and 10.0, with increasing survival rates in between, and suggested maintaining pH levels at 10.0 for 6 hours or more by injection of NaOH to reduce and eliminate colonies. Chlorine treatments negatively affect *Cordylophora* growth, but treatments as high as 5 mg/L for periods of 105 minutes have been unsuccessful in completely eradicating colonies (Mant et al. 2011). Furthermore, chlorine use is highly regulated at water treatment facilities where *Cordylophora* most frequently cause problems. Hydroids are sensitive to vanadium leeching from slag stones used in riverbank reinforcement, and are sensitive to heavy metals in general, especially mercury, copper, cadmium, and arsenic, though it is unlikely that these will be useful in control (Ringelband and Karbe 1996).

Craspedacusta sowerbyi Lankester, 1880

Regulations

There are no known regulations for this species.

Control

Craspedacusta sowerbyi has spread across temperate climates for more than a century, but despite experimental observation of its possible contribution to trophic cascade effects (Jankowski et al. 2005), and studies on predation habits (Dodson and Cooper 1983, Spadinger and Maier 1999, Dendy 1978), little research on control is available.

Biological

There are no known biological control methods for this species. *Craspedacusta sowerbyi* populations are not checked by predation (Jankowski et al. 2005).

Physical

There are no known physical control methods for this species. Hydrozoan hydromedusae blooms are known to be temperature dependent (Ma and Purcell 2005), but polyps and especially the overwintering podocysts are more resistant to varying physical conditions (Peard 2002).

Chemical

There are no known chemical control methods for this species.

A.6 Amphipods

Echinogammarus ischnus (Stebbing, 1899)

Regulations

There are no known regulations for this species.

Control

Biological

Benthic invertebrates including *Echinogammarus ischnus* are a major part of the native yellow perch (*Perca flavescens*) diet (Gonzalez and Burkart 2004). *Echinogammarus ischnus* has also become prey of the invasive Round Goby, *Neogobius melanostomus* (Gonzalez and Burkart 2004). The spread of dreissenid-covered substrate across the Great Lakes region has created an ideal habitat for *Echinogammarus ischnus*, where it is less susceptible to predation, indicating that efforts to control *Dreissena* spp. could also aid control of *Echinogammarus ischnus* (Gonzalez and Burkart 2004).

A parasitic water mold (oomycete) detected in the upper St. Lawrence river is likely responsible for reducing *Echinogammarus ischnus* abundance despite favorable physical and chemical conditions (Kestrup et al. 2011). The oomycete also infects the native amphipod *Gammarus fasciatus*, but its effects are significantly less severe than in *Echinogammarus ischnus* (Kestrup et al. 2011).

Physical

Echinogammarus ischnus can tolerate a maximum water temperature between 31.0°C and 32.2°C before irreversible physiological damage and mortality occur (Wijnhoven et al. 2003).

Electron beam irradiation can be used to control microorganisms in aquatic pathways, including *Echinogammarus ischnus* (USACE 2012b). Electron beam irradiation is a non-selective control

method which exposes water to low doses of radiation using gamma-sterilizers or electron accelerators, breaking down DNA in living organisms while leaving behind no by-products (USACE 2012b). Ultraviolet (UV) light can also effectively control microorganisms including *E. ischnus* in water treatment facilities and narrow channels, where UV filters can be used to emit UV light into passing water, penetrating cell walls and rearranging DNA of microorganisms (USACE 2012b).

Chemical

There are no known chemical control methods for this species

***Gammarus tigrinus* Sexton 1939**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Note: Check federal, state/provincial, and local regulations for the most up-to-date information.

Control

Biological

There are no known biological control methods for this species

Physical

Gammarus tigrinus can tolerate maximum water temperatures between 32.2°C and 34.2°C before irreversible physiological damage and mortality occur (Wijnhoven et al. 2003). In a study of the effects of tide gate operation on hypoxic conditions in the Back River and Savannah River estuaries in Savannah, Georgia, it was found that oxygen saturation levels below 30% are lethal to experimental populations of amphipods, and that *Gammarus tigrinus* is especially susceptible to low dissolved oxygen levels, exhibiting mortality within three hours of exposure to levels between 12 and 18% (Winn and Knott 1992).

Chemical

There are no known chemical control methods for this species.

A.7 Cladocerans

***Bosmina coregoni* Baird, 1857**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

Young-of-year bloaters (*Coregonus hoyi*) have been shown to surface feed on *Eubosmina coregoni* in Lake Michigan, but their effectiveness as a control is unknown, especially because they have been documented moving to the benthos earlier to avoid competition from the invasive Alewife (*Alosa pseudoharengus*) (Crowder and Crawford 1984). *Bythotrephes* spp. are known to consume *E. coregoni* in Russia and the United States (Grigorovich et al. 1998), and *E. coregoni*

populations declined significantly immediately following the invasion of the waterflea *Bythotrephes longimanus* in the Laurentian Great Lakes Michigan, Huron, and Erie, with direct predation by *B. longimanus* the most likely explanation (Barbiero and Tuchman 2004). *E. coregoni* levels in *B. longimanus*-invaded areas have remained at low levels, indicating a significant population-wide impact, but the long-term effectiveness of *B. longimanus* as a biological control method is unknown (Barbiero and Tuchman 2004).

Physical

E. coregoni may be transported over land by recreational boaters (Suchy and Hann 2007). Though it is not as likely to foul gear and attach to equipment as the more commonly known Spiny Waterflea (*Bythotrephes longimanus*) and Fishhook Waterflea (*Cercopagis pengoi*), the same responsible maintenance and cleaning methods are recommended to prevent spread between water bodies, including cleaning all aquatic equipment with high pressure water (>250 psi) or hot water (>50°C) after each use (Ontario's Invading Species Awareness Program). Electron beam irradiation and ultraviolet light treatments have been used to control Spiny and Fishhook Waterfleas in aquatic pathways, and are likely effective against *E. coregoni* (USACE 2012b). Another possible non-selective pathway control is high water turbidity, which may decrease zooplankton (especially cladoceran) abundances due to the negative effects of suspended clay particles on filtering and assimilation rates (Suchy and Hann 2007).

Chemical

E. coregoni is a freshwater cladoceran. Mortality has been documented quickly at salinities $\geq 3\%$ (Nauwerck 1991). Gemza (1995) documented a shift from copepods to cladocerans as dominant zooplankton at increasingly eutrophic sites in Severn Sound, Lake Huron. Zooplankton biomass generally increases with increasing eutrophication, so reduction of excessive nutrient pollution causing abnormal eutrophication could help control *E. coregoni* (Gemza 1995). A study of the effects of cadmium and zinc on Lake Michigan zooplankton found that *E. coregoni* was significantly reduced by separate and combined treatments of 2 $\mu\text{g Cd/L}$ and 100 $\mu\text{g Zn/L}$, with negative effects primarily due to zinc (Marshall et al. 1981). A more recent study on the effects of copper sulfate (used to control algal biomass in eutrophic water bodies) and Carbaryl (used to control aquatic pests) on zooplankton found that levels of 50 $\mu\text{g/L Cu}$ and 20 $\mu\text{g/L Carbaryl}$ individually reduced *E. coregoni* biomass by $\geq 50\%$ (Havens 1994).

Bythotrephes longimanus

Regulations (pertaining to the Great Lakes region)

In Wisconsin, the Spiny Waterflea is a prohibited invasive species ([WI Administrative Code § NR 40.04](#)), which indicates that it is likely to survive and spread if introduced into the state, potentially causing economic or environmental harm or harm to human health ([WI Administrative Code § NR 40.02](#)). With certain exceptions, it is unlawful to transport, possess, transfer or introduce a prohibited invasive species in Wisconsin ([WI Administrative Code § NR 40.04](#)). In Minnesota, the Spiny Waterflea is a regulated invasive species ([MN Administrative Rules § 6216.0260](#)). It is legal to possess, sell, buy, and transport regulated invasive species, but no person may introduce a regulated invasive species without a permit ([MN Administrative Rules § 6216.0265 Subpart 1](#)).

Control

Like the confamilial Fishhook Waterflea (*Cercopagis pengoi*), the Spiny Waterflea is most likely to be spread on aquatic equipment, especially fishing lines. Consequently, public education is a significant method of control which can greatly reduce incidences of species transfer by unaware or incautious anglers (Jacobs and MacIsaac 2007, Lui et al. 2010).

Biological

Bythotrephes longimanus is consumed by rainbow smelt, lake herring, lake hitefish, Yellow Perch, White Perch, White Bass, Walleye, Alewife, Bloater Chub, Emerald Shiner, Spottail Shiner, Deepwater Sculpin, and Chinook Salmon in the Great Lakes (Bur et al. 1986, Makarewicz and Jones 1990, Branstrator and Lehman 1996). *Bythotrephes longimanus*' defensive tailspine has been observed increasing in size throughout the summer in response to predation pressure (Straile and Halbach 2000). Consequently, larger fish are more likely to be successful predators (Branstrator and Lehman 1996). The opossum shrimp (*Mysis relicta*) has been observed eating *B. longimanus* in Ontario lakes, but the frequency of consumption appeared related to abundance of the invader and alternate prey (Nordin et al. 2008).

Physical

Bythotrephes longimanus collects in gelatinous clumps on fishing lines, downrigger cables, and other aquatic equipment (Lui et al. 2010). Responsible maintenance and cleaning methods are recommended to prevent spread between water bodies, including cleaning all aquatic equipment with high pressure (>250 psi) or hot (>50°C) water after each use (Ontario's Invading Species Awareness Program). The acute upper lethal temperature level for *B. longimanus*, at which death occurs rapidly, is 40°C (USACE 2012b), and a study found that *B. longimanus* specifically requires 10 minutes treatment with 43°C water to ensure 100% mortality (Beyer et al. 2011). Fishing lines designed specifically to prevent the spread of waterfleas, such as the Flea Flicker brand, have been proven effective in significantly reducing fouling on lines, indicating their importance as a management tool (Jacobs and MacIsaac 2007).

Electron beam irradiation has been used to control microorganisms in aquatic pathways, including *Bythotrephes longimanus* (USACE 2012b). Electron beam irradiation is a non-selective control method which exposes water to low doses of radiation using gamma-sterilizers or electron accelerators, breaking down DNA in living organisms while leaving behind no by-products (USACE 2012b). Ultraviolet (UV) light can also effectively control microorganisms including *B. longimanus* in water treatment facilities and narrow channels, where UV filters can be used to emit UV light into passing water, penetrating cell walls and rearranging DNA of microorganisms (USACE 2012b).

Chemical

There are no known chemical control methods for this species

Cercopagis pengoi

Regulations (pertaining to the Great Lakes region)

In Wisconsin, the Fishhook Waterflea is a prohibited invasive species ([WI Administrative Code § NR 40.04](#)), meaning that it is unlawful to transport, possess, transfer, or introduce the species

within or into the state without a permit as defined under [Wisconsin Administrative Code § NR 40.06](#).

Control

Cercopagis pengoi is most likely to be spread on aquatic equipment, especially fishing lines. Consequently, public education is a significant method of control which can greatly reduce incidences of species transfer by unaware or incautious anglers (Jacobs and MacIsaac 2007).

Biological

Pothoven et al. (2007) found that adult large Alewives (*Alosa pseudoharengus*) (>100 mm) consume *Cercopagis pengoi* in Lake Michigan, but not significantly enough to control the species, concluding that the Alewife prefers *Bythotrephes longimanus* due to its larger size and conspicuousness. In contrast, a study of *C. pengoi* as a prey item in Lake Ontario found that at least 70% of Alewives larger than 70 mm contained *C. pengoi* spines (Bushnoe et al. 2003). The same study also found spines in Rainbow Smelt (*Osmerus mordax*) stomachs (Bushnoe et al. 2003). Rainbow Smelt historically consume cladocerans in the Great Lakes, but prefer larger prey and may select *Bythotrephes longimanus* over *C. pengoi* where both occur (Pothoven et al. 2009). Gorokhova et al. (2004) found that in the northern Baltic proper, Herring (*Clupea harengus* L.) and Sprat (*Sprattus sprattus* L.) are the dominant predators of *C. pengoi*, and a possible source of biological control through fisheries management, though it is possible that fully mature resting eggs may survive passage through fish digestive systems as has been observed with *B. longimanus* eggs in Yellow Perch (*Perca flavescens*). *B. longimanus* is also known to consume *C. pengoi*, but not as a main prey item (Cavaletto et al. 2010).

Physical

Cleaning all aquatic/fishing equipment, including downrigger lines and monofilament on reels, is important in areas where this species is present. Responsible maintenance and cleaning methods are recommended to prevent spread between water bodies, including cleaning all aquatic equipment with high pressure (>250 psi) or hot (>50°C) water after each use (Ontario's Invading Species Awareness Program). *Bythotrephes longimanus* has been documented spreading by transfer of diapausing eggs on fishing gear, which are more resilient than adult waterfleas (Jacobs and MacIsaac 2007). Because of its similar life history to *B. longimanus*, it is likely that *Cercopagis pengoi* can also be spread by introduction of diapausing eggs to previously uninvaded waters as well as by transfer of fully developed adult specimens (Jacobs and MacIsaac 2007). Fishing lines designed specifically to prevent the spread of waterfleas, such as the Flea Flicker brand, have been proven effective in significantly reducing fouling on lines, indicating their importance as a management tool (Jacobs and MacIsaac 2007).

Electron beam irradiation has been used to control microorganisms in aquatic pathways, including *C. pengoi* (USACE 2012b). Electron beam irradiation is a non-selective control method which exposes water to low doses of radiation using gamma-sterilizers or electron accelerators, breaking down DNA in living organisms while leaving behind no by-products (USACE 2012b). Ultraviolet (UV) light can also effectively control microorganisms, including *C. pengoi*, in water treatment facilities and narrow channels, where UV filters can be used to emit UV light into passing water, penetrating cell walls and rearranging DNA of microorganisms (USACE 2012b).

Chemical

There are no known chemical control methods for this species.

***Daphnia galeata galeata* Sars, 1864**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There is little control information available on a species-specific level for *Daphnia galeata galeata*, especially as related to its presence in North America. However, many control methods used for more commonly known waterfleas may work for *D. g. galeata* as well.

Biological

Various parasites have been shown to reduce host density and population survival in experimental *Daphnia* populations in Europe (Ebert 2005). However, it is unclear whether parasites regulate natural *Daphnia* populations, as all experiments have been completed under lab conditions. Research on *D. g. galeata* is lacking, but many invertebrates are likely predators of *Daphnia* spp. where they occur in Europe and North America, including Great Lakes species such as the predacious phantom midge *Chaoborus flavicans*, the waterfleas *Leptodora* spp. and *Bythotrephes longimanus*, (Lysebo 1995). Many small fish of species such as Yellow Perch (*Perca flavescens*), Three-Spined Stickleback (*Gasterosteus aculeatus*), Alewife (*Alosa pseudoharengus*), Bluegill (*Lepomis macrochirus*), and ciscos (*Coregonus* spp.) have been documented consuming daphnids and other zooplankton in Canada and the United States (Mills and Forney 1983, Post and McQueen 1987, Hulsmann and Mehner 1997), but predation may be insufficient for control in most cases.

Physical

Electron beam irradiation has been used to control microorganisms in aquatic pathways, including *D. g. galeata* (USACE 2012b). Electron beam irradiation is a non-selective control method which exposes water to low doses of radiation using gamma-sterilizers or electron accelerators, breaking down DNA in living organisms while leaving behind no by-products (USACE 2012b). Ultraviolet (UV) light can also effectively control microorganisms including *D. g. galeata* in water treatment facilities and narrow channels, where UV filters can be used to emit UV light into passing water, penetrating cell walls and rearranging DNA of microorganisms (USACE 2012b). Hot water (e.g., boat washing) may be a viable method for killing adults, but resting stages are likely resistant.

Chemical

There are no known chemical control methods for this species.

Daphnia lumholtzi

Regulations (pertaining to the Great Lakes region)

In Wisconsin, *Daphnia lumholtzi* is a prohibited invasive species ([WI Administrative Code § NR 40.04](#)), meaning that it is unlawful to transport, possess, transfer, or introduce the species within or into the state without a permit as defined under [Wisconsin Administrative Code § NR 40.06. Control](#)

Biological

Daphnia lumholtzi is preyed upon by a variety of zooplanktivorous fishes, including Inland Silversides (*Menidia beryllina*) in Lake Texoma (Oklahoma-Texas), Bluegill (*Lepomis macrochirus*), White Bass (*Morone chrysops*), White Crappie (*Pomoxis annularis*), and Black Crappie (*Pomoxis nigromaculatus*) in Lake Chautauqua, Illinois (Lienesch and Gophen 2001, Lemke et al. 2003). The degree to which these fishes may be able to control *D. lumholtzi* populations is not certain.

Physical

D. lumholtzi is likely transferred through anthropogenic vectors, including on recreational boats (Frisch et al. 2013). Its resting eggs (ephippia) feature a long point and hairs on the margin that serve as hooks, possibly aiding in attachment to boats or macrophytes caught on boats (Dzialowski et al. 2000). Therefore, the same precautions recommended to prevent the spread of more commonly known waterfleas should be taken to help prevent *D. lumholtzi* distribution, namely cleaning all aquatic/fishing/boating equipment, including downrigger lines and monofilament on reels, using high pressure (>250 psi) or hot (>50°C) water after each use (OFAH n.d.).

While *D. lumholtzi* is not listed as a target species by the United States Army Corps of Engineers Great Lakes and Mississippi River Interbasin Study (USACE GLMRIS), the methods suggested to control Spiny and Fishhook Waterfleas in aquatic pathways would likely control *D. lumholtzi* as well. Electron beam irradiation has been used to control microorganisms in aquatic pathways by exposing water to low doses of radiation using gamma-sterilizers or electron accelerators, breaking down DNA in living organisms while leaving behind no by-products (USACE 2012b). Ultraviolet (UV) light can also effectively control microorganisms, in water treatment facilities and narrow channels, where UV filters can be used to emit UV light into passing water, penetrating cell walls and rearranging DNA of microorganisms (USACE 2012b).

Chemical

There are no known chemical control methods for this species.

***Eubosmina maritima* P.E. Müller, 1867**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Note: Check federal, state/provincial, and local regulations for the most up-to-date information.

Control

Biological

The mysid shrimps, *Mysis mixta* and *Mysis relicta*, consume *Eubosmina maritima* diapausing eggs (ephippia) selectively in the northern Baltic Sea, but only at a rate which can affect local abundances of *E. maritima* (Viitasalo and Viitasalo 2004).

Physical

It is possible that *Eubosmina maritima* is transported over land by recreational boaters. Though it is not as likely to foul gear and attach to equipment as the more commonly known Spiny Waterflea (*Bythotrephes longimanus*) and Fishhook Waterflea (*Cercopagis pengoi*), the same responsible maintenance and cleaning methods are recommended to prevent spread between water bodies, including cleaning all aquatic equipment with high pressure water (>250 psi) or hot water (>50°C) after each use (OFAH n.d.). Electron beam irradiation and ultraviolet light treatments have been used to control Spiny and Fishhook Waterfleas in aquatic pathways, and are likely effective against *E. maritima* (USACE 2012b). Another possible non-selective pathway control is high water turbidity, which may decrease zooplankton (especially Cladoceran) abundances due to the negative effects of suspended clay particles on filtering and assimilation rates (Suchy and Hann 2007).

Chemical

Santagata et al. (2008) found that ballast water exchange methods which flush freshwater organisms into euhaline seawater are effective against *Eubosmina maritima* at a minimum of 24 PSU (practical salinity units) for two hours in a laboratory simulation. However, *E. maritima* ephippia may remain in residual unpumpable sediment in ballast tanks, and Gray et al. (2005) found that exposing zooplankton ephippia to open ocean saline water of 32 ppt (parts per thousand) did not reduce egg abundances or consistently affect richness of invertebrates hatched from exposed eggs.

Zooplankton biomass generally increases with increasing eutrophication, so reduction of excessive nutrient pollution causing abnormal eutrophication could help control *E. coregoni* (Gemza 1995).

A.8 Copepods

Argulus japonicus Thiele, 1900

Gizzard Shad (*Dorosoma cepedianum*) is a host fish (Poly, 1998) as well as Yellow Perch (*Perca flavescens*), Koi Carp (*Cyprinus carpio*), and Goldfish (*Carassius auratus*) (Lesko et al. 2003). *Cyclops strenuus* Fischer, 1851

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

Treasurer (1992) found that Eurasian perch (*Perca fluviatilis*) larvae selectively prey on *Cyclops strenuus abyssorum* in the Scottish lochs Kinord and Davan. The total zooplankton reduction observed was minimal, but Treasurer suggested that grazing by larvae is likely to impact copepod populations (1992).

Physical

There are no known physical control methods for this species.

Chemical

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of copepods. It should be noted that the effectiveness of these methods is likely significantly diminished against copepod resting stages.

***Eurytemora affinis* Poppe, 1880**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

Maes et al. (2005) found that juvenile Herring (*Clupea harengus*) and Sprat (*Sprattus sprattus*) exhibit top down control of *Eurytemora affinis* through predation pressure in the Scheldt estuary in Belgium.

Physical

There are no known physical control methods for this species.

Chemical

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of copepods. It should be noted that the effectiveness of these methods is likely significantly diminished against copepod ephippia. Lindley et al. (1999) found that exposure to the organochlorine compounds pentachlorophenol (PCP) and 1, 2-dichlorobenzene (DCB), (both common industrial pollutants) accumulated in sediment significantly reduced hatching success and nauplii viability of *E. affinis* eggs. In a study of the effect of salinity on toxicity of cadmium to Chesapeake Bay organisms, Hall et al. (1995) found that *E. affinis* is very sensitive to cadmium compared to other estuarine aquatic biota. The study documents 96 h LC50 (lethal concentration to 50% of organisms tested) values of 51.6, 213.2, and 82.9 $\mu\text{g L}^{-1}$ at 5, 15, and 25 ppt (parts per thousand) salinities, respectively (Hall et al. 1995). Sullivan et al. observed a 96 h LC50 of $>120 \mu\text{g L}^{-1}$ for cadmium and $\sim 30 \mu\text{g L}^{-1}$ for copper on *E. affinis* at 10 ppt salinity (1983). Sullivan et al. (1983) also noted that reduced

growth rates of *E. affinis* occur at Cu and Cd doses below the 96 h LC50, which has been documented as extending generation length and eventually reducing population size in previous studies. The negative effects of the insecticide diflubenzuron (Dimilin®) on *E. affinis* nauplii were documented by Savitz and Wright, who noted a 48 h LC50 of 2.2 µg L⁻¹. The insecticide is approved for use against the gypsy moth (*Lymantria dispar*) and other insect pests by the United States Environmental Protection Agency (USEPA), and enters *E. affinis* habitat through runoff or by direct spraying (Savitz and Wright 1994). Diflubenzuron specifically targets the arthropod molting process, so the most explicit effects are expected in sub-adult crustaceans. *E. affinis* was lethally affected at levels as low as 0.78 µg L⁻¹ (Savitz and Wright 1994).

***Heteropsyllus nr. nunni* Coull**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of copepods. It should be noted that the effectiveness of these methods is likely significantly diminished against copepod resting stages.

***Megacyclops viridis* Jurine, 1820**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

Megacyclops viridis is an important prey item for introduced Ruffe (*Gymnocephalus cernuus*), but the Ruffe feeds on a wide variety of benthic organisms, so its feasibility as a biocontrol is unknown (Ogle et al. 1995). Yellow Perch (*Perca flavescens*) and trout perch (*Percopsis omiscomaycus*) are potential predators of *M. viridis* (Ogle et al. 1995). Hansen and Jeppesen (1992) found that a 50% reduction of planktivorous fish biomass (roach, *Rutilus rutilus*, and bream, *Abramis brama*) affected cyclopoid copepod population directly through reduction in fish predation pressure and changes in biological structure of Lake Væng, Denmark.

Physical

There are no known physical control methods for this species. *M. viridis* is tolerant to salinity of a wide range, up to 7.9 g L⁻¹ in one study (Wolfram et al. 1999).

Chemical

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of copepods. It should be noted that the effectiveness of these methods is likely significantly diminished against copepod ephippia.

***Neoergasilus japonicus* Harada, 1930**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

Electron beam irradiation can be used to control microorganisms in aquatic pathways, including *Neoergasilus japonicus* (USACE 2012b). Electron beam irradiation is a non-selective control method which exposes water to low doses of radiation using gamma-sterilizers or electron accelerators, breaking down DNA in living organisms while leaving behind no by-products (USACE 2012b). Ultraviolet (UV) light can also effectively control microorganisms including *N. japonicus* in water treatment facilities and narrow channels, where UV filters can be used to emit UV light into passing water, penetrating cell walls and rearranging DNA of microorganisms (USACE 2012b).

Chemical

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of copepods. It should be noted that the effectiveness of these methods is likely significantly diminished against copepod resting stages.

***Nitokra hibernica* Brady, 1880**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Ballast exchange regulations for international vessels entering the Great Lakes are highly efficient at preventing introduction of international nonindigenous species (NIS), but Briski et al. have noted that a lack of ballast regulations in domestic shipping within the Great Lakes poses a threat to further spread of NIS between lakes (2012). Specifically, they sampled *Nitokra hibernica* from ballast water destined for discharge in Lake Superior, where it has not yet been

documented. They suggest domestic regulations be based on ecological boundaries rather than geographic and political borders to help prevent spread of NIS within the Great Lakes (Briski et al. 2012).

Control

Biological

Nitokra hibernica has been found in the stomach of Slimy Sculpin (*Cottus cognatus*) at Yankee Reef, Lake Huron, and it is known to be consumed by Rainbow Smelt (*Osmerus mordax*) in Lake Huron, but the significance of this predation on biological control is unknown (Lesko et al. 2003).

Physical

There are no known physical control methods for this species.

Chemical

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of copepods. It should be noted that the effectiveness of these methods is likely significantly diminished against copepod resting stages.

Nitokra incerta Richard, 1893

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species

Physical

There are no known physical control methods for this species.

Chemical

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of copepods. It should be noted that the effectiveness of these methods is likely significantly diminished against resting stages.

Salmincola lotae Olsson, 1877

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species

Physical

There are no known physical control methods for this species.

Chemical

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of copepods. It should be noted that the effectiveness of these methods is likely significantly diminished against resting stages.

***Schizopera borutzkyi* Monchenko, 1967**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

Schizopera borutzkyi is found in shallow muds and sands at a temperature of 21°C and pH 7.6 in its native habitat of the Black Sea Danube River delta, and can tolerate a wide variety of salinities (0.04-6%), but there is no research available at this point on the significance of these parameters to control (Horvath et al. 2001).

Electron beam irradiation can be used to control microorganisms in aquatic pathways, including *S. borutzkyi* (USACE 2012b). Electron beam irradiation is a non-selective control method which exposes water to low doses of radiation using gamma-sterilizers or electron accelerators, breaking down DNA in living organisms while leaving behind no by-products (USACE 2012b). Ultraviolet (UV) light can also effectively control microorganisms, including *S. borutzkyi*, in water treatment facilities and narrow channels, where UV filters can be used to emit UV light into passing water, penetrating cell walls and rearranging DNA of microorganisms (USACE 2012b).

Chemical

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of copepods. It should be noted that the effectiveness of these methods is likely significantly diminished against resting stages.

***Skistodiaptomus pallidus* Herrick, 1879**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of copepods. It should be noted that the effectiveness of these methods is likely significantly diminished against resting stages.

A.9 Mysids

***Hemimysis anomala* G.O. Sars, 1907**

Regulations (pertaining to the Great Lakes region)

In Wisconsin, *Hemimysis anomala* is a prohibited invasive species ([WI Administrative Code § NR 40.04](#)), meaning that it is unlawful to transport, possess, transfer, or introduce the species within or into the state without a permit as defined under [Wisconsin Administrative Code § NR 40.06](#).

Control

Biological

In Lake Ontario, *H. anomala* has been documented in the stomachs of Alewife (*Alosa pseudoharengus*), Rock Bass (*Ambloplites rupestris*), Yellow Perch (*Perca flavescens*), and White Perch (*Morone americana*) (Brooking et al. 2010, Lantry et al. 2010). Alewives were the only predators exhibiting significant consumption of *H. anomala*, likely due to their nocturnal feeding habits (*H. anomala* exhibits diel vertical migration, remaining near the lakebed during the day and emerging at night), and their prior experience consuming the Great Lakes native mysid, *Mysis relicta*, which exhibits similar swimming behavior to *H. anomala* (Lantry et al. 2010). Lantry et al. (2010) suggest that as the density and spatial distribution of *H. anomala* expands, more Great Lakes fishes will become successful predators. Round Goby (*Apollonia melanostoma*) are accomplished molluscivores, but have also demonstrated specific predatory behavior enabling them to consume *M. relicta*, and may become significant predators of *H. anomala* in the absence of dreissenids (Lantry et al. 2010). *H. anomala* has also been documented in Yellow Perch and White Perch diets in Lake Oneida, New York (Brooking et al. 2010). The significance of fish predation on control of *H. anomala* is currently unknown, but there is potential for adaptation towards consumption among Great Lakes zooplanktivores, especially because it is considered a high-energy food source (Borcherding et al. 2006).

Physical

H. anomala exhibits mortality at temperatures below 0° C (Borcherding et al. 2006). Because *H. anomala* is a new, quickly spreading invasive species, preventative measures are encouraged for boaters traveling between water bodies, including visually inspecting boats, trailers, and equipment for plants, animals, and mud after each use, draining water from the motor, live well, bilge, and transom wells while on land, and rinsing all equipment with high pressure (>250 psi) or hot (>50°C) water (OFAH n.d.).

Electron beam irradiation can be used to control microorganisms in aquatic pathways, including *H. anomala* (USACE 2012b). Electron beam irradiation is a non-selective control method which exposes water to low doses of radiation using gamma-sterilizers or electron accelerators, breaking down DNA in living organisms while leaving behind no by-products (USACE 2012b). Ultraviolet (UV) light can also effectively control microorganisms including *H. anomala* in water treatment facilities and narrow channels, where UV filters can be used to emit UV light into passing water, penetrating cell walls and rearranging DNA of microorganisms (USACE 2012b).

Chemical

H. anomala tolerates salinity of 0-19 ppt (parts per thousand) (Bij de Vaate et al. 2002). Ellis and MacIsaac (2009) tested the salinity tolerance of Great Lakes Invaders in ballast water exchange (BWE) simulations. They documented 100% mortality for *H. anomala* after five hours in a simultaneous BWE treatment, in which salinity was gradually increased from 4-30 ppt, and 100% mortality after three hours in a sequential BWE treatment, in which species are immediately exposed to 30 ppt salinity.

The Great Lakes and Mississippi River Interbasin Study (USACE 2012b) suggests that alteration of water quality using carbon dioxide, ozone, nitrogen, and/or sodium thiosulfate could be effective in preventing upstream and downstream movement of crustaceans.

A.10 Fishes

***Alosa aestivalis* Mitchill, 1814**

Regulations (pertaining to the Great Lakes region)

New York, under federal law, is required to follow the Interstate Fishery Management Plan for Shad and River Herring established by the Atlantic States Marine Fisheries Commission ([Enacted May 2009](#); ASMFC) and, therefore, are forced to close any non-sustainable commercial and recreational fisheries by January 1, 2012 until the Department can prove New York fisheries are self-sustaining (NYSDEC 2012b). Management plans are still in review and final amendments for each New York fisheries is unknown. New York restricts the use of anadromous river Herring (including Blueback) as bait in most waters ([6 NYCRR § Part 19](#)). While not listed by name, in Ohio it is illegal for any person to possess, import or sell exotic species of fish (including *Alosa aestivalis*) or hybrids thereof for introduction or to release into any body of water that is connected to or otherwise drains into a flowing stream or other body of water that would allow egress of the fish into public waters, or waters of the state, without first having obtained permission ([Ohio Administrative Code § 1501:31-19](#)). Ontario takes a whitelist

approach to bait under which all unlisted species (including *Alosa aestivalis*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Most management research relating to *Alosa aestivalis* appears focused on maintaining populations within the native range. Little species-specific information is available on how to control this species where it is invasive.

Biological

There are no known biological control methods for this species. Top-down control by salmonids is effective for the related species Alewife, *Alosa pseudoharengus*.

Physical

There are no known physical control methods for this species.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered “general” piscicides, but no studies have been found of their effects on *Alosa aestivalis* (USACE 2012b).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

***Alosa pseudoharengus* Wilson, 1811**

Regulations (pertaining to the Great Lakes region)

Alosa pseudoharengus is a regulated invasive species in Minnesota ([MN Administrative Rules § 6216.0260](#)). New York restricts the use of Alewife as bait in most waters ([6 NYCRR § Part 19](#)). While not listed by name, in Ohio it is illegal for any person to possess, import or sell exotic species of fish (including *Alosa pseudoharengus*) or hybrids thereof for introduction or to release into any body of water that is connected to or otherwise drains into a flowing stream or other body of water that would allow egress of the fish into public waters, or waters of the state, without first having obtained permission ([Ohio Administrative Code Chapter 1501:31-19](#)).

Ontario takes a whitelist approach to bait under which all unlisted species (including *Alosa pseudoharengus*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

The management response to Great Lakes Alewife overabundance and recurring die-offs was to invest in Sea Lamprey (*Petromyzon marinus*) control and planting of hatchery-reared Pacific salmonids (*Oncorhynchus* spp.) to re-establish top open-water predators (Kocik and Jones 1999, Hansen and Holey 2002). Older and larger fish tend to be most heavily affected by piscivores, while smaller and younger fish remain abundant (Hewett and Stewart 1989). Alewives are now managed in part to support the valuable salmonid fishery.

Physical

There are no known physical control methods for this species.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered “general” piscicides (USACE 2012b).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

***Apeltes quadracus* Mitchill, 1815**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species. Ontario takes a whitelist approach to bait under which all unlisted species (including *Apeltes quadracus*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods for this species.

Physical

Netting can be used method of control for some systems, though small size of these fish might limit the viability of this option.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered “general” piscicides, but no studies have been found of their effects on *Apeltes quadracus* (USACE 2012b).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fishes with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Boogaard et al. (2003) found that the lampricides 3-trifluoromethyl-4-nitrophenol (TFM) and 2',5-dichloro-4'-nitrosalicylanilide (niclosamide) demonstrate additive toxicity when combined. In another study on cumulative toxicity, combinations of Bayer 73 (niclosamide) and TFM with contaminants common in the Great Lakes (pesticides, heavy metals, industrial organics, phosphorus, and sediments) were found to be mostly additive in toxicity to Rainbow Trout, and one combination of TFM, Delnav, and malathion was synergistic, with toxicity magnified 7.9 times (Marking and Bills 1985). This highlights the need for managers to conduct on-site toxicity testing and to give serious consideration to determining the total toxic burden to which organisms may be exposed when using chemical treatments (Marking and Bills 1985). Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

***Carassius auratus* Linnaeus, 1758**

Regulations (pertaining to the Great Lakes region)

Canadian federal law dictates that no person shall use as bait, or possess for use as bait, in any province any live or dead Goldfish ([Maritime Provinces Fishery Regulations § SOR 93-55](#)). Provincial laws in Ontario and Quebec also state that Goldfish are not to be used as bait ([Ontario Fishery Regulations § SOR/2007-237](#) and [Quebec Fishery Regulations § SOR 90/214](#)).

In the state of New York, it is illegal to use or sell Goldfish larvae for bait, and Goldfish larvae taken in nets operated pursuant to baitfishing are to be destroyed immediately ([NY Environmental Conservation Law § 11-1315](#)). In Minnesota, Goldfish are a regulated invasive species, making introduction of the species without a permit illegal ([MN Administrative Rules §](#)

[6216.0260](#), [MN Administrative Rules § 6216.0265](#)). In the state of Pennsylvania, it is unlawful to use or possess Goldfish as baitfish while fishing ([58 PA Code § 63.44](#)). In the state of Wisconsin, Goldfish are a restricted invasive species ([WI Administrative Code § NR 40.05](#)).

Control

Biological

There are no known biological control methods for this species.

Physical

Yamamoto et al. (2006) noted that physical drawdown of water levels has significant negative effects on cyprinid spawning abilities in Lake Biwa, Japan. *Carassius* spp. and *Cyprinus carpio* eggs were notably reduced after collection when water levels were lowered by 30 cm, and as little as a 10 cm reduction can significantly reduce available shallow, litter-accumulated spawning areas preferred by cyprinids (Yamamoto et al. 2006). Potential impacts to native species may be significant. This species may be managed by removal (Morgan et al. 2005), although this would have to be an ongoing program.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered “general” piscicides (USACE 2012b).

Goldfish are somewhat more resistant to antimycin A than other fishes (Lennon and Berger 1964), thus higher concentrations may be required and potential kill of native fishes should be given careful consideration. Potassium permanganate (KMnO₄) is effective for detoxifying antimycin in laboratory waters of pH 6.5 to 9.5. The half-life for antimycin exposed to 1.0 mg/liter of KMnO₄ ranges from 7 to 11 minutes in the different pH waters at 12 C. The 96-h LC₅₀'s for potassium permanganate (KMnO₄) is 3.60 mg/liter with Goldfish, *Carassius auratus*, in laboratory tests. The toxicity of KMnO₄ to fish is greatest in water of lower temperatures, in harder water, or in higher pH water (Marking and Bills 1975). Treating with antimycin A followed by detoxification with potassium permanganate allows restocking within 2 hours (Gilderhus et al. 1981).

Relative to other fish species, Goldfish are highly resistant to rotenone. They were the most resistant of 21 species tested by Marking and Bills (1976) and second only to Yellow Bullhead in Turner's study (1959).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fishes with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008). Liming has also been used to control Goldfish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. This is especially true for Goldfish, which demonstrate a high

resistance to most chemical controls and tolerance for degraded environmental conditions. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Boogaard et al. (2003) found that the lampricides 3-trifluoromethyl-4-nitrophenol (TFM) and 2',5-dichloro-4'-nitrosalicylanilide (niclosamide) demonstrate additive toxicity when combined. In another study on cumulative toxicity, combinations of Bayer 73 (niclosamide) and TFM with contaminants common in the Great Lakes (pesticides, heavy metals, industrial organics, phosphorus, and sediments) were found to be mostly additive in toxicity to Rainbow Trout, and one combination of TFM, Delnav, and malathion was synergistic, with toxicity magnified 7.9 times (Marking and Bills 1985). This highlights the need for managers to conduct on-site toxicity testing and to give serious consideration to determining the total toxic burden to which organisms may be exposed when using chemical treatments (Marking and Bills 1985). Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly. Goldfish are particularly tolerant of low oxygen levels and most other species would be expected to die first.

***Cyprinus carpio* Linnaeus, 1758**

Regulations (pertaining to the Great Lakes region)

Cyprinus carpio are a regulated invasive species in Minnesota, making introduction of the species without a permit illegal ([MN Administrative Rules § 6216.2060](#), [MN Administrative Rules § 6216.0265](#)). In Wisconsin, *Cyprinus carpio* is a “restricted species” under [NR 40](#). “Restricted species are also subject to a ban on transport, transfer and introduction, but possession is allowed, with the exception of fish and crayfish.” Carp may not be collected or used for bait in New York or Pennsylvania ([NY Environmental Conservation Law § 11-1315](#); [58 PA Code § 63.44](#)). Ontario takes a whitelist approach to bait under which all unlisted species (including *Cyprinus carpio*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Management and control of Common Carp has been well documented through much of

North America (Meronek et al. 1996, Wydoski and Wiley 1999) with millions of dollars invested on research and control (Pimentel et al. 2000).

Biological

Northern Pike, *Esox Lucius*, have been used as a biological tool to control Common Carp recruitment in the Sandhill lakes in Nebraska (Paukert et al. 2003). Spring Viremia of Carp (SVC) has been suggested as a control of Common Carp in Australia. However, releasing water-borne viral control agents would likely lead to serious disagreement in the scientific and management communities, along with the general public due to potential impacts to nontarget species (Koehn et al. 2000). Inducible Fatality Genes (IFG) involve breeding carp with a fatal genetic weakness to a trigger substance, such as zinc. The fatal gene technology appears to be a potentially viable and long-term strategy for the environmentally benign control of carp (Koehn et al. 2000).

Physical

Removal projects included mechanical harvest by netting (Ritz 1987, Pinto et al. 2005), water level manipulation to disrupt spawning (Summerfelt 1999), and exclusion from spawning habitat (Lougheed and Chow-Fraser 2001).

Common Carp display jumping behavior when trying to escape entrapment. The Williams cage exploits this behavior by selectively removing the jumping carp from other fishes. Tests of the Williams cage in Australia proved to be extremely successful. Over the three year testing, the Williams cage successfully separated 88% of adult Common Carp and allowed 99.9% native species to pass through. The Williams cage is useful in controlling dispersal and abundance of Common Carp.

Barriers including electric, bubble curtain, and sonic have been used to exclude carp from industrial cooling intake structures (Koehn et al. 2000). Harvesting is only effective if carp are of importance by fisheries and anglers. Even if carp are beneficial for harvest, this method is one of the least effective methods available (Linfield 1980, Vacha 1998, Koehn et al. 2000, Wedekind et al. 2001, Arlinghaus and Mehner 2003). Other physical control methods include traps and water level manipulation. Seasonal movements of Common Carp can be exploited to enhance management actions, such as removal or interrupting spawning (Taylor et al. 2012). When possible, carp can be excluded from an area and then kept out through sorting of fish, which has been done since 1997 at the Cootes Paradise Marsh in Hamilton, Ontario (Lougheed et al. 2004). Although labor intensive, this method is effective at keeping carp from returning to the marsh.

Chemical

Rotenone is a widely used non-selective chemical used to eliminate Common Carp from a water body (Koehn et al. 2000, Sorensen and Stacey 2004, Fajt and Grizzle 1998).

Antimycin-impregnated baits have been used to target Common Carp (Rach et al. 1994, Clearwater et al. 2008). The bait pellets consisted of fish meal, a binding agent, antimycin and water. Doses of 10 mg antimycin/g bait caused low (19%) to high (74%) mortalities in fish feeding voluntarily on 50 g of the toxic bait in each of three earthen ponds.

In laboratory trials, a combination of pH 6.5 and 642 mg/L NaHCO₃ was the most effective treatment for Rainbow Trout, Brook Trout, and Common Carp, causing the fish to cease locomotion and slowing opercular rate within five minutes (Brooke et al. 1978).

Application of different pheromones such as migratory, alarm, and sex may be useful in the integrated management of carp (Sorensen and Stacey 2004).

Enneacanthus gloriosus Holbrook, 1855

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species. Ontario takes a whitelist approach to bait under which all unlisted species (including *Enneacanthus gloriosus*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods specific to this species. Top-down control (trophic cascade) may be effective for control as it is for native sunfishes.

Physical

There are no known physical control methods for this species.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered “general” piscicides, but no studies have been found of their effects on *Enneacanthus gloriosus* (USACE 2012b).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fishes with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

Gonzalez and Dunson (1989) found that *Enneacanthus gloriosus* exposed to pH 4.25 and 4.0 grew at a lower rate (30-40% less growth) than fish of the same species exposed to higher pH (4.5, 5.0, 5.8). Survival of *E. gloriosus* was reduced at pH 3.5 and eliminated at pH 3.25, and the authors concluded that *E. gloriosus* would be excluded from habitats that regularly drop below pH 4.0 (Gonzalez and Dunson 1989).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Boogaard et al. (2003) found that the lampricides 3-trifluoromethyl-4-nitrophenol (TFM) and 2',5-dichloro-4'-nitrosalicylanilide (niclosamide) demonstrate additive toxicity when combined. In another study on cumulative toxicity, combinations of Bayer 73 (niclosamide) and TFM with contaminants common in the Great Lakes (pesticides, heavy metals, industrial organics, phosphorus, and sediments) were found to be mostly additive in toxicity to Rainbow Trout, and one combination of TFM, Delnav, and malathion was synergistic, with toxicity magnified 7.9 times (Marking and Bills 1985). This highlights the need for managers to conduct on-site toxicity testing and to give serious consideration to determining the total toxic burden to which organisms may be exposed when using chemical treatments (Marking and Bills 1985). Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

Esox niger Lesueur, 1818

Regulations (pertaining to the Great Lakes region)

The sale of dead Chain Pickerel is prohibited in **Quebec** under the [Quebec Regulation Respecting Aquaculture and the Sale of Fish § RRQ, c C-61.1, r 7](#). Ontario takes a whitelist approach to bait under which all unlisted species (including *Esox niger*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods for this species.

Physical

The Maine Department of Inland Fisheries and Wildlife cites removal of fishing bag limits for Chain Pickerel as a management measure in certain lakes, noting that the rationale is biologically sound but negligibly effective because few anglers take advantage of the law and capture more than 10 pickerel per day (Brokaw 2008).

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered “general” piscicides (USACE 2012b).

Esox niger exhibited partial mortality in trials ranging from 0.4-12.1ppb antimycin (Lennon and Berger 1970).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fishes with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Boogaard et al. (2003) found that the lampricides 3-trifluoromethyl-4-nitrophenol (TFM) and 2',5-dichloro-4'-nitrosalicylanilide (niclosamide) demonstrate additive toxicity when combined. In another study on cumulative toxicity, combinations of Bayer 73 (niclosamide) and TFM with contaminants common in the Great Lakes (pesticides, heavy metals, industrial organics, phosphorus, and sediments) were found to be mostly additive in toxicity to Rainbow Trout, and one combination of TFM, Delnav, and malathion was synergistic, with toxicity magnified 7.9 times (Marking and Bills 1985). This highlights the need for managers to conduct on-site toxicity testing and to give serious consideration to determining the total toxic burden to which organisms may be exposed when using chemical treatments (Marking and Bills 1985). Other non-selective alterations of

water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

Gambusia affinis Baird and Girard, 1853

Regulations (pertaining to the Great Lakes region)

The Western Mosquitofish is prohibited in **Wisconsin** under [Wisconsin Administrative Code § NR 40.04](#), meaning that no person may transport, possess, transfer, or introduce the species without authorization. Ontario takes a whitelist approach to bait under which all unlisted species (including *Gambusia affinis*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered “general” piscicides, but no studies have been found of their effects on *Gambusia affinis* (USACE 2012b).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

Gymnocephalus cernua Linnaeus, 1758

Regulations (pertaining to the Great Lakes region)

Aquarium fish-keeping, production, keeping in captivity, breeding, stocking, transport, sale, or purchase of live Ruffe is prohibited in **Quebec** under the [Quebec Regulation Respecting Aquaculture and the Sale of Fish § RRQ, c C-61.1, r 7](#). In **Ontario**, Ruffe is an invasive fish

under [Ontario Fishery Regulations § SOR/2007-237](#), and, therefore, may not be possessed without a license and shall not be used or possessed for use as baitfish.

In **Pennsylvania**, it is unlawful to possess live Ruffe or to import or introduce live Ruffe to Pennsylvania waters under [58 PA Code § 71.6](#). It is unlawful to sell, purchase, offer for sale or barter for live Ruffe under [58 PA Code § 63.46](#). Ruffe may not be transported from another state, province, or country into Pennsylvania, liberated into a Pennsylvania watershed, or transferred between Pennsylvania waters without written permission from the Pennsylvania Fish and Boat Commission under [58 PA Code § 73.1](#). In **Ohio**, it is unlawful to possess, import, or sell live individuals of Ruffe except for research, education, or public display when authorized ([Ohio Administrative Code § 1501:31-19-01](#)). In **Michigan**, Ruffe is a prohibited species under [Michigan NREPA 451 § 324.41301](#). No person shall knowingly possess a live prohibited organism in Michigan except for education, research, or identification purposes as listed in Michigan [NREPA 451 § 324.41303](#). It is also unlawful to introduce prohibited organisms in Michigan under [Michigan NREPA 451 § 324.41305](#). In Michigan, a violation involving a prohibited species is a felony, and a knowing introduction violation with intent to harm is punishable with up to five years imprisonment and a \$2,000 to \$1,000,000 fine ([MI NREPA § 324.41309](#)). In **Indiana**, Ruffe is classified as an exotic fish under [312 Indiana Administrative Code 9-6-7](#), meaning except as otherwise provided, no individual can import, possess, propagate, buy, sell, barter, trade, transfer, loan, or release into public or private waters live fish, recently hatched juveniles, viable eggs, or genetic material. In **Illinois**, Ruffe is listed as an injurious species under [Illinois Administrative Code 17 § 805.20](#). It is unlawful to possess, propagate, buy, sell, barter, or offer to be bought, sold, bartered, transported, traded, transferred, or loaned an injurious species to any person or institution unless a permit is obtained from the Illinois DNR ([IL Administrative Code 17 § 805.30](#)). In **Wisconsin**, Ruffe is a restricted species as an established non-native, and therefore cannot be transported, possessed, transferred, or introduced without a permit ([WI Administrative Code § NR 40.05](#)). In **Minnesota**, Ruffe is a prohibited invasive species, meaning it is unlawful (a misdemeanor) to possess, import, purchase, transport, or introduce an organism except under permit for control, research, or education ([MN Administrative Rules § 6216.0250](#)).

Control

Biological

Minnesota and Wisconsin, with advice from the United States Fish and Wildlife Service, implemented a top-down control program for Ruffe in the St. Louis River, western Lake Superior, in 1989, using Northern Pike (*Esox lucius*), Walleye (*Sander vitreus*), Smallmouth Bass (*Micropterus dolomieu*), Brown Bullhead (*Ameiurus nebulosus*), and Yellow Perch (*Perca flavescens*) (Mayo et al. 1998). A bioenergetics modeling evaluation of the top-down control program revealed that although predators ate as much as 47% of Ruffe biomass in one year, they avoided Ruffe and were selective for native prey, and were thus unable to halt the increase in Ruffe abundance (Mayo et al. 1998). However, the authors noted that Northern Pike and Walleye appeared to have potential for top-down control of Ruffe due to a combination of their diets and population sizes, and due to indications that they may learn to prey more selectively on Ruffe (Mayo et al. 1998).

As Mayo et al. (1998) noted, caution is advised when considering top-down biological control as a management tool because the stability properties of a system do not just depend on predation, but also on the life histories of component species and their interactions.

Physical

There are no known physical control methods for this species.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered general piscicides, but no studies have been found of their effects on *Oncorhynchus nerka* (USACE 2012b).

Evaluation of the effects of common piscicides on Ruffe revealed that the lampricide 3-trifluoromethyl-4-nitrophenol (TFM) has potential for selective control of the species (Boogaard et al. 1996). Ruffe was three to six times more sensitive to TFM than both Yellow Perch (*Perca flavescens*) and Brown Trout (*Salmo trutta*) (Boogaard et al. 1996). Toxicity tests in May and August 1992 on the Brule River, Wisconsin revealed a 12h LC99.9 (concentration at which 99.9% of organisms are killed after 12 hours) of 5.9 mg/L at normal pH levels (~8.4) and 2.80 mg/L at low pH levels (Boogaard et al. 1996). Furthermore, at low pH levels (7.7-7.9) 12h LC25's of 7.2 mg/L and 4.6 mg/L were recorded for Yellow Perch and Brown Trout, respectively, but at normal pH levels no Brown Trout or Yellow Perch mortality was recorded at the highest tested concentration of 8.8 mg/L (Boogaard et al. 1996). A cost benefit analysis of a United States Ruffe control program supported TFM as a promising chemical control (Leigh 1998). However, Dawson et al. (1998) suggest that TFM may have more application for treating entire bodies of water rather than localized areas because it tended to repel Ruffe in preference tests, allowing them to move to untreated areas. Bottom-release formulations of bayluscide and antimycin showed promise for effectiveness in treating localized concentrations of Ruffe, but more field testing is needed (Dawson et al. 1998).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Boogaard et al. (2003) found that the lampricides 3-trifluoromethyl-4-nitrophenol (TFM) and 2',5-dichloro-4'-nitrosalicylanilide (niclosamide) demonstrate additive toxicity when combined. In another study on cumulative toxicity, combinations of Bayer 73 (niclosamide) and TFM with contaminants common in the Great Lakes (pesticides, heavy metals, industrial organics, phosphorus, and sediments) were found to be mostly additive in toxicity to Rainbow Trout, and one combination

of TFM, Delnav, and malathion was synergistic, with toxicity magnified 7.9 times (Marking and Bills 1985). This highlights the need for managers to conduct on-site toxicity testing and to give serious consideration to determining the total toxic burden to which organisms may be exposed when using chemical treatments (Marking and Bills 1985). Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

Ictiobus cyprinellus Valenciennes in Cuvier and Valenciennes, 1844

Ditching and draining for farmlands, which eliminated shallow lakes, may have reduced Bigmouth Buffalo populations within their native range in the first half of the 20th century. The species is not listed as threatened or endangered in any region of its native or introduced distribution.

Regulations (*pertaining to the Great Lakes region*)

Commercial harvests are regulated in many states. Although rarely caught via hook and line, state fishing regulations may apply. Ontario takes a whitelist approach to bait under which all unlisted species (including *Ictiobus cyprinellus*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species

Chemical

There are no known chemical control methods specific to this species. General piscicides (such as rotenone) may be used for control, but expect significant kill of non-target species.

Lepisosteus platostomus Rafinesque, 1820

Regulations (*pertaining to the Great Lakes region*)

In Ohio, a class B aquaculture permit is required to engage in propagation, culture, or sale of Shortnose Gar, and two levels of escapement prevention are required if cultured in the Lake Erie drainage basin (Ohio Administrative Code § 1501:31-39-01). Ontario takes a whitelist approach to bait under which all unlisted species (including *Lepisosteus platostomus*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered general piscicides, but no studies have been found of their effects on *Lepisosteus platostomus* (USACE 2012b).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

Lepomis microlophus Günther, 1859

Regulations (pertaining to the Great Lakes region)

The Redear Sunfish is listed as an approved species for aquaculture production in Michigan ([Aquaculture Development Act, Michigan Compiled Law § 286.875](#)). In Wisconsin, the Redear Sunfish is listed as a restricted species under the definition “nonnative fish in the aquaculture industry” ([WI Administrative Code § NR 40.02](#), Ontario takes a whitelist approach to bait under which all unlisted species (including *Lepomis microlophus*) are prohibited to use as bait (Ontario Fishing Guide 2014). [40.05](#)).

Control

Biological

There are no known biological control methods for this species.

Physical

Removal of catch limits for anglers and public information campaigns could aid control of this species.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered “general” piscicides, but no studies have been found of their effects on *Lepomis microlophus* (USACE 2012b).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate

CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

***Misgurnus anguillicaudatus* Cantor, 1842**

Regulations (pertaining to the Great Lakes region)

In Michigan, *Misgurnus anguillicaudatus* is a prohibited species under [Michigan Natural Resources and Environmental Protection Act \(NREPA\) 451 § 324.41301](#). No person shall knowingly possess a live prohibited organism in Michigan except for educational, research, or identification purposes as listed in [Michigan NREPA 451 § 324.41303](#). It is also unlawful to introduce a prohibited organism in Michigan under [Michigan NREPA 451 § 324.41305](#). In Michigan, a violation involving a prohibited species is a felony, and a knowing introduction violation with intent to harm is punishable with up to five years imprisonment and a \$2,000 to \$1,000,000 fine ([MI NREPA § 324.41309](#)). In Wisconsin, *M. anguillicaudatus* is restricted as a nonnative viable fish species in the aquarium trade ([WI Administrative Code § NR 40.02 and 40.05](#)). Ontario takes a whitelist approach to bait under which all unlisted species (including *Misgurnus anguillicaudatus*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered general piscicides, but no studies have been found of their effects on *M. anguillicaudatus* (USACE 2012b).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States,

not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

When planning control and management of this species, special attention should be given to the high physiological tolerances which place it in the profile of a successful invader. The Oriental Weatherfish can survive temperatures that range from 0-38° C, utilize atmospheric oxygen as a facultative air-breather to survive hypoxic conditions, and has been documented surviving desiccation for over 81 days with no food, likely perishing from desiccation before starvation (Koetsier and Urquhart 2012).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

Morone americana Gmelin, 1789

Regulations (pertaining to the Great Lakes region)

Minnesota lists White Perch as a prohibited invasive species ([MN Administrative Rules § 6216.0250](#)). In Ohio it is unlawful for any person to possess, import or sell live White Perch ([Ohio Administrative Code § 1501:31-19](#)). Indiana ([312 IN Administrative Code § 9-6-7](#)) designates White Perch as an exotic fish - an individual must not import, possess, propagate, buy, sell, barter, trade, transfer, loan, or release into public or private waters live fish or recently hatched or juvenile live fish or their viable eggs or genetic material. Ontario takes a whitelist approach to bait under which all unlisted species (including *Morone americana*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

For most waters, the only management recommendation for White Perch is unlimited harvest (Smith et al. 2002).

Biological

Bottom-up control (reduction in food supply) of White Perch usually results in stunting accompanied by an increase in population so that the population consists of many small fish (Smith et al. 2002).

Physical

There are no known physical control methods for this species.

Chemical

The IJC (2011) recommends rotenone for control of White Perch in rapid response scenarios.

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered “general” piscicides, but no studies have been found of their effects on *Morone americana* (USACE 2012b).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

***Neogobius melanostomus* Pallas, 1814**

Regulations (pertaining to the Great Lakes region)

In Ohio ([Ohio Administrative Code § 1501:31-19](#)) it is unlawful for any person to possess, import or sell live Round Gobies. Michigan ([MI NREPA 451, Section 324.41301](#)) and Minnesota ([MN Administrative Rules § 6216.0250](#)) list Round Goby as a prohibited species. Illinois ([IL Administrative Code § 17: 805.20](#)) lists Round Gobies as an injurious species. In Pennsylvania ([58 PA Code §71.6](#)), it is illegal to possess, import or introduce round gobies. New York ([6 NYCRR Part 10, Paragraph 10.1\(c\)\(3\)](#)) prohibits the use of round gobies as bait. Indiana ([312 IAC 9-6-7](#)) lists round goby as an exotic fish - an individual must not import, possess, propagate, buy, sell, barter, trade, transfer, loan, or release into public or private waters any exotic fish (including recently hatched or juvenile live fish or their viable eggs or genetic material). Ontario takes a whitelist approach to bait under which all unlisted species (including *Neogobius melanostomus*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

Although many other species consume round goby, no effective and species-specific biocontrol has been identified. Among other species, native burbot are being investigated for their potential to control goby populations (Madenjian et al 2011).

Physical

Electrical barriers may be successful at limiting the movement of round gobies. In tank studies, round gobies did not move through such a barrier (Savino et al 2001).

Chemical

The IJC (2011) recommends rotenone for control of round goby in rapid response scenarios.

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered “general” piscicides, but no studies have been found of their effects on *Neogobius semilunaris* (GLMRIS 2012).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the US, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for 5 min. is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

Other

Rollo et al (2007) reported round gobies will approach a speaker emitting conspecific male calls in the field, and female round gobies showed significant attractions to speakers emitting conspecific male calls in the laboratory. Therefore round goby phonotaxis could be used to lure gravid females to traps. As round gobies will spawn multiple times throughout late spring and summer, they should remain receptive to male calls and bioacoustic capture for the entire breeding season.

***Notropis buchanani* Meek, 1896**

Regulations

In Pennsylvania, the ghost shiner is an endangered species. The catching, taking, killing, possessing, importing to or exporting from the Commonwealth of Pennsylvania, selling or offering for sale or purchasing of any individual of an endangered species, alive or dead, or any part thereof, without a special permit, is prohibited. Ontario takes a whitelist approach to bait under which all unlisted species (including *Notropis buchanani*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered general piscicides, but no studies have been found of their effects on this species (GLMRIS 2012).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the US, not for use as euthanasia, and exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

Oncorhynchus gorbuscha Walbaum, 1792

Unlike other Pacific salmon, Pink Salmon were not deliberately stocked for Alewife biocontrol, nor are they deliberate stocked. Nonetheless, Pink Salmon descendants of an accidental release have become a valued part of the Great Lakes recreational fishery and they are managed alongside the other Pacific salmonids. Therefore, Pink Salmon management objectives are not geared towards the removal or eradication of the species like with most invaders.

Regulations (pertaining to the Great Lakes region)

Direct Regulations:

Great Lakes states and provinces have their own specific fishing regulations. Generally, the overall goals and objectives of Pacific salmon fishing regulations are the same throughout the region i.e., to maintain or enhance a healthy and sustainable salmonid fisheries. Pacific salmon fishing regulations include daily and season bag limits, size limits, permitted baitfish, manner of taking i.e., snagging or hook and line, and designated season dates (See New York DEC, Pennsylvania F&BC, Ohio DNR, Michigan DNR, Indiana DNR, Illinois DNR, Minnesota DNR, Wisconsin DNR, Ontario MNR, and Quebec MRNF websites for specific fishing regulations). Ontario takes a whitelist approach to bait under which all unlisted species (including *Oncorhynchus gorbuscha*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Indirect Regulations:

Typically, Pacific salmon regulations are not species specific, but rather regulate the salmonid fisheries as a whole. Indirect Pacific salmon regulations include mandated salmonid pathogen screening tests and baitfish regulations.

Mandatory salmonid pathogen screening tests are implemented in all Great Lakes states and provinces. The importation, exportation, and transportation of Pacific salmon is highly regulated to control the spread of infectious diseases and parasites such as VHS, Bacterial Kidney Disease (BKD), and whirling disease (See USGS nonindigenous diseases and parasites factsheets for state and provincial regulations).

State and provincial baitfish regulations have aided in preventing the spread of infectious disease. Specific and or stricter regulations are placed on baitfish species that are known carriers of salmonid pathogens.

Control

Biological

Like other Pacific salmon, Pink Salmon prey heavily upon two non-native species in the Great Lakes, the Alewife (*Alosa pseudoharengus*) and Rainbow Smelt (*Osmerus mordax*). Alewives remain a key food source and crucial to the survival of Pacific salmon. Over the past several decades, Pacific salmon populations have fluctuated with fluctuating Alewife populations. Managing one species significantly impacts the other. Pacific salmon and Alewives have significant environmental, socio-economic, and beneficial effects in the Great Lakes and therefore integrated management is essential. Rainbow Smelt are also a major component of Pacific salmon diet. Similar to Alewives, Pacific salmon and Rainbow Smelt management should be integrated. Rainbow Smelt have a high environmental impact and high beneficial effect in the Great Lakes. The presence or absence of this species significantly alters predator-prey relationships and competition between native species. Managers can also attempt to increase less harmful native prey species stocks while allowing harmful invasive prey species to decrease. Implementation of this bio-control has potential significant beneficial effects in the Great Lakes with few negative impacts (See USGS fact sheets on [Alewife](#) and [Rainbow Smelt](#)).

Of the 23 nonindigenous diseases and parasites in the Great Lakes, *Aeromonas salmonicida*, *Renibacterium salmoninarum*, *Myxobolus cerebralis*, and *Novirhabdovirus* sp. infections have been realized in Great Lakes Pacific salmon, while *Heterosporosis* sp. and *Piscirickettsia cf. salmonis* infections have been realized clinically or outside the Great Lakes. *Glugea hertwigi*, a microsporidian, is known to cause mortality in Rainbow Smelt. Therefore, Pacific salmon management must include the management of the above pathogens and parasites (See USGS factsheets on *Aeromonas salmonicida*, *Renibacterium salmoninarum*, *Myxobolus cerebralis*, *Novirhabdovirus* sp., *Heterosporosis* sp., *Piscirickettsia cf. salmonis*, and *Glugea hertwigi* for information on Great Lakes impacts and management).

Physical

Aquaculture facilities manage wild and cultured Pacific salmon stocks through wild stock assessments and other methods. Managers are then able to make informed decisions on stocking strategies. Research, pathogen screening, and pathogen treatment, etc. is conducted in aquaculture facilities (See New York DEC, Pennsylvania F&BC, Ohio DNR, Michigan DNR,

Indiana DNR, Illinois DNR, Minnesota DNR, Wisconsin DNR, Ontario MNR, and Quebec MRNF websites for information on salmonid aquaculture and state hatcheries).

Chemical

Chemical controls for Pacific salmon are not intended to eradicate or kill the species but rather to protect it against infectious disease. Typically, depending on the target species, chemicals controls are only effective in aquaculture or similar systems. Examples of chemicals used and include Furogen®, chlorination, and disinfectants.

Oncorhynchus kisutch Walbaum, 1792

Pacific salmon were first introduced to the Great Lakes in the 1960s to manage Alewife populations. Soon after, the multi-million dollar Great Lakes Pacific salmon sportfishery was established and is now one of the largest economic sectors in the region. Therefore, Pacific salmon management objectives are not geared towards the removal or eradication of the species like with most invaders, but rather to maintain or enhance the health and stability of the fisheries. Managers and citizens understand that with over 180 nonindigenous species, the Great Lakes are not the same ecosystem they once were. Management efforts still focus on the prevention and eradication of harmful invaders, but also realize that non-native Pacific salmon fisheries are one of the driving economic forces in the Great Lakes and managers need to account for this. Pacific salmon management is extremely diverse, integrated, and cascading and is therefore these are the most heavily regulated species (direct and indirectly) in the Great Lakes.

Regulations (pertaining to the Great Lakes region)

Direct Regulations:

Great Lakes states and provinces have their own specific fishing regulations. Generally, the overall goals and objectives of Pacific salmon fishing regulations are the same throughout the region i.e., to maintain or enhance a healthy and sustainable salmonid fisheries. Pacific salmon fishing regulations include daily and season bag limits, size limits, permitted baitfish, manner of taking i.e., snagging or hook and line, and designated season dates (See New York DEC, Pennsylvania F&BC, Ohio DNR, Michigan DNR, Indiana DNR, Illinois DNR, Minnesota DNR, Wisconsin DNR, Ontario MNR, and Quebec MRNF websites for specific fishing regulations). Ontario takes a whitelist approach to bait under which all unlisted species (including *Oncorhynchus kisutch*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Indirect Regulations:

Typically, Pacific salmon regulations are not species specific, but rather regulate the salmonid fisheries as a whole. Indirect Pacific salmon regulations include mandated salmonid pathogen screening tests and baitfish regulations.

Mandatory salmonid pathogen screening tests are implemented in all Great Lakes states and provinces. The importation, exportation, and transportation of Pacific salmon is highly regulated to control the spread of infectious diseases and parasites such as VHS, Bacterial Kidney Disease (BKD), and whirling disease (See USGS nonindigenous diseases and parasites factsheets for state and provincial regulations).

State and provincial baitfish regulations have aided in preventing the spread of infectious disease. Specific and or stricter regulations are placed on baitfish species that are known carriers of salmonid pathogens.

Control

Biological

Pacific salmon prey heavily upon two non-native species in the Great Lakes, the Alewife (*Alosa pseudoharengus*) and Rainbow Smelt (*Osmerus mordax*). Alewives remain a key food source and crucial to the survival of Pacific salmon. Over the past several decades, Pacific salmon populations have fluctuated with fluctuating Alewife populations. Managing one species significantly impacts the other. Pacific salmon and Alewives have significant environmental, socio-economic, and beneficial effects in the Great Lakes and therefore integrated management is essential. Rainbow Smelt are also a major component of Pacific salmon diet. Similar to Alewives, Pacific salmon and Rainbow Smelt management should be integrated. Rainbow Smelt have a high environmental impact and high beneficial effect in the Great Lakes. The presence or absence of this species significantly alters predator-prey relationships and competition between native species. Managers can also attempt to increase less harmful native prey species stocks while allowing harmful invasive prey species to decrease. Implementation of this bio-control has potential significant beneficial effects in the Great Lakes with few negative impacts (See USGS fact sheets on [Alewife](#) and [Rainbow Smelt](#)).

Of the 23 nonindigenous diseases and parasites in the Great Lakes, *Aeromonas salmonicida*, *Renibacterium salmoninarum*, *Myxobolus cerebralis*, and *Novirhabdovirus* sp. infections have been realized in Great Lakes Pacific salmon, while *Heterosporosis* sp. and *Piscirickettsia cf. salmonis* infections have been realized clinically or outside the Great Lakes. *Glugea hertwigi*, a microsporidian, is known to cause mortality in Rainbow Smelt. Therefore, Pacific salmon management must include the management of the above pathogens and parasites (See USGS factsheets on *Aeromonas salmonicida*, *Renibacterium salmoninarum*, *Myxobolus cerebralis*, *Novirhabdovirus* sp., *Heterosporosis* sp., *Piscirickettsia cf. salmonis*, and *Glugea hertwigi* for information on Great Lakes impacts and management).

Physical

Aquaculture facilities manage wild and cultured Pacific salmon stocks through wild stock assessments and other methods. Managers are then able to make informed decisions on stocking strategies. Research, pathogen screening, and pathogen treatment, etc. is conducted in aquaculture facilities (See New York DEC, Pennsylvania F&BC, Ohio DNR, Michigan DNR, Indiana DNR, Illinois DNR, Minnesota DNR, Wisconsin DNR, Ontario MNR, and Quebec MRNF websites for information on salmonid aquaculture and state hatcheries).

Chemical

Chemical controls for Pacific salmon are not intended to eradicate or kill the species but rather to protect it against infectious disease. Typically, depending on the target species, chemicals controls are only effective in aquaculture or similar systems. Examples of chemicals used and include Furogen®, chlorination, and disinfectants.

***Oncorhynchus mykiss* Walbaum, 1792**

Regulations (pertaining to the Great Lakes region)

Federal law in Canada regulates Rainbow Trout as a game fish ([CRC § c.1120](#)). In Quebec, it is illegal to stock Rainbow Trout in certain bodies of water as listed by [Quebec RRQ § c C-61.1, r 7](#). The sale of dead Rainbow Trout is also prohibited in Quebec ([Quebec RRQ § c C-61.1, r 7](#)). In Ontario, Rainbow Trout is regulated as an eligible species for aquaculture ([Ontario Regulation § 664/98](#)). Ontario takes a whitelist approach to bait under which all unlisted species (including *Oncorhynchus mykiss*) are prohibited to use as bait (Ontario Fishing Guide 2014).

In New York, trout, including Rainbow Trout, shall not be bought and sold, excepting cases in which a hatchery permit is issued, as described under [New York Environmental Conservation Law § 11-1909](#). In Ohio, it is unlawful to take or possess Rainbow Trout less than 12 inches in length while on Lake Erie or its tributaries, including all streams in the entire drainage basin, excepting Cold Creek upstream of state route two located in Erie county, and Beaver creek in Seneca County. It is also unlawful to take or possess Rainbow Trout less than twelve inches in length while on the Mad River or its tributaries ([Ohio Administrative Code § 1501:31-13-09](#)). In Michigan, Rainbow Trout is an approved species for aquaculture production ([MI Compiled Law § 286.875](#)). In Wisconsin, Rainbow Trout is restricted as a nonnative fish species in the aquaculture industry, and therefore cannot be transported, possessed, transferred, or introduced without a permit ([WI Admin Code § NR 40.05](#)).

Control

Biological

There are no known biological control methods for this species.

Physical

In streams and rivers, barriers can be constructed and natural barriers augmented to prevent upstream migration of trout and aid management and eradication efforts. In a report on a successful trout removal program involving a combination of piscicides (rotenone) for eradication and barriers for prevention of re-invasion, Lintermans and Raadik (2003) noted three key aspects of successful barriers: a 1.5 m or greater vertical drop; direction of water flow towards the middle in higher flows with no slower overland flow passing down the banks; and no deep pool below the barrier from which trout could jump. Rainbow Trout often rely on spawning streams and small tributaries for reproduction, and removal of access to such streams could reduce or potentially eliminate populations in downstream bodies of water (Champion et al. 2002). The United States National Park Service uses physical removal through electrofishing to manage Rainbow Trout and Brown Trout populations that threaten native Brook Trout in Shenandoah National Park, Virginia (NPS 2011).

Chemical

Antimycin A (available as Fintrol®) is a registered piscicide in the United States that is documented as highly effective against scaled fishes, including Rainbow Trout (Finlayson et al. 2002). Elimination of trout is achievable in a contact time of two hours at 5 µg/L (5 ppb), or in one hour at 10 ppb (Gilderhus 1972, Finlayson et al. 2002). Antimycin is most effective in small streams, shallow ponds, and alpine lakes where there is ample mixing and an adequate contact time can be achieved (Finlayson et al. 2002, Gilderhus 1972). Antimycin does not seem to repel fish like rotenone (another registered piscicide, available as Noxfish®) does, and rapidly breaks

down by hydrolysis in natural waters (Finlayson et al. 2002). Disadvantages of antimycin include increasing ineffectiveness in waters with higher pH (>8), streams with significant gradients (80-150 m elevation drop), and large lakes where good mixing and contact time cannot be established (Finlayson et al. 2002). Rotenone is also effective against Rainbow Trout, but at much higher concentrations, with 50 µg/L required to eliminate trout in a 2 hour contact time (Gilderhus 1972, Finlayson et al. 2002). Antimycin may be preferred because of the lower dose required. Antimycin and rotenone are non-selective, and toxicity to other fishes and aquatic invertebrates will vary.

Lintermans and Raadik (2003) provide a detailed account of Rainbow Trout elimination programs using rotenone in order to protect fishes of the family Galaxiidae, conducted in two separate areas of Australia. In 1992, Rainbow Trout were removed from 2.4 km of Lees Creek, Australian Capital Territory using a 5% rotenone emulsion at concentrations of approximately 0.05 parts per million (ppm) (Lintermans and Raadik 2003). The creek was treated in 500 m sections, with 300-350 mL of rotenone added over a 15-minute period to each section, and with mesh stop nets placed after each section to prevent downstream reinvasion of trout (Lintermans and Raadik 2003). An oxidant (350-500 g potassium permanganate) was added to the stream when rotenone reached the downstream limit of treatment sections to remove the toxicant (Lintermans and Raadik 2003). To prevent trout reinvasion of the treated area, a downstream weir was augmented with a heavy steel grill to present a 1.75 m vertical barrier (Lintermans and Raadik 2003). Complete eradication was accomplished at the Lees Creek site, and despite heavy impacts on aquatic macroinvertebrates, benthic macroinvertebrates remained in significant numbers (Lintermans and Raadik 2003). In 1995, a total of 20 km of stream length (seven different streams) was treated in Victoria, with a total of 60 L rotenone used, neutralized with 1100 kg of potassium permanganate, and with rotenone volumes ranging from 0.3 to 0.5 L per 100 m of stream (Lintermans and Raadik 2003). Areas in which trout and galaxiids overlapped were not treated with piscicides; they were instead intensively electrofished to more selectively remove Rainbow Trout (Lintermans and Raadik 2003).

The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood before pursuing chemical treatment options. Boogaard et al. (2003) found that the lampricides 3-trifluoromethyl-4-nitrophenol (TFM) and 2',5-dichloro-4'-nitrosalicylanilide (niclosamide) demonstrate additive toxicity when combined, with Rainbow Trout demonstrating 12h LC50s of 8.40-10.6 mg/L in response to treatments of TFM and 5.00-5.05 mg/L in response to treatments of a TFM/1% niclosamide combination in lab tests (Boogaard et al. 2003). In another study on cumulative toxicity, combinations of Bayer 73 (niclosamide) and TFM with contaminants common in the Great Lakes (pesticides, heavy metals, industrial organics, phosphorus, and sediments) were found to be mostly additive in toxicity to Rainbow Trout, and one combination of TFM, Delnav, and malathion was synergistic, with toxicity magnified 7.9 times (Marking and Bills 1985). This highlights the need for managers to conduct on-site toxicity testing and to give serious consideration to determining the total toxic burden to which organisms may be exposed when using chemical treatments (Marking and Bills 1985).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate

CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). Laboratory trials demonstrated a combination of pH 6.5 and 642 mg/L NaHCO₃ was the most effective treatment for Rainbow Trout (Clearwater et al. 2008). CO₂ is approved only for use as an anaesthetic for cold, cool, and warm water fishes in the United States. It is not approved for use as euthanasia (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

Oncorhynchus nerka (Walbaum in Artedi, 1792)

Regulations (pertaining to the Great Lakes region)

In Canada, Sockeye Salmon is a game fish as designated by the [National Parks of Canada Fishing Regulations § CRC, c. 1120](#). Sockeye Salmon is also a game fish in the provinces of Ontario and Quebec ([Ontario Fish Regulations § SOR/2007-237](#); [Quebec Fish Regulations § SOR/90-214](#)). Ontario takes a whitelist approach to bait under which all unlisted species (including *Oncorhynchus nerka*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods for this species.

Physical

Sockeye Salmon are a homing species, returning to their native stream for reproduction. Barriers can be constructed and/or natural barriers augmented to prevent upstream migration and aid management and eradication efforts, though little research exists on effective barriers for *Oncorhynchus nerka*. Lintermans and Raadik (2003) noted three key aspects of successful barriers in relation to a Rainbow Trout control program: a 1.5 m or greater vertical drop; direction of water flow towards the middle in higher flows with no slower overland flow passing down the banks; and no deep pool below the barrier from which fish could jump.

The United States Army Corps of Engineers Great Lakes and Mississippi River Interbasin Study (USACE GLMRIS) notes the potential effectiveness of sensory deterrent systems in providing barriers to fish migration or eliciting fish movements (USACE 2012b). Specifically, the success of underwater strobe lights as studied by Maiolie et al. (2001) is cited. Testing conducted on wild, free-ranging *O. nerka* in their natural pelagic habitat in two large Idaho lakes revealed that fish moved an average of 30-136 m away from lights in waters with secchi transparency of 2.8 to 17.5 m, with an 80% reduction in fish density within 30 m of the strobe lights (Maiolie et al. 2001). Many large scale strobe systems consist of four individual lights that flash at a rate of 450 flashes/minute, with an approximate intensity of 2634 lumens/flash (USACE 2012b). Maiolie et al. (2001) tested flash rates of 300, 360, and 450 flashes/minute.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered general piscicides, but no studies have been found of their effects on *O. nerka* (USACE 2012b).

Exposure to niclosamide, registered in the United States as a granular lampricide, wettable powder, technical grade product, and an emulsifiable concentrate, is known to be toxic to all fish species at 0.5 mg/L after a 48-h exposure (Clearwater et al. 2008). There are no available studies of its specific effects on Sockeye Salmon, though it has been used for control of aquatic snails, Zebra Mussels, and oligochaetes, and is also toxic to many crayfish, frogs, clams, algae, and other amphibian and fish species (Clearwater et al. 2008).

In a study on removal of toxic chemicals from water using activated carbon, Dawson et al. (1976) found that granular activated carbon is saturated by rotenone at 0.1 mg of Noxfish per gram carbon, and cited other studies documenting 0.94-1.32 mg of Noxfish adsorbed per gram of carbon. Antimycin was efficiently absorbed and did not saturate carbon because of the low doses used (Dawson et al. 1976).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). Salmonids are considered to be among the most sensitive fishes to low dissolved oxygen levels, with a DO concentration of 1-3 mg/L sufficient to cause mortality or loss of equilibrium (Clearwater et al. 2008). However, CO₂ is approved only for use as an anaesthetic for cold, cool, and warm water fishes the United States, for use as euthanasia (Clearwater et al. 2008). Exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

Low pH is known to affect fish behavior. Ikuta et al. (2003) documented the effects of low pH on Sockeye Salmon, noting that salmon would not swim upstream into areas of pH lower than 6.0. Acute exposure to low pH levels can directly kill fish by discharge of sodium and chloride ions from body fluid, and sub-lethal levels can affect reproduction (Ikuta et al. 2003). In the case of Sockeye Salmon, weak acidic conditions of ≤pH 6 were enough to depress the prespawning behavior of swimming upstream (Ikuta et al. 2003).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target organisms, such as macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna, and their potential harmful effects should therefore be evaluated thoroughly.

Oncorhynchus tshawytscha Walbaum in Artdi, 1792

Pacific salmon were first introduced to the Great Lakes in the 1960s to manage Alewife populations. Soon after, the multi-million dollar Great Lakes Pacific salmon sportfishery was

established and is now one of the largest economic sectors in the region. Therefore, Pacific salmon management objectives are not geared towards the removal or eradication of the species like with most invaders, but rather to maintain or enhance the health and stability of the fisheries. Managers and citizens understand that with over 180 nonindigenous species, the Great Lakes are not the same ecosystem they once were. Management efforts still focus on the prevention and eradication of harmful invaders, but also realize that non-native Pacific salmon fisheries are one of the driving economic forces in the Great Lakes and managers need to account for this. Pacific salmon management is extremely diverse, integrated, and cascading and is therefore these are the most heavily regulated species (direct and indirectly) in the Great Lakes.

Regulations (pertaining to the Great Lakes region)

Direct Regulations:

Great Lakes states and provinces have their own specific fishing regulations. Generally, the overall goals and objectives of Pacific salmon fishing regulations are the same throughout the region i.e., to maintain or enhance a healthy and sustainable salmonid fisheries. Pacific salmon fishing regulations include daily and season bag limits, size limits, permitted baitfish, manner of taking i.e., snagging or hook and line, and designated season dates (See New York DEC, Pennsylvania F&BC, Ohio DNR, Michigan DNR, Indiana DNR, Illinois DNR, Minnesota DNR, Wisconsin DNR, Ontario MNR, and Quebec MRNF websites for specific fishing regulations). Ontario takes a whitelist approach to bait under which all unlisted species (including *Oncorhynchus tshawytscha*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Indirect Regulations:

Typically, Pacific salmon regulations are not species specific, but rather regulate the salmonid fisheries as a whole. Indirect Pacific salmon regulations include mandated salmonid pathogen screening tests and baitfish regulations.

Mandatory salmonid pathogen screening tests are implemented in all Great Lakes states and provinces. The importation, exportation, and transportation of Pacific salmon is highly regulated to control the spread of infectious diseases and parasites such as VHS, bacterial kidney disease (BKD), and whirling disease (See USGS nonindigenous diseases and parasites factsheets for state and provincial regulations).

State and provincial baitfish regulations have aided in preventing the spread of infectious disease. Specific and or stricter regulations are placed on baitfish species that are known carriers of salmonid pathogens.

Control

Biological

Pacific salmon prey heavily upon two non-native species in the Great Lakes, the Alewife (*Alosa pseudoharengus*) and Rainbow Smelt (*Osmerus mordax*). Alewives remain a key food source and crucial to the survival of Pacific salmon. Over the past several decades, Pacific salmon populations have fluctuated with fluctuating Alewife populations. Managing one species significantly impacts the other. Pacific salmon and Alewives have significant environmental, socio-economic, and beneficial effects in the Great Lakes and therefore integrated management is essential. Rainbow Smelt are also a major component of Pacific salmon diet. Similar to Alewives, Pacific salmon and Rainbow Smelt management should be integrated. Rainbow Smelt have a high environmental impact and high beneficial effect in the Great Lakes. The presence or

absence of this species significantly alters predator-prey relationships and competition between native species. Managers can also attempt to increase less harmful native prey species stocks while allowing harmful invasive prey species to decrease. Implementation of this bio-control has potential significant beneficial effects in the Great Lakes with few negative impacts (See USGS fact sheets on [Alewife](#) and [Rainbow Smelt](#)).

Of the 23 nonindigenous diseases and parasites in the Great Lakes, *Aeromonas salmonicida*, *Renibacterium salmoninarum*, *Myxobolus cerebralis*, and *Novirhabdovirus* sp. infections have been realized in Great Lakes Pacific salmon, while *Heterosporosis* sp. and *Piscirickettsia* cf. *salmonis* infections have been realized clinically or outside the Great Lakes. *Glugea hertwigi*, a microsporidian, is known to cause mortality in Rainbow Smelt. Therefore, Pacific salmon management must include the management of the above pathogens and parasites (See USGS factsheets on *Aeromonas salmonicida*, *Renibacterium salmoninarum*, *Myxobolus cerebralis*, *Novirhabdovirus* sp., *Heterosporosis* sp., *Piscirickettsia* cf. *salmonis*, and *Glugea hertwigi* for information on Great Lakes impacts and management).

Physical

Aquaculture facilities manage wild and cultured Pacific salmon stocks through wild stock assessments and other methods. Managers are then able to make informed decisions on stocking strategies. Research, pathogen screening, and pathogen treatment, etc. is conducted in aquaculture facilities (See New York DEC, Pennsylvania F&BC, Ohio DNR, Michigan DNR, Indiana DNR, Illinois DNR, Minnesota DNR, Wisconsin DNR, Ontario MNR, and Quebec MRNF websites for information on salmonid aquaculture and state hatcheries).

Chemical

Chemical controls for Pacific salmon are not intended to eradicate or kill the species but rather to protect it against infectious disease. Typically, depending on the target species, chemical controls are only effective in aquaculture or similar systems. Examples of chemicals used and include Furogen®, chlorination, and disinfectants.

***Osmerus mordax* Mitchill, 1814**

Regulations (pertaining to the Great Lakes region)

Use of Rainbow Smelt is regulated in the Canadian province of Quebec under [Quebec Fish Regulations § SOR/90-214](#). A population of Rainbow Smelt in an area south of the St. Lawrence estuary is designated a vulnerable wildlife species in Quebec under [Quebec Statutes and Regulations RRQ § c E-12.01, r 2](#). The sale of dead Rainbow Smelt is prohibited in Quebec by [Quebec Statutes and Regulations RRQ § c C-61.1, r 7](#). In Ontario, Rainbow Smelt use as bait and non-angling fishing methods are regulated by [Canada Federal Statutes and Regulations SOR § 2007-237](#). Ontario takes a whitelist approach to bait under which all unlisted species (including *Osmerus mordax*) are prohibited to use as bait (Ontario Fishing Guide 2014).

In the state of New York, it is unlawful to use Rainbow Smelt as bait except as provided in [New York Codes, Rules, and Regulations 6 § 19.2](#). Furthermore, it is unlawful to take Rainbow Smelt for sale as bait or to sell as bait, except as otherwise provided as without a pursuant license as defined in [New York Environmental Conservation Law § 11-1315](#). In Pennsylvania, the use of commercial trap nets under license to capture Rainbow Smelt is regulated by [Pennsylvania Administrative Code § 69.33](#). In Ohio, Rainbow Smelt is defined as a commercial fish and an

unrestricted species under [Ohio Administrative Code 1501 § 31-1-02](#). Commercial fish are permitted to be taken, possessed, bought, or sold unless otherwise restricted in Ohio code. In Indiana, the Rainbow Smelt sport fishing season on Lake Michigan is defined as March 1-May 30, with capture allowed only by the use of dip nets, seines, or nets with limitations provided in [312 Indiana Administrative Code § 9-7-2](#). There is otherwise no bag limit, possession limit, or size limit, as defined under [312 Indiana Administrative Code § 9-7-14](#). In Illinois, the sport-fishing season of Rainbow Smelt is defined as March 1-April 30 under [Illinois Administrative Code 17-1 § 810.10](#). In Wisconsin, Rainbow Smelt is defined as an established non-native fish species in [Wisconsin Administrative Code § NR 40.02](#), and is restricted per the above definition by [Wisconsin Admin Code § NR 40.05](#). In Minnesota, Rainbow Smelt is a regulated invasive species under [Minnesota Administrative Rules § 6216.0260](#).

Control

Biological

Several species of non-native salmonids have been introduced to the Great Lakes, beginning in the 1960s, to control invasive Rainbow Smelt (USACE 2012b). Rainbow Smelt is heavily consumed by Atlantic Salmon (*Salmo salar*), Lake Trout (*Salvelinus namaycush*), Brook Trout (*S. fontinalis*), Coho Salmon (*Oncorhynchus kisutch*), Chinook Salmon (*O. tshawytscha*), Rainbow Trout (*O. mykiss*), Brown Trout (*Salmo trutta*), Splake (Brook Trout x Lake Trout), Burbot (*Lota lota*), Walleye (*Sander vitreus*), Northern Pike (*Esox lucius*), and many other freshwater piscivores (Stewart et al. 1981, Brandt and Madon 1986, Crossman 1991, He and LaBar 1994, Kirn and LaBar 1996, USACE 2012b). However, the significance of piscivore predation on Rainbow Smelt has only been studied for a few species. Observed Atlantic Salmon predation on smaller Rainbow Smelt, as well as bioenergetics modeling suggesting that by age four, cumulative piscivory by Atlantic Salmon was nearly 10-fold greater than that of Lake Trout of the same age, implies its greater usefulness for management of Rainbow Smelt (Kirn and LaBar 1996). While Lake Trout consume large amounts of Rainbow Smelt, almost exclusively so in some studies, the species is believed to provide little potential for responsive management manipulation outside of stabilizing fluctuating prey populations, due to the long cycle of its predatory effect (peaking 3-5 years after stocking, lasting 7-8 years) (Stewart et al. 1981, He and LaBar 1994, Kirn and LaBar 1996). Chinook Salmon have been successfully used to eradicate Rainbow Smelt from small lakes in New Hampshire in 1936 (Stewart et al. 1981). Because the trade-off between fish species as agents of biological control is not directly correlated with consumption, management decisions involving shifts between species should not take consumption solely into account (Stewart et al. 1981).

Physical

The United States Army Corps of Engineers Great Lakes and Mississippi River Interbasin Study notes the potential effectiveness of sensory deterrent systems in providing barriers to fish migration or eliciting fish movements (USACE 2012b). *In situ* testing of two models of strobe lights as a deterrent preventing entrainment of Rainbow Smelt through Oahe Dam, Lake Oahe, South Dakota demonstrated successful avoidance of 15-21 m horizontally and six meters vertically by Rainbow Smelt (Hamel et al. 2008). Many large scale strobe systems consist of four individual lights that flash at a rate of 450 flashes/minute, with an approximate intensity of 2634 lumens/flash (USACE 2012b). Hamel et al. (2008) tested the AGL FH-901 flashhead, which consists of four horizontal lights positioned at 90 degree angles, flashing 450 times/min at 2,634 lumens/flash, and the newer AGL FH-920 flashhead, which consists of an omnidirectional

vertical light tube, covering a full 360 degrees with 360 flashes/min at 6,585 lumens. When using physical deterrents as barriers, combining methods can increase effectiveness, as was the case for Patrick et al. (1985), who found that Rainbow Smelt and other pelagic fishes were successfully deterred by a barrier combining air bubbles and strobe lights.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered general piscicides (USACE 2012b). Marking et al. (1983) found that the three most effective registered chemicals for potential use in control of Rainbow Smelt eggs and larvae are rotenone, potassium permanganate, and chlorine, respectively. In exposures of 6-24 hours, all chemicals were effective at concentrations from 5 to >10 mg/L (Marking et al. 1983). Rotenone demonstrated a 96h LC50 of 0.015 mg/L for Rainbow Smelt eggs and 0.001 mg/L for larvae (derived calculating only the activity of rotenone in 5% Noxfish solution) (Marking et al. 1983). Potassium permanganate demonstrated 96h LC50s of 0.074 mg/L and 0.075 mg/L for eggs and larvae, respectively. Chlorine demonstrated 96h LC50s of 0.14 mg/L for eggs and 0.31 mg/L for larvae (Marking et al. 1983). Temperature, pH, and hardness of water all affected toxicity of rotenone and potassium permanganate, with higher temperatures, softer water, and higher pH increasing toxicity (Marking et al. 1983). It should be noted that tests were carried out in a laboratory, but natural waters usually contain oxidizable material, which produces a demand on chlorine and reduces it to a less active form (Marking et al. 1983).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia (Clearwater et al. 2008). Exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

Petromyzon marinus (Linnaeus, 1758)

Regulations (pertaining to the Great Lakes region)

In Minnesota, sea lamprey is a prohibited species and therefore it is unlawful to possess, import, purchase, transport, or introduce this species except under a permit for disposal, control, research, or education (MDNR 2012). In Ohio it is illegal to possess, import or sell live lamprey ([OAC Chapter 1501:31-19](#)). New York ([NY ECL 11-1315, 6a](#)) prohibits the use of lamprey

larvae as bait. Ontario takes a whitelist approach to bait under which all unlisted species (including *Petromyzon marinus*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Since the bi-national Sea Lamprey Control program (managed by the Great Lakes Fishery Commission) was started in the 1950s, populations have been reduced by 90%, and fish survival and spawning have increased (Page and Laird 1993, Smith 1985). It is impossible to completely eradicate sea lamprey from the Great Lakes; however, continuous control efforts can minimize their impacts on the ecosystem and fisheries (FOC 2009).

Biological

An effective bio-control of sea lamprey is the implementation of the sterile-male-release program. Male sea lamprey are captured during spawning runs, sterilized using bisazir, and released to compete with fertile males for mating; thus reducing egg fertilization. Released males are sterilized past their parasitic phase and do not return to the lake. (FOC 2009, GLFHC 2000a). A potential alternative to bisazir is a lamprey GnRH antagonist. However, further research of this alternative sterilizing agent is necessary (Bergstadt and Twohey 2007).

Physical

Barriers and traps have been effective controls of sea lamprey since the 1950s. Barrier options include mechanical weirs, electrical barriers, low-head barriers, adjustable crest barriers, and velocity barriers (GLFHC 2000, Scott and Crossman 1973, Smith and Tibbles 1980). Traps are often used in association with barriers to capture sea lamprey while allowing desired species to continue upstream (FOC 2009, GLFHC 2000b). Barriers have reduced the need for lampricide applications (GLFC 2012). Once captured, sea lamprey are killed, used for research, or used in sterile-male -release programs.

Chemical

Beginning in the late 1950s, sea lampreys began to be successfully controlled by use of the lampricide 3-trifluoromethyl-4-nitrophenol (TFM), a chemical agent that kills larval lampreys in their stream habitats (Smith and Tibbles 1980). The lampricide has reduced the population by over 90% of the 1961 peak (Scott and Crossman 1973). However, continued use of TFM is required to keep populations under control (Becker 1983, Scott and Crossman 1973). TFM is sometimes harmful to other fish (e.g., walleye) as well as to the larvae of nonparasitic native lamprey species (Becker 1983). Bayluscide (niclosamide) treatments in deltas are also a widely used and an effective control of sea lamprey larvae (NYSDEC 2012).

Other

To increase the efficacy of lampricide treatments, streams and rivers are frequently assessed for larvae density to help determine the application sites (FOC 2009, GLFC 2012).

Note: Check state/provincial and local regulations for the most up-to-date information regarding permits for control methods. Follow all label instructions.

***Phenacobius mirabilis* (Girard, 1856)**

Regulations

There are no known regulations for this species. Ontario takes a whitelist approach to bait under which all unlisted species (including *Phenacobius mirabilis*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered general piscicides (GLMRIS 2012). There are no available studies of their effects on suckermouth minnow at the time of this writing.

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the US, not for use as euthanasia (Clearwater et al. 2008). Exposure to NaHCO₃ concentration of 142-642 mg/L for 5 min. is sufficient to anaesthetize most fish (Clearwater et al 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

***Proterorhinus semilunaris* (Heckel, 1837)**

Regulations

In Canada, tubenose goby is listed as an invasive species under Canadian Federal Statutes and Regulations, and is thus prohibited from being possessed, released, or used as bait without a license. In Quebec, aquarium fish keeping, production, keeping in captivity, breeding, stocking, transport, sale, or purchase of live tubenose goby is prohibited by .

In the commonwealth of Pennsylvania, it is unlawful to possess, sell, introduce, or import tubenose goby under . Sale, purchase, or barter of injurious, nonnative species, including tubenose goby, is prohibited by , and transportation in or through the commonwealth is

prohibited by . In the state of Ohio, it is unlawful for a person to possess, import, or sell live tubenose goby under . In the state of Michigan, tubenose goby is a prohibited species under . Tubenose goby is regulated as an exotic fish in the state of Indiana under , meaning an individual must not import, possess, propagate, buy, sell, barter, trade, transfer, loan, or release into public or private waters any tubenose goby, including recently hatched or juvenile live fish or their viable eggs or genetic material. In the state of Illinois, tubenose goby is an injurious species under . It is unlawful to possess, propagate, buy, sell, barter, or offer to buy, sell, barter, transport, trade, transfer, or loan tubenose goby to any person or institution without a permit in Illinois. Tubenose goby is a restricted invasive species in Wisconsin under . In the state of Minnesota, tubenose goby is a prohibited invasive species as defined in . Ontario takes a whitelist approach to bait under which all unlisted species (including *Proterorhinus semilunaris*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods for this species.

Physical

The USACE Great Lakes and Mississippi River Interbasin Study notes the potential effectiveness of acoustic fish deterrents in controlling or deterring *Proterorhinus semilunaris* populations (GLMRIS 2012). Acoustic deterrents include continuous wave and pulsed wave technology, which use sound/pressure waves to influence the behavior of aquatic organisms. Similarly, sensory deterrent systems such as acoustic air bubble curtains, electric barriers, and underwater strobe lights may prove useful in controlling populations in waterways and small bodies of water, but there are no studies of their effects on *P. semilunaris* at the present time (GLMRIS 2012). When using physical deterrents as barriers, combining methods can increase effectiveness, as was the case for Patrick et al. (1985), who found that pelagic estuarine and freshwater fishes were successfully deterred by a barrier combining air bubbles and strobe lights.

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered general piscicides, but there are no studies of the effects of chemical treatment on *P. semilunaris* at the present time (GLMRIS 2012).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the US, not for use as euthanasia (Clearwater et al. 2008). Exposure to NaHCO₃ concentration of 142-642 mg/L for 5 min. is sufficient to anaesthetize most fish (Clearwater et al 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations

of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

***Salmo trutta* Linnaeus, 1758**

Regulations

In Wisconsin, brown trout is restricted as a nonnative fish species in the aquaculture industry. Ontario takes a whitelist approach to bait under which all unlisted species (including *Salmo trutta*) are prohibited to use as bait (Ontario Fishing Guide 2014).

Control

Biological

There are no known biological control methods for this species.

Physical

Barriers can be constructed and/or natural barriers augmented to prevent upstream migration and aid management and eradication efforts, though little research exists on effective barriers for *Salmo trutta*. Lintermans and Raadik (2003) noted 3 key aspects of successful barriers in relation to a Rainbow Trout control program: a 1.5 m or greater vertical drop; direction of water flow towards the middle in higher flows with no slower overland flow passing down the banks; and no deep pool below the barrier from which fish could jump.

The United States Army Corps of Engineers Great Lakes and Mississippi River Interbasin Study (USACE GLMRIS) notes the potential effectiveness of sensory deterrent systems in providing barriers to fish migration or eliciting fish movements, including underwater strobe lights, acoustic air bubble barriers, continuous and pulsed wave acoustic deterrents, and electric barriers (USACE 2012b). Most large-scale strobe systems consist of four individual lights that flash at a rate of 450 flashes/minute, with an approximate intensity of 2634 lumens/flash (USACE 2012b).

The United States National Park Service uses physical removal through electrofishing to manage Rainbow Trout and Brown Trout populations that threaten native Brook Trout in Shenandoah National Park, Virginia (NPS 2011).

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered general piscicides (USACE 2012b).

In 1995, a total of 20 km of stream length (seven different streams) was treated in Victoria, Australia with a total of 60 L rotenone used, neutralized with 1100 kg of potassium permanganate, and with rotenone volumes ranging from 0.3 to 0.5 L per 100 m of stream, in order to eradicate Rainbow Trout (*Oncorhynchus mykiss*) and Brown Trout and protect native Galaxiids (Lintermans and Raadik 2003). Areas in which Trout and Galaxiids overlapped were not treated with Piscicides; they were instead intensively electrofished to more selectively remove Trout (Lintermans and Raadik 2003). Rotenone has also been used to eradicate Brown Trout in two streams of Kaiwharawhara catchment in Wellington, New Zealand using a concentration of 200 µg/L rotenone over initial treatment times of 4 and 5.5 hours for the smaller

and larger stream, respectively (Pham et al. 2013). Ling (2003) noted that Brown Trout has an LC50 of 5.5 µg/L at 17°C for 1h exposure to rotenone.

Boogaard et al. (2003) found that Brown Trout are among the least sensitive fishes to the lampricide TFM and a TFM/1% niclosamide mixture in laboratory exposures, with a 12 hour LC50 of 8.90 mg/L for TFM and 12 hour LC50s of 5.01 and 5.68 mg/L for TFM/1% niclosamide. Sensitivity was slightly decreased in field tests (Boogaard et al. 2003).

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). Salmonids are considered to be among the most sensitive fishes to low dissolved oxygen levels, with a DO concentration of 1-3 mg/L sufficient to cause mortality or loss of equilibrium (Clearwater et al. 2008). However, CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States, not for use as euthanasia (Clearwater et al. 2008). Exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

Low pH is known to affect fish behavior. Ikuta et al. (2003) documented the effects of low pH on *S. trutta*, noting that trout would not swim upstream into areas of pH lower than 5.5. Acute exposure to low pH levels can directly kill fish by discharge of sodium and chloride ions from body fluid, and sub-lethal levels can affect reproduction (Ikuta et al. 2003). In the case of Brown Trout, weak acidic conditions of <pH 6.4 were enough to depress prespawning digging behavior. Salmonids in general show significant avoidance of acidic environments (Ikuta et al. 2003).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Boogaard et al. (2003) found that the lampricides 3-trifluoromethyl-4-nitrophenol (TFM) and 2',5-dichloro-4'-nitrosalicylanilide (niclosamide) demonstrate additive toxicity when combined. In another study on cumulative toxicity, combinations of Bayer 73 (niclosamide) and TFM with contaminants common in the Great Lakes (pesticides, heavy metals, industrial organics, phosphorus, and sediments) were found to be mostly additive in toxicity to Rainbow Trout, and one combination of TFM, Delnav, and malathion was synergistic, with toxicity magnified 7.9 times (Marking and Bills 1985). This highlights the need for managers to conduct on-site toxicity testing and to give serious consideration to determining the total toxic burden to which organisms may be exposed when using chemical treatments (Marking and Bills 1985). Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

***Scardinius erythrophthalmus* Linnaeus, 1758**

Regulations (pertaining to the Great Lakes region)

In Quebec, aquarium fish-keeping, production, keeping in captivity, breeding, stocking, transport, sale or purchase of live Rudd is prohibited ([Quebec Statutes and Regulations RRW § c C-61.1, r7](#)). In Ontario, Rudd is listed as an invasive fish ([Ontario Fishery Regulations § SOR/2007-237](#)) and its use as bait is prohibited. In Pennsylvania, it is unlawful to possess, import, or introduce Rudd ([58 PA Code § 71.6](#)). In Ohio, it is unlawful to possess, import, or sell Rudd ([Ohio Administrative Code § 1501:31-19](#)). In Michigan, Rudd is a prohibited fish species ([MI NREPA 451 § 324.41301](#)). In Indiana, it is unlawful to import, possess, propagate, buy, sell, barter, trade, transfer, loan, or release into public or private waters any adult or recently hatched or juvenile Rudd or their genetic material ([312 IN Administrative § 9-6-7](#)). In Illinois, Rudd is an injurious species, meaning it shall not be possessed, propagated, bought, sold, bartered or offered to be bought, sold, bartered, transported, traded, transferred or loaned to any other person or institution unless a permit is first obtained from the Department of Natural Resources in accordance with [Illinois Administrative Code § 805.40](#), except persons engaged in interstate transport for lawful commercial purposes who do not buy, sell, barter, trade, transfer, loan or offer to do so in Illinois ([IL Administrative Code § 805](#)). In Wisconsin, Rudd is a prohibited species under [Wisconsin Administrative Code § NR 40.04](#), meaning that no person may transport, possess, transfer, or introduce Rudd, except as otherwise provided in paragraphs (b) to (h) of [Wisconsin Administrative Code § NR 40.04](#). In Minnesota, Rudd is a prohibited species, meaning it is unlawful (a misdemeanor) to possess, import, purchase, transport, or introduce except under a permit for disposal, control, research, or education ([MN Administrative Rules § 6216.0250](#)).

Control

Biological

There are no known biological control methods for this species.

Physical

Fine-mesh monofilament gill nets have been used to control Rudd in three shallow lakes in Waikato, New Zealand, but elimination was not achievable (Neilson et al. 2004). Small, potentially fecund fish in dense littoral vegetation proved challenging to net, presenting a problem for total eradication, but removal of larger Rudd likely affected breeding success and netting is seen as a highly cost effective control method with low environmental impact (Neilson et al. 2004).

Chemical

Of the four chemical piscicides registered for use in the United States, antimycin A and rotenone are considered general piscicides (USACE 2012b). Ling (2003) noted that rotenone has an LC50 of 24.5 µg/L for one hour exposure to Rudd at 20°C.

Increasing CO₂ concentrations, either by bubbling pressurized gas directly into water or by the addition of sodium bicarbonate (NaHCO₃) has been used to sedate fish with minimal residual toxicity, and is a potential method of harvesting fish for removal, though maintaining adequate CO₂ concentrations may be difficult in large/natural water bodies (Clearwater et al. 2008). CO₂ is approved only for use as an anesthetic for cold, cool, and warm water fishes the United States,

not for use as euthanasia (Clearwater et al. 2008). Exposure to NaHCO₃ concentration of 142-642 mg/L for five minutes is sufficient to anaesthetize most fish (Clearwater et al. 2008).

It should be noted that chemical treatment will often lead to non-target kills, and so all options for management of a species should be adequately studied before a decision is made to use piscicides or other chemicals. Potential effects on non-target plants and organisms, including macroinvertebrates and other fishes, should always be deliberately evaluated and analyzed. The effects of combinations of management chemicals and other toxicants, whether intentional or unintentional, should be understood prior to chemical treatment. Other non-selective alterations of water quality, such as reducing dissolved oxygen levels or altering pH, could also have a deleterious impact on native fishes, invertebrates, and other fauna or flora, and their potential harmful effects should therefore be evaluated thoroughly.

A.11 Insects

Acentria ephemerella Olivier, 1791

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

Acentria ephemerella is used for biological control of Eurasian Watermilfoil (*Myriophyllum spicatum*). Its population is best controlled by elimination of its host plants, which are predominantly Eurasian Watermilfoil but may to a lesser extent, include a variety of other native and nonindigenous plants (Cornell 2004).

Physical

Mechanical harvesting, herbicide applications, benthic barriers, and water drawdowns all remove either *A. ephemerella* individuals or their habitat (aquatic plants) from waterways (Cornell 2004).

Chemical

This species is susceptible to herbicide control of its host plants (Cornell 2004).

Tanysphyrus lemnae Paykull/Fabricius, 1792

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological, physical or chemical control methods for this species.

A.12 Mollusks

Corbicula fluminea O. F. Müller, 1774

Regulations (pertaining to the Great Lakes region)

Corbicula fluminea is a prohibited species in Wisconsin ([Wisconsin Administrative Code § NR 40.04](#)). In Indiana an individual may not import, possess or release Asiatic clams ([312 IN Administrative Code § 9-9-3 and § 14-22-17-3](#)).

Control

Eradication of Asian Clams from infested open waters is unlikely – emphasis is generally on preventing further spread.

Biological

There are no known biological control methods for this species.

Physical

Screens and traps are commonly employed to prevent *C. fluminea* colonization of water intakes (GISD 2013).

Diver assisted suction removal and bottom barriers are being researched as potential methods for physical control of *C. fluminea* populations in Lake Tahoe (UC Davis TERC, 2004). Benthic barriers have been demonstrated to be effective for short-term control of *C. fluminea*, but non-target mortality to other benthic invertebrates may be high (Wittmann et al. 2012).

Chemical

A wide array of chemical molluscicides are available, but are not species-specific and may harm native species to a greater extent than non-natives.

Molluscicides are typically classified as either oxidizing or non-oxidizing compounds. Oxidizing chemicals include chlorine, chlorine dioxide, chloramines, ozone, bromine, hydrogen peroxide, and potassium permanganate. Non-oxidizing chemicals (including organic film-forming antifouling compounds, gill membrane toxins, and nonorganics) can be classified into several distinct groups: quaternary and polyquaternary ammonium compounds; aromatic hydrocarbons; endothal as the mono (N,N-dimethylalkyl amine) salt; metals and their salts (e.g., copper sulfate formulations); and niclosamide (including some formulations of Bayluscide). Bayluscide was initially developed as a Sea Lamprey larvicide, but has molluscicidal activity. While some of these products are biodegradable, many require detoxification or deactivation to meet state and Federal discharge requirements (USACE 2012a).

Low concentration of chlorine or bromine will kill juvenile Asian Clams (GISD 2013).

C. fluminea is not tolerant of fluctuating environmental conditions (particularly temperature and oxygen) and is prone to massive die-offs (Menninger 2013), this suggests that short-term chemical manipulation may be useful in controlling *C. fluminea* populations. *C. fluminea* may be controlled at intake pipes by heating influent water to 37°C (GISD 2013).

***Dreissena polymorpha* Pallas, 1771**

Regulations (pertaining to the Great Lakes region)

Federal law (Lacey Act 1990) prohibits the possession and transportation of Zebra Mussels in the United States unless intended for research.

The following regulations apply to all vessels equipped with ballast water tanks that enter a United States port on the Great Lakes after operating in waters beyond the exclusive economic zone. Vessels are required to exchange ballast water beyond the exclusive zone prior to entering any Great Lakes port. Ballast exchange may also be conducted in waters that are considered a non-threat to the infestation of an aquatic nuisance species in the Great Lakes (Allowable waters stated under [ANS Task Force section 1102\(a\)\(1\)](#)). Vessels are also required to use environmentally sound ballast water management methods if deemed necessary (NASPCA 2000).

In New York, it is unlawful to intentionally release Zebra Mussels into state waters ([New York ECL § 11-0507](#)). In Pennsylvania, it is unlawful to possess, introduce, import, transport, sell, purchase, offer for sale, or barter Zebra Mussels ([58 PA Code § 63.46](#), [71.6](#), and [73.1](#)). In Ohio, it is unlawful to possess, import, or sell Zebra Mussels ([Ohio Administrative Code § 1501:31-19-01\(K\)\(4\)](#)). In Indiana, it is unlawful to import, possess, or release Zebra Mussels into public or private waters ([312 IN Administrative Code § 9-9-3\(d\) and \(e\)](#)). In Michigan, Zebra Mussels are a restricted species ([MI Compiled Laws § 324.41301](#)) and therefore cannot be possessed unless it is to identify, eradicate, or control the species ([MI Compiled Laws § 324.41303](#)). In Wisconsin, it is unlawful to transport, transfer, or introduce Zebra Mussels ([WI Administrative Code § NR 40.05](#)). In Minnesota, it is unlawful to place or attempt to place a watercraft, trailer, or plant harvesting equipment that has Zebra Mussels attached into state water ([MN Statutes § 84D.10](#)). Persons leaving the state are required to drain boats and related equipment during transportation on a public road ([MN Statutes § 84D.10](#), [MN Administrative Rules 6216.0500](#), Kaminski Leduc 2011). Illinois lists Zebra Mussels as injurious species ([IL Administrative Code § 805](#)).

Control

Given the widespread established of Zebra Mussels in the Great Lakes, total eradication is considered impossible with current technologies. No control methods are currently available for open water applications. Control efforts focus primarily on protection of human infrastructure (such as water intakes) and along vectors of spread (such as boats, trailers, gear, etc). Controlling Zebra Mussels to minimize effect on natural and anthropologic systems is expensive, regardless of the method(s) chosen.

Biological

Biological control so far has proven to be ineffective in controlling *Dreissena* species. Predation by migrating diving ducks, fish species, and crayfish may reduce mussel abundance, though the effects are short-lived (Bially and MacIsaac 2000). Other biological controls being researched are selectively toxic microbes and parasites that may play a role in management of *Dreissena* populations (Molloy 1998). Laboratory testing shows strain CL145A of *Pseudomonas fluorescens* (a bacterium) to be highly lethal to Zebra Mussels; capable of eliminating over 90% of adults and 100% of larvae (Molloy 2002, Abdel-Fattah 2011). Commercially, this product is known as Zequanox® and is developed by Marrone Bio Innovations (Abdel-Fattah 2011). Interfering with the synchronization of spawning by adults in their release of gametes could also

offer control of *Dreissena* populations (Snyder et al. 1997). Another approach would be to inhibit the planktonic veliger (larvae) from settling and attaching to a surface to begin development (Kennedy 2002).

Physical

Effective physical controls of *Dreissena* include drawing water from public sources or a groundwater well, infiltration intakes or sandfilter intakes (filter out veligers), thermal treatments, carbon dioxide pellet blasting, high-pressure water jet cleaning, mechanical cleaning, freezing, scraping, scrubbing, pigging, and desiccation. Potential controls include the use of electrical fields, pulse acoustics, low-frequency electromagnetism, ultraviolet light (UV light), and reduced pressure (USACE 2002).

Physical removal of visible vegetation (which may harbor small mussels) from boats, trailers and other equipment being moved from one water body to another is an important method in controlling the spread of Zebra Mussels. Flushing engines, cooling systems, live wells and bilge with water over 43°C will kill veligers and 60°C will kill adults. Air drying equipment for five days will kill most larvae and smaller mussels, but large mussels may survive two weeks out of water.

Placement of water intakes (in areas too deep or otherwise unsuitable for Zebra Mussel colonization) has been used as a form of physical control, but this is less successful in areas which also have Quagga Mussels.

Chemical

Oxidizing chemical control treatments effective against *D. polymorpha* include hypochlorite reaction, chloramine, chlorine dioxide, bromine, ozone, potassium, permanganate, and sodium chlorite. Non-oxidizing chemicals include molluscicides, detoxification, and potassium ions. However, application of these chemical can be extremely detrimental to ecosystem and human health so possible effects should be thoroughly evaluated before use (Boelman et al. 1997, Sprecher and Getsinger 2000).

Various chemical coatings – including copper-based, tributyltin-based, copolymer, vinyl/epoxy, resin and other films - can be applied to structures to prevent the attachment of Zebra Mussels. Tributyltin-based antifoulants are extremely toxic and restricted by federal law (Ohio Sea Grant 1992).

***Dreissena rostriformis bugensis* Andrusov, 1897**

Regulations (pertaining to the Great Lakes region)

Federal law ([Lacey Act 1990](#)) prohibits the possession and transportation of Quagga Mussels in the United States unless intended for research.

The following regulations apply to all vessels equipped with ballast water tanks that enter a United States port on the Great Lakes after operating in waters beyond the exclusive economic zone. Vessels are required to exchange ballast water beyond the exclusive zone prior to entering any Great Lakes port. Ballast exchange may also be conducted in waters that are considered a non- threat to the infestation of an aquatic nuisance species in the Great Lakes (Allowable waters stated under [ANS Task Force section 1102\(a\)\(1\)](#)). Vessels are also required to use

environmentally sound ballast water management methods if deemed necessary (NASPCA 2000).

In Pennsylvania, it is unlawful to possess, introduce, import, transport, sell, purchase, offer for sale, or barter Quagga Mussels ([58 PA Code § 63.46](#), [71.6](#), and [73.1](#)). In Indiana, it is unlawful to import, possess, or release Quagga Mussels into public or private waters ([312 IN Administrative Code § 9-9-3\(d\) and \(e\)](#)). In Michigan, Quagga Mussels are a restricted species ([MI Compiled Laws § 324.41301](#)) and therefore cannot be possessed unless it is to identify, eradicate, or control the species ([MI Compiled Laws § 324.41303](#)). In Wisconsin, it is unlawful to transport, transfer, or introduce Quagga Mussels ([WI Administrative Code § NR 40.05](#)). In Minnesota, it is unlawful to place or attempt to place a watercraft, trailer, or plant harvesting equipment that has Quagga Mussels attached into state water ([MN Statutes § 84D.10](#)). Persons leaving the state are required to drain boats and related equipment during transportation on a public road ([MN Statutes § 84D.10](#), [MN Administrative Rules 6216.0500](#), Kaminski Leduc 2011). Violation penalties can range from a civil fine of \$250-\$1,000 and/or a misdemeanor ([MN Statutes § 84D.13](#)). Illinois lists Quagga Mussels as injurious species ([IL Administrative Code § 805](#)).

Control

Given the widespread established of quagga mussels in the Great Lakes, total eradication is considered impossible with current technologies. No control methods are currently available for open water applications. Control efforts focus primarily on protection of human infrastructure (such as water intakes) and along vectors of spread (such as boats, trailers, gear, etc). Controlling quagga mussels to minimize effect on natural and anthropologic systems is expensive, regardless of the method(s) chosen.

Biological

Biological control so far has proven to be ineffective in controlling *Dreissena* species. Predation by migrating diving ducks, fish species, and crayfish may reduce mussel abundance, though the effects are short-lived (Bially and MacIsaac 2000). Other biological controls being researched are selectively toxic microbes and parasites that may play a role in management of *Dreissena* populations (Molloy 1998). Laboratory testing shows strain CL145A of *Pseudomonas fluorescens* (a bacterium) to be highly lethal to quagga mussels; capable of eliminating over 90% of adults and 100% of larvae (Abdel-Fattah 2011, Molloy 2002). Commercially, this product is known as Zequanox® and is developed by Marrone Bio Innovations (Abdel-Fattah 2011).

Interfering with the synchronization of spawning by adults in their release of gametes could also offer control of *Dreissena* populations (Snyder et al. 1997). Another approach would be to inhibit the planktonic veliger (larvae) from settling and attaching to a surface to begin development (Kennedy 2002).

Physical

Effective physical controls of *Dreissena* include drawing water from public sources or a groundwater well, infiltration intakes or sandfilter intakes (filter out veligers), thermal treatments, carbon dioxide pellet blasting, high-pressure water jet cleaning, mechanical cleaning, freezing, scraping, scrubbing, pigging, and desiccation. Potential controls include the use of electrical fields, pulse acoustics, low-frequency electromagnetism, ultraviolet light (UV light), and reduced pressure USACE 2002).

A recent study has confirmed that thermal treatment of residual water in boats and other water vehicles may be a viable option for managing quagga mussel spread. Increasing temperature and exposure time was found to increase the level of veliger mortality (Craft and Myrick 2011).

Other methods of physical control include exposure and desiccation, manual scraping, high-pressure jetting (including with high temperature water), mechanical filtration, removable substrates, and sonic vibration.

Chemical

Prechlorination has been the most common treatment for control of *Dreissena mussels*, but chlorine concentrations needed for effective control of quagga mussels may reach hazardous levels (Grime 1995). Potassium permanganate has been used as an alternative control, especially for drinking water sources. Ozone is also a potential control. Other molluscicides and anti-fouling coatings may be effective; however, application of these chemical can be extremely detrimental to ecosystem and human health so possible effects should be thoroughly evaluated before use (Boelman et al. 1997, Sprecher and Getsinger 2000).

Other

Other potential methods of Quagga Mussel control include oxygen deprivation, radiation, and electric currents.

***Lasmigona subviridis* Conrad, 1835**

Regulations (pertaining to the Great Lakes region)

NatureServe lists this species as ‘global vulnerable’ at moderate risk of extinction in its native range.

There are no regulations for this species as an invasive in the Great Lakes region.

Control

Management research focuses primarily on preservation of populations within the native range.

Biological

Competition with dreissenid mussels will likely limit its expansion in the Great Lakes.

Chemical

A wide array of chemical molluscicides are available, but are not species-specific and may harm native species to a greater extent than non-natives.

Molluscicides are typically classified as either oxidizing or non-oxidizing compounds. Oxidizing chemicals include chlorine, chlorine dioxide, chloramines, ozone, bromine, hydrogen peroxide, and potassium permanganate. Non-oxidizing chemicals (including organic film-forming antifouling compounds, gill membrane toxins, and nonorganics) can be classified into several distinct groups: quaternary and polyquaternary ammonium compounds; aromatic hydrocarbons; endothal as the mono (N, N-dimethylalkyl amine) salt; metals and their salts (e.g., copper sulfate formulations); and niclosamide (including some formulations of Bayluscide). Bayluscide was initially developed as a Sea Lamprey larvicide, but has molluscicidal activity. While some of these products are biodegradable, many require detoxification or deactivation to meet state and Federal discharge requirements (USACE 2012a).

***Pisidium amnicum* Müller, 1774**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

A wide array of chemical molluscicides are available, but are not species-specific and may harm native species to a greater extent than non-natives.

Molluscicides are typically classified as either oxidizing or non-oxidizing compounds. Oxidizing chemicals include chlorine, chlorine dioxide, chloramines, ozone, bromine, hydrogen peroxide, and potassium permanganate. Non-oxidizing chemicals (including organic film-forming antifouling compounds, gill membrane toxins, and nonorganics) can be classified into several distinct groups: quaternary and polyquaternary ammonium compounds; aromatic hydrocarbons; endothal as the mono (N, N-dimethylalkyl amine) salt; metals and their salts (e.g., copper sulfate formulations); and niclosamide (including some formulations of Bayluscide). Bayluscide was initially developed as a Sea Lamprey larvicide, but has molluscicidal activity. While some of these products are biodegradable, many require detoxification or deactivation to meet state and Federal discharge requirements (USACE 2012a).

***Pisidium henslowanum* Shepard, 1825**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

A wide array of chemical molluscicides are available, but are not species-specific and may harm native species to a greater extent than non-natives.

Molluscicides are typically classified as either oxidizing or non-oxidizing compounds. Oxidizing chemicals include chlorine, chlorine dioxide, chloramines, ozone, bromine, hydrogen peroxide, and potassium permanganate. Non-oxidizing chemicals (including organic film-forming antifouling compounds, gill membrane toxins, and nonorganics) can be classified into several distinct groups: quaternary and polyquaternary ammonium compounds; aromatic hydrocarbons; endothal as the mono (N, N-dimethylalkyl amine) salt; metals and their salts (e.g., copper sulfate

formulations); and niclosamide (including some formulations of Bayluscide). Bayluscide was initially developed as a Sea Lamprey larvicide, but has molluscicidal activity. While some of these products are biodegradable, many require detoxification or deactivation to meet state and Federal discharge requirements (USACE 2012a).

Pisidium moitessierianum Paladilhe, 1866

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Research on drawdown as a potential control technology for *P. moitessierianum* suggests that it will be in-effective as a control in most situations (Mouthon 2011). Pisidiid clams showed poor resistance to a brief period of drying of the habitat, but also demonstrated high rates of recovery. In the study sites, drawdown did cause a decline in *P. moitessierianum*, but the population recovered quickly, becoming the dominant bivalve in the following year.

Chemical

A wide array of chemical molluscicides are available, but are not species-specific and may harm native species to a greater extent than non-natives.

Molluscicides are typically classified as either oxidizing or non-oxidizing compounds. Oxidizing chemicals include chlorine, chlorine dioxide, chloramines, ozone, bromine, hydrogen peroxide, and potassium permanganate. Non-oxidizing chemicals (including organic film-forming antifouling compounds, gill membrane toxins, and nonorganics) can be classified into several distinct groups: quaternary and polyquaternary ammonium compounds; aromatic hydrocarbons; endothal as the mono (N, N-dimethylalkyl amine) salt; metals and their salts (e.g., copper sulfate formulations); and niclosamide (including some formulations of Bayluscide). Bayluscide was initially developed as a Sea Lamprey larvicide, but has molluscicidal activity. While some of these products are biodegradable, many require detoxification or deactivation to meet state and Federal discharge requirements (USACE 2012a).

Pisidium supinum Schmidt 1850

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

A wide array of chemical molluscicides are available, but are not species-specific and may harm native species to a greater extent than non-natives.

Molluscicides are typically classified as either oxidizing or non-oxidizing compounds. Oxidizing chemicals include chlorine, chlorine dioxide, chloramines, ozone, bromine, hydrogen peroxide, and potassium permanganate. Non-oxidizing chemicals (including organic film-forming antifouling compounds, gill membrane toxins, and nonorganics) can be classified into several distinct groups: quaternary and polyquaternary ammonium compounds; aromatic hydrocarbons; endothal as the mono (N, N-dimethylalkyl amine) salt; metals and their salts (e.g., copper sulfate formulations); and niclosamide (including some formulations of Bayluscide). Bayluscide was initially developed as a Sea Lamprey larvicide, but has molluscicidal activity. While some of these products are biodegradable, many require detoxification or deactivation to meet state and Federal discharge requirements (USACE 2012a).

Sphaerium corneum Linnaeus, 1758

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

A wide array of chemical molluscicides are available, but are not species-specific and may harm native species to a greater extent than non-natives.

Molluscicides are typically classified as either oxidizing or non-oxidizing compounds. Oxidizing chemicals include chlorine, chlorine dioxide, chloramines, ozone, bromine, hydrogen peroxide, and potassium permanganate. Non-oxidizing chemicals (including organic film-forming antifouling compounds, gill membrane toxins, and nonorganics) can be classified into several distinct groups: quaternary and polyquaternary ammonium compounds; aromatic hydrocarbons; endothal as the mono (N, N-dimethylalkyl amine) salt; metals and their salts (e.g., copper sulfate formulations); and niclosamide (including some formulations of Bayluscide). Bayluscide was initially developed as a Sea Lamprey larvicide, but has molluscicidal activity. While some of these products are biodegradable, many require detoxification or deactivation to meet state and Federal discharge requirements (USACE 2012a).

In short-term experiments, *S. corneum* can reduce the bioaccumulation of 2, 4, 5-trichlorophenol (TCP) by closing their shell valves (Heinonen et al. 1997) – this reaction to chemical stimuli generally may limit the usefulness of chemical molluscicides against this species.

Bithynia tentaculata

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are currently no documented successful methods for the control of Faucet Snails in open water ecosystems.

Biological

There are no known biological control methods for this species.

Physical

In an attempt to limit the number of Faucet Snails in the Upper Mississippi River National Wildlife and Fish Refuge, biologists experimented with covering colonies of Faucet Snails with sand (Williams 2007). The success of this method was undocumented.

Chemical

There are no known chemical control methods for this species.

Cipangopaludina chinensis malleata

Regulations (pertaining to the Great Lakes region)

Chinese Mysterysnail is a regulated invasive species in Minnesota ([MN Administrative Rules § 6216.0260](#)) and a restricted species in Wisconsin ([Wisconsin Administrative Rules § 40.05](#)).

Control

Specific control methods for the Chinese Mysterysnail have yet to be developed.

Biological

Manipulation of predator fishes and turtles that eat snails may be useful in the control of snail populations. However, as a relatively large snail species, *Cipangopaludina chinensis* may escape predation by smaller fishes.

Physical

Preliminary research demonstrates that *C. chinensis* will not migrate upstream against a small current (Rivera 2008). Authors suggest that acceleration of current may be an important management tool for preventing upstream spread.

Dessication (drying) is not an effective control method for *C. chinensis*. Field experiments under mesic conditions indicated that this snail can survive exposure to air for at least four weeks (Havel 2011).

Chemical

There are copper compounds that are sold as snailicides but they are usually not selective in the snails they kill. With Chinese Mysterysnails possessing the ability to “close up”, more damage would probably occur to native snails in the treatment area than to the target pest.

Cipangopaludina japonica

Regulations (pertaining to the Great Lakes region)

Japanese Mysterysnail is a regulated invasive species in Minnesota ([MN Administrative Rules § 6216.0260](#))

Control

Specific control methods for the Japanese Mysterysnail have yet to be developed – most available control suggestions are based on research with the closely related *Cipangopaludina chinensis*.

Biological

Manipulation of predator fishes and turtles that eat snails may be useful in the control of snail populations. However, as a relatively large snail species, *Cipangopaludina japonica* may escape predation by smaller fishes.

Physical

Preliminary research demonstrates that *C. chinensis* will not migrate upstream against a small current (Rivera 2008) – it is not known whether *C. japonica* is similarly restricted. Acceleration of current may be an important management tool for preventing upstream spread.

Dessication (drying) is not likely to be an effective control method for *C. japonica*. Field experiments under mesic conditions indicated that *C. chinensis* and other snails with opercula can survive exposure to air for at least four weeks (Havel 2011).

Chemical

There are copper compounds that are sold as snailicides but they are usually not selective in the snails they kill. With Japanese Mysterysnails possessing opercula, more damage would probably occur to native snails in the treatment area than to the target pest.

Elimia virginica

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Specific control methods for *Elimia virginica* have yet to be developed.

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

There are copper compounds that are sold as snailicides but they are usually not selective in the snails they kill. With *E. virginica* possessing the ability to close their shells (opercula), more damage would probably occur to native snails in the treatment area than to the target pest.

Gillia altilis

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Specific control methods for *Gillia altilis* have yet to be developed.

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

There are copper compounds that are sold as snailicides but they are usually not selective in the snails they kill.

Potamopyrgus antipodarum

Regulations (pertaining to the Great Lakes region)

New Zealand Mudsnailes are listed as a prohibited species in Wisconsin ([Wisconsin Administrative Rules § 40.05](#)) and Minnesota ([MN Administrative Rules § 6216.0260](#)).

Control

Many times New Zealand Mudsnailes may be in a river or lake where chemical eradication will not be feasible and physical eradication difficult. Areas where eradication may be possible include small lakes and ponds, waterbodies that can be temporarily hydrologically separated.

Biological

Parasites of New Zealand Mudsnailes from New Zealand may also become useful to control population size by inhibiting reproduction. Studies of the efficacy and specificity of a trematode parasite from the native range of New Zealand Mudsnailes as a biological control agent have shown positive results so far (Dybdahl et al. 2005).

Physical

New Zealand Mudsnailes easily hitchhike with fish and aquatic plants. Inspection of boats/trailers/gear is essential, but equipment should also be dried thoroughly before moving from infected to uninfected waters. Putting fishing gear in a freezer for 6-8 hours will kill all attached New Zealand Mudsnailes (Medhurst 2003, Richards et al. 2004). Putting fishing gear in water maintained at 49°C for a few minutes will eliminate New Zealand Mudsnailes (Medhurst 2003). Mudsnailes can survive at 43.3°C so the water temperature needs to be accurate. Dry fishing gear at 28-30°C for at least 24 hours or at 40°C for at least two hours (Richards et al. 2004).

For (aquaculture) facilities where no known New Zealand Mudsnaile contamination occurs, close visual inspection of water systems, raceways, stocking equipment, as well as regular gut content analysis can detect the arrival of snails before they can be spread .

Physical treatments include the use of temperature, humidity or desiccation to kill the target species. This includes draining the infested areas. New Zealand Mudsnaills can survive for long periods in a cool damp environment; however, draining the areas where they are congregated and exposing them to sunlight during the summer months may be sufficient for eradication. Using a flame thrower in a hatchery situation against the walls of raceways will kill any mudsnails attached. Mudsnaills cannot withstand warm temperatures (Dwyer et al. 2003, Richards et al. 2004) or low humidity situations (Dwyer and Kerans, unpublished, Richards et al. 2004). Alternately, if an infested area could be drained in the winter and the substrate is frozen to a depth containing the mudsnails, then total eradication will occur. There is preliminary evidence that hydrocyclonic separators may also be a useful tool to decontaminate fish hatchery water supplies and prevent the spread of New Zealand Mudsnaills within a hatchery.

It has been suggested that barriers such as copper stripping or electrical weirs may limit volitional movement of New Zealand Mudsnaills, particularly as a means of protecting high risk sites like fish hatchery water systems. Some investigations are underway but there is no applicable tool available yet.

Chemical

Chemical methods used to eradicate New Zealand Mudsnaills include: Bayer 73, copper sulfate, and 4-nitro-3-trifluoromethylphenol sodium salt (TFM). The only molluscicide known to have been tested against New Zealand Mudsnaills is Bayluscide (niclosamide). Preliminary investigations also suggest that copper and carbon dioxide under pressure may prove useful in both decontaminating fish hatchery water supplies and preventing spread into uncontaminated areas of a hatchery. Ozone has not been shown to be effective in killing New Zealand Mudsnaills in a hatchery environment.

The most effective solutions for killing New Zealand Mudsnaills which can be used in the field, according to this research are copper sulfate (252mg/L Cu), benzethonium chloride (1,940 mg/L) and 50% Commercial Solutions Formula 409® Cleaner Degreaser Disinfectant.

Copper sulfate, hyamine and hydrogen peroxide, have all been used to control New Zealand mud snails (IJC 2011).

Radix auricularia

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Specific control methods for *Radix auricularia* have yet to be developed.

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

There are copper compounds that are sold as snailicides but they are usually not selective in the snails they kill.

Valvata piscinalis

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

Specific control methods for *Valvata piscinalis* have yet to be developed.

Biological

Manipulation of predator fishes and turtles that eat snails may be useful in the control of snail populations. However, as a relatively large snail species, *V. piscinalis* may escape predation by smaller fishes.

Physical

There are no known physical control methods for this species.

Chemical

There are copper compounds that are sold as snailicides but they are usually not selective in the snails they kill. With *V. piscinalis* possessing the ability to “close up”, more damage would probably occur to native snails in the treatment area than to the target pest.

Viviparus georgianus I. Lea, 1834

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

Specific control methods for *Viviparus georgianus* have yet to be developed.

Biological

Manipulation of predator fishes and turtles that eat snails may be useful in the control of snail populations. However, as a relatively large snail species, *V. georgianus* may escape predation by smaller fishes.

Physical

There are no known physical control methods for this species.

Chemical

There are copper compounds that are sold as snailicides but they are usually not selective in the snails they kill. With *V. georgianus* possessing the ability to “close up”, more damage would probably occur to native snails in the treatment area than to the target pest.

A.13 Plants

Agrostis gigantea Roth

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species. However, the Michigan Department of Natural Resources (MIDNR and MIDEQ 2009) recommends use of Redtop for erosion control in forested land. Moreover, Redtop's close cousin, Creeping Bentgrass (*A. stolonifera*), is commonly put to commercial use on golf course putting greens, tees, and fairway turf (Banks et al. 2004).

Control

Biological

There are no known biological control methods for this species.

Physical

This species has rhizomal roots that can grow to a depth of four feet (Tilley et al. 2010), making mechanical removal of Redtop difficult, time-consuming, and unlikely to be successful, as any significant portions of the rhizome that are not removed from the soil could generate new plants.

Continuous grazing or mowing to a height of less than three inches may result in temporary control (Tilley et al. 2010).

Chemical

Due to the tendency for this species to form dense, monospecific turfs (Tilley et al. 2010) herbicide application should be easy to manage. Increased grass chlorosis (yellowing of the plant due to reduced chlorophyll production) results when herbicides are applied during cool weather (10°C) (McCullough and Hart 2006).

Among available herbicides, Redtop is very susceptible to atrazine (Carey 1995). It can also be well controlled by glyphosate. However, glyphosate-resistant strains of the related Creeping Bentgrass (*A. stolonifera*) do exist and are becoming more common (Hart et al. 2005); commercial release is likely to increase the potential for unintended transfer of the herbicide resistance gene to *A. gigantea* (NatureServe 2008).

Given the ease of hybridization with other *Agrostis* species (Tilley et al. 2010), if control methods listed above are not successful, alternative chemical control methods may also be considered. For instance, in tests conducted in Virginia, the closely related Creeping Bentgrass experienced at least a 92% die-off after applying isoxaflutole, imazaquin, or mesotrione in two to three sequential applications (Beam et al. 2006). Fluazifop-P has also been effective in controlling bentgrass species (Hart et al. 2005). In addition, Creeping Bentgrass seems to be susceptible to rimsulfuron, especially when the herbicide is kept dry after application (Barker et al. 2005).

***Alnus glutinosa* (L.) Gaertn.**

Regulations (pertaining to the Great Lakes region)

There are conflicting recommendations on the regulation of Black Alder. Indiana recommends that this species be inter-planted to improve soil quality and “protect other valuable trees” (IDNR n.d.). The state also includes Black Alder on a list of invasive exotics plants whose “use in landscaping and re-vegetation projects should be avoided or limited when possible” (Homoya 2010). Black Alder is also listed as a “plant to avoid” in Wisconsin’s planting guide (WIDNR n.d.). It is a recommended tree for urban environments in Minnesota (Johnson and Himanga 2009).

Control

For the most effective control, the United States Department of Agriculture Natural Resources Conservation Service (2006) recommends treating Black Alder with a combination of physical and chemical methods.

Biological

There are no known biological control methods for this species.

Physical

Trees should be felled and then herbicide applied to the stumps to prevent sprouting (USDA NRCS 2006).

Chemical

Effective herbicides include napropamide (preemergence) (Willoughby et al. 2007), glyphosate via exposed stump application (Kelly and Southwood 2006) triclopyr triethylamine via foliar application (Champion et al. 2008).

***Alopecurus geniculatus* L.**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

Toogood et al. (2008) found that Water Foxtail tolerates periods of waterlogging and flooding. This species does not grow as well under a drier water regime. Moreover, in locations where water level manipulation is used for wetland management, flooding should not be used as a method for controlling this species.

Alopecurus geniculatus has the ability to reproduce via seed dispersal and vegetatively from roots nodes (Klinkenberg 2010). Each individual seed has a mass of approximately 0.8 mg, which is light enough to be transported short distances by wind (Klein 2011). If seeds are driven

below this depth, they are unlikely to germinate. A variety of tilling methods can be used both to kill seedlings after emergence and to bury seeds deep into the soil to prevent germination (Curran 2009). Seeds of Water Foxtail can remain viable in soil for at least three years (Roberts 1986). Therefore, management activity would be most effective if performed before seeds have a chance to disperse.

Chemical

Complete eradication of a related species, *Alopecurus pratensis* L. (Meadow Foxtail), was achieved when treated in late summer with glyphosate at 1.0 kg/ha (0.89 lbs/ac). Use of fluazifop also controls *A. pratensis* L., but has also been reported to be toxic to fishes and aquatic invertebrates. Absorption of sethoxydon, another herbicide used to control *A. pratensis* L., can be increased when it is simultaneously applied with oil adjuvant and a non-ionic surfactant (OSU 2005). If managers expect dense populations of foxtail species, grass herbicide rates may need to be increased or the application timing altered to include split applications or postemergence control (Curran 2009).

***Butomus umbellatus* L.**

Regulations (pertaining to the Great Lakes region)

Butomus umbellatus is listed as a prohibited species in Michigan, Minnesota, and Illinois, and as a restricted species (but still available) in Wisconsin (GLPANS 2008, Jensen 2011). A recent survey of Minnesota Nursery and Landscape Association Members revealed that 80% of respondents were incorrect or unsure when judging the non-native status of *B. umbellatus* despite its prohibited use in Minnesota (Peters et al. 2006). It is on New York State's Interim Invasive Species Plant List (NYSDEC 2011). In Pennsylvania it is labeled as a medium-high threat to native ecosystem and in Ohio it is classified as a "well established invasive plant" (Ohio Division of Natural Areas and Preserves and Nature Conservancy 2000, Higman and Campbell 2009).

Control

Butomus umbellatus has a similar appearance to some native plants, such as common bulrush (*Typha latifolia*) (Jensen 2011). Care should be taken to first identify the plants in question before control actions are taken.

Biological

In its native range in Europe, ducks have been known to graze extensively on Flowering Rush (Hroudová et al. 1996).

Physical

B. umbellatus spreads by floating seeds and vegetatively by rhizomes and root pieces (Campbell et al. 2010, Jensen 2011). Great care to remove all parts of the plant should be taken when implementing a physical method of control. To ensure removed plants will not sent out shoots, thoroughly dry all plant pieces (Jensen 2011).

Cutting Flowering Rush below the water surface will not kill the plant, but it will reduce its abundance, and therefore, its ability to spread. Multiple cuts may be needed throughout the

growing season (Jensen 2011). Cutting later may remove most of that season's rhizome growth (Hroudová et al. 1996). Hand digging may be a viable option for removing isolated plants (Jensen 2011). Techniques involving raking or pulling are not recommended (Jensen 2011).

Decreases in water level promotes establishment of Flowering Rush, suggesting that water level reductions is not a method of control (Parkinson et al. 2010).

Chemical

Control with the use of herbicides is difficult because these chemicals easily miss or fall off of the narrow, slightly twisted leaves of Flowering Rush (Seizer 2009, Parkinson et al. 2010). There is currently no herbicide that targets *B. umbellatus*, but initial testing shows that a mid-summer application of imazapyr (labeled as the herbicide 'Habitat') during calm weather may be effective (Parkinson et al. 2010, MN DNR 2012b). Some applicators report control of *Butomus umbellatus* with glyphosate; others report control of submerged *B. umbellatus* with diquat in sites that have limited dilution potential.

Cabomba caroliniana

Regulations (pertaining to the Great Lakes)

Cabomba caroliniana is prohibited in Wisconsin, Illinois and Michigan (GLPANS 2008). In Minnesota, *C. caroliniana* can be possessed, sold, bought, and transported, but it is illegal to release it into the environment (MN DNR 2013b). The New York Invasive Species Council ranks this species as posing a high ecological threat (NYISC 2010).

The Great Lakes Indian Fish & Wildlife Commission have not found *C. caroliniana* in their ceded territories, but recommended immediate control upon detection (Falck and Garske 2003).

Control

Biological

When fed on by crayfish and snails, *C. caroliniana* induces a chemical defense mechanism deter both herbivores and microbes that typically attack plants via openings left by herbivores (Morrison and Hay 2011).

Grass Carp, *Ctenopharyngodon idella*, will eat *C. caroliniana*, but it is not their preferred food source.

Diploid (fertile) Grass Carp are illegal for use in some states, such as Minnesota (MN DNR 2013a). The use of certified triploid (sterile) Grass Carp is allowed in New York and Pennsylvania, with the correct permits (NY DEC 2013, Shiels and Hartle n.d.)

Physical

Cabomba caroliniana becomes brittle late in the growing season. Physical control efforts should not be tried during this time because broken pieces can develop into new plants (IISCTC 2007).

Physical cutting and removal of *C. caroliniana* is most effective on large infestations in closed water bodies (ADEH 2003). In areas of sufficient size and depth, this can be done with floating

mechanical (CSIRO Entomology 2011). However, given the low probability of removing every plant fragment, this method is likely to only provide nuisance relief for a few weeks (ADEH 2003). Efficacy can be improved by using tools such as a venturi dredge, which acts like a vacuum cleaner to *C. caroliniana* fanwort fragments and the root ball (WIDNR 2012d).

Water-level drawdowns have reduced growth of *C. caroliniana* populations in some areas in southern Wisconsin (WIDNR 2012d). Extreme drying, in which the root ball dries completely, is needed or the plant will return (Forest Health Staff 2006b, IISCTC 2007).

Ensuring wash-downs of boats, trailers, and other equipment can reduce the spread of Carolina Fanwort (IISCTC 2007).

Chemical

Chemical defense also indicates that the use of biocontrol agents on invasive populations of *C. caroliniana* may not be a viable approach (Morrison and Hay 2011).

Precise application of appropriate herbicides to submerged *C. caroliniana* can be problematic and should be done with great care to avoid desired species (ADEH 2003).

Herbicides containing endothall or fluridone have been effective in controlling *C. caroliniana* (ADEH 2003, Forest Health Staff 2006b). In laboratory tests, the application of diquat and flumioxazin (separately) resulted in a greater than 50% reduction in photosynthesis of *C. caroliniana*, however, these trials did not include field testing (Bultemeier et al. 2009).

Michigan reports *Cabomba* has been controlled in one lake using whole lake 20 ppb fluridone. Carolina Fanwort has also been reported to be sensitive to 2,4-D (Wilson et al. 1997). Michigan further reports *Cabomba* has been controlled using spot treatment of 200 ppb flumioxazin in many waterbodies. However, they report that *Cabomba* populations have not been controlled in field trials with diquat or endothall.

***Carex acutiformis* Ehrh.**

Regulations (pertaining to the Great Lakes region)

Based on its competitive dominance in Stony Swamp, Ottawa, Ontario, *Carex acutiformis* was identified as a high priority invasive plant by the Canada Botanical Association in 2004, ranking 14th overall among invasive plants (Catling 2005).

Control

Biological

There are no known biological control methods for this species.

Physical

Harvesting, either mechanically or by hand, may be a viable option, depending on site conditions. Extreme care should be taken to remove all parts of the roots system and rhizomes to prevent further spread of the species. This method is unlikely to eradicate Swamp Sedge, but it will help to prevent population expansion. Physical control methods are most effective when completed before seed production (USACE 2012b).

Seeds do not germinate when they are seven cm below the soil surface (Schütz 1998). Thus, tilling the soil to push the existing seeds deeper into the substrate may be an effective method for reducing the prevalence of Swamp Sedge (Curran et al. 2009).

Carex acutiformis does not extend into open water deeper than 55 cm, and it is able to cope with low water levels. Stands of *C. acutiformis* did not decrease immediately after moderate reduction in water level, suggesting that water level alteration may not be an effective form of control (Lawniczak et al. 2010).

Chemical

Currently, no peer-reviewed literature examines the efficacy of herbicides against Swamp Sedge. According to the Center for Ecology and Hydrology all sedges are susceptible to glyphosate (CEH 2004a); an application in mid to late summer will maximize translocation and control of rhizomes (USACE 2012b). Imazapyr has also been effective in controlling some sedge species (USACE 2012b).

***Carex disticha* Huds.**

Regulations (pertaining to the Great Lakes region)

Tworank Sedge has not been found in the United States, but it is classified as an “introduced species” in Ontario, Canada (Canadensys 2012, USDA NRCS 2012).

Control

Biological

There are no known biological control methods for this species.

Physical

Carex spp. are able to withstand water-logged conditions, suggesting that water level increase may not be an effective form of control (Riutta et al. 2007).

Chemical

There are no known chemical control methods for *Carex disticha*; however, imazapyr has been effective in controlling other sedge species (USACE 2012b).

Chenopodium glaucum

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

The occurrence of *Chenopodium glaucum* can be minimized in crop fields by timely spring tillage and early autumn plowing. Crop rotation with winter cereal grains may also be effective in controlling *C. glaucum* (Larina 2008).

Chemical

There are no known chemical control methods for this species.

Cirsium palustre

Regulations (pertaining to the Great Lakes region)

Cirsium palustre is considered to be a medium to high threat species in New York and Michigan

(Higman and Campbell 2009, NYISC 2010). *C. palustre* became a prohibited species in some Wisconsin counties with the creation of Wisconsin's Invasive Species Identification, Classification and Control Rule ([Wisconsin Administrative Rules § 40.05](#); Terrestrial Herbaceous Plants Species Assessment Group 2007). Even though it is not present in Minnesota, it is characterized as a severe threat to native ecosystems based on its impact in other locations (Minnesota Invasive Species Advisory Council 2009). Marsh Thistle is listed as an introduced species in Ontario and Quebec (Canadensys 2012). It is considered noxious in some regions of British Columbia under the Weed Control Act (OLA and MAFF 2002). Marsh Thistle was categorized as a priority species for removal by the Great Lakes Indian Fish & Wildlife Commission (GLIFWC) in 2006, but this priority was reduced in subsequent comprehensive reports (Falck et al. 2006, 2009ab).

Control

Biological

While there are no specific biocontrol agents for *C. palustre* (GLIFWC 2006), herbivory by a variety of species may be beneficial but requires additional research.

Promising biocontrol candidates include a European seedhead fly, *Terellia ruficauda* (Fraser 2000, OLA and MAFF 2002); the seed-eating weevil, *Rhinocyllus conicus*, currently undergoing experimental trial in the Robson Valley Forest District, British Columbia (OLA and MAFF 2002, USDA Forest Service 2005b); and the glassy cutworm, *Apamea devastator* (native in New York and Ohio; Volger and Stressler 2011). The latter is an indiscriminate herbivore known to feed on *C. palustre* and may help control Marsh Thistle; however, this moth feeds on a broad spectrum of additional plants.

Larvae of the artichoke plume moth (*Platyptilia carduidactyla*) also feed on Marsh Thistle, but as its common name suggests, this species is considered a pest to artichokes. Furthermore, the moth's native range is south of the Great Lakes (Winston et al. 2008). Occasionally *Cheilosisia corydon*, a fly native to Italy, feeds on Marsh Thistle (Winston et al. 2008). This fly was released in Oregon in 1991 to control several invasive thistle populations. However, since its release, *C. corydon* populations have attacked native and exotic thistles indiscriminately (ODA Plant Division 2011). Additional insects that feed on and/or use *C. palustre* for part of their life cycle are listed on these websites (Lindsey 2005, Hogan n.d.).

Goats are attracted to the flowering stage of many thistles, including *C. palustre*. Only about 0.5% of thistle seeds that pass through their digestive systems remain viable, making it unlikely that they would aid in the spread of this species. Effective grazing could reduce Marsh Thistle populations, although it is unclear whether grazing would ultimately control *C. palustre* via the trampling of rosettes or facilitate its spread through the creation of safe sites for germination (Fraser 2000). Reseeding of native vegetation may enhance the success of prior control efforts. Moreover, goats do not select for Marsh Thistle and may also eat native thistles in intermingled communities (Popay and Field 1996).

Van Leeuwen found that a combination of European grazers (rabbits, the hoverfly *Cheilosisia grossa*, and *Epiblema scutula*) resulted in an approximately 30% reduction in flower heads on *Cirsium palustre*. Furthermore, plants that had suffered predation had a reduced stem height,

resulting in a reduced seed dispersal distance of surviving achenes (van Leeuwen 1983). Additional research is needed to determine if native species of rabbits and insects could have similar results on controlling *C. palustre* in the Great Lakes.

Physical

All physical control efforts need to be carefully executed and monitored for several years before reducing *C. palustre* infestations (GLIFWC 2006, Sheehan 2007).

Where infestations are small, hand pulling may be effective. In Chequamegon-Nicolet National Forest (CNNF), Wisconsin, individual plants were mechanically controlled by cutting the root just below the surface with a spade (USDA Forest Service 2005a, GLIFWC 2006). This method is most effective if completed before flowering so that all plant material can be left on site to decompose (Invasive Plant Council of British Columbia 2008). If this method is implemented while flowers and seeds are present, flower heads must be bagged and removed from site; the remaining plant material can be left on site (Invasive Plant Council of British Columbia 2008).

Mowing before plants flower may reduce the release of seeds (OLA and MAFF 2002); however, there is a risk of regrowth with extra flower heads, and extensive mowing of rosettes could promote growth of this early successional species once mowing ceases (Fraser 2000, Nordin et al. 2008). Repeated close mowing can reduce a *C. palustre* infestation in three to four years (Gumbart 2012.). Mowing a minimum of three times per growing season can be enough to weaken the following year's population (Boos et al. 2010). However, in a study by Falinska (1999), the density of *C. palustre* seeds in the soil decreased dramatically as time since last mowing increased, indicating that frequent mowing may actually increase the density of Marsh Thistle in the seedbank.

Little is known about Marsh Thistle response to fire as a control strategy (Gucker 2009).

Chemical

Foliar spray with clopyralid (e.g., Transline®, Curtail®, Reclaim®) or metsulfuron-methyl is the preferred chemical treatment for *C. palustre* in CNNF, Wisconsin (USDA Forest Service 2005b). However, glyphosate (e.g., Rodeo®, Aquamaster®, Roundup®) must be used in areas that are wet or near open water (USDA Forest Service 2005b). Either of these two treatments are most effective if applied in the spring when plants are 6 to 10 inches tall and still in the budding stage or applied directly to the flower heads in the fall (USDA Forest Service 2005b, Boos et al. 2010). To minimize damaging other non-target species when using glyphosate, stems should be cut close to the ground and a small amount sprayed onto the cut area (Sheehan 2007).

Conium maculatum

Regulations (pertaining to the Great Lakes region)

Conium maculatum is a well-established invasive species in Ohio and has been designated as a prohibited noxious weed (Ohio Division of Natural Areas and Preserves and Nature Conservancy 2000, USDA NRCS 2012). The possession, transportation, transfer, or introduction of Poison Hemlock is restricted in all Wisconsin counties except: Crawford, Dane, Grant, Green, Iowa, Lafayette, Richland, Rock, and Sauk (WIDNR 2010a). The Robert W. Freckman Herbarium of

the University of Wisconsin lists an eradication notice for *C. maculatum* (Robert W. Freckman Herbarium 2012). However, as of 2007 it was not illegal to sell Poison Hemlock in Wisconsin (Annen 2007).

Regionally, the Great Lakes Indian Fish & Wildlife Commission (GLIFWC) classify this species as capable of causing moderate to severe ecological impacts and/or having limited effective control options available (Falck and Garske 2003).

Control

Physical or chemical removal of *C. maculatum* individuals is relatively easy, but complete eradication may be difficult due a viable seed bank and reintroductions. Management efforts may need to be continued for several years to be effective (J. McHenry pers. comm. in Pitcher 1985).

Biological

The Poison Hemlock defoliating moth (*Agonopterix alstroemeriana*) was accidentally introduced to the United States, but it is now being investigated as a potential biocontrol agent because of its monophagous (feeding on a single food source) association with *C. maculatum* (Castells and Berenbaum 2006). Hemlock Moth larvae feed on the young stem tissue, flowers, and seeds (Forest Health Staff 2006d). High densities of *A. alstroemeriana* have been effective drivers of plant mortality in *C. maculatum* stands in the western United States, where several hundred larvae have been reported from a single plant. However, as a chemical defense, alkaloid production appears to increase with *A. alstroemeriana* herbivory, potentially driving surviving populations to higher levels of toxicity over time (Castells et al. 2005). Furthermore, *A. alstroemeriana* was found to be targeted by a predatory wasp (*Euodynerus foraminatus*) in Illinois, suggesting that the effectiveness of biocontrol may be lessened in the Midwest and other locations where *E. foraminatus* exerts top-down pressure on *A. alstroemeriana* (Castells and Berenbaum 2008). Although *A. alstroemeriana* is widespread in the United States, larvae may still be difficult to obtain for biocontrol purposes (Castells and Berenbaum 2006).

Trichoplusia ni, the cabbage looper, is a generalist lepidopteran that is found throughout the United States and occasionally feeds on *C. maculatum*. Overall growth of *T. ni* is not stunted, but larvae raised on diets enriched with the piperidines found in *C. maculatum* develop slower. A prolonged larval stage makes *T. ni* more vulnerable to predators and could reduce overall biocontrol capabilities (Castells and Berenbaum 2008).

Papilio poluxenes, Black Swallowtail Butterfly, will lay eggs on *C. maculatum*, but a study conducted in central New York found low larvae survivorship (Feeny et al. 1985).

Conium maculatum is capable of being infected by multiple viruses, including Ring Spot Virus, Carrot Thin Leaf Thin Virus (CTLV), Alfalfa Mosaic Virus (AMV), and Celery Mosaic Virus (CeMV) (Howell and Mink 1981). However, viral infections appear to stunt growth rather than cause mortality, diminishing their potential for biocontrol (Howell and Mink 1981, Pitcher 2004). Another disadvantage to using these types of biological control agents is the potential for them to escape in neighboring habitats, especially agricultural fields (J. McHenry pers. comm. in Pitcher 1985).

Physical

Care should be taken in handling this toxic plant; it is recommended that gloves are worn (Pitcher 2004). If any body part comes into contact with any part of the plant, be sure to wash it thoroughly (OLA and MAFF 2002). Hand pulling or digging out the taproot are effective methods of control for small populations, especially when the soil is moist (WIDNR 2008). Mowing close to the ground is another option of mechanical control if the blade is close to the ground (WIDNR 2008). A dust mask should be worn for protection to avoid inhaling toxins while mowing (King County 2011). In both cases, efforts are most effective if completed before the plants flower and multiple follow-up efforts should be taken to prevent regrowth or new growth (Parsons 1973, Pitcher 2004, WIDNR 2008, Woodard 2008). Poison Hemlock remains for several years after death and should be removed where there is a risk of consumption by livestock, wildlife, or children (Pitcher 2004).

Chemical

Effective control of large infestations may require chemical agents (WIDNR 2008, King County 2011). Chemical control of rosettes (before flowering) is a common form of management for *Conium maculatum* because it is such a prolific seed producer (Forest Health Staff 2006d, Woodard 2008). Application of herbicides early in spring when sprouts are just emerging may also result in effective control, but if *C. maculatum* has a large presence in the seed bank, multiple applications may be needed (Forest Health Staff 2006d).

Several herbicides—including chlorsulfuron, hexazinone, imazapic, glyphosate (Round-Up®), metribuzin, metsulfuron, picloram, triclopyr, terbacil, imazapic plus glyphosate, and metsulfuron plus 2,4-D plus dicamba—were found to be the most effective chemical agents (Pitcher 1985, Jeffery and Robinson 1990, OLA and MAFF 2002, Forest Health Staff 2006d, Woodward 2008, King County 2011). The surrounding plant community should be surveyed prior to selecting a herbicide. The effectiveness of all herbicides declines over time, so multiple applications are recommended from spring to fall (Woodard 2008).

Extra care should be taken when using herbicides near desired wild vegetation or agricultural crops. Foliar herbicides applied with a wick will minimize damage to other nearby plants if *C. maculatum* is growing amongst favorable vegetation (Jeffery and Robinson 1990, OLA and MAFF 2002). Glyphosate and metsulfuron is not recommended for use near croplands (Monsanto Company 2007, WIDNR 2008). In a study conducted by Jeffery and Robinson (1990), hexazinon, metribuzin, and terbacil controlled Poison Hemlock with damaging alfalfa when applied while the alfalfa was still dormant. For more in-depth instructions on the use of 2,4-D, glyphosate, and/or metsulfuron, refer to the Pacific Northwest Weed Management Handbook (Prather et al. 2014).

***Echinochloa crus-galli* L. P. Beauvois**

Regulations (pertaining to the Great Lakes region)

In Minnesota, *E. crus-galli* is considered to pose a “minimal” threat to ecosystems: poses insignificant competition with native species, may naturalize, alters ecosystems insignificantly, and has little possibility of spread within or to other sites (Minnesota Invasive Species Advisory Council 2009).

In 2003, the Great Lakes Indian Fish & Wildlife Commission (GLIFWC) reported that *E. crus-galli* has widespread established populations, limited effective control methods, and causes low to moderate ecological impacts; as a result, GLIFWC does not require its regulation (Falck and Garske 2003).

Control

Biological

The fungal pathogen *Exserohilum monoceras* has shown some success in controlling Barnyard Grass (Catindig et al. 2009).

Physical

Echinochloa crus-galli seeds need to be near the surface to germinate; less than one cm of soil will inhibit germination (Adamus et al. 2001). Shallow tillage repeated during the spring can reduce emergence of Barnyard Grass (OLA and MAFF 2002). Mowing is unlikely to be effective because it will stimulate new growth from lateral buds (OLA and MAFF 2002).

Placing mulch over areas where *E. crus-galli* is expected to emerge will keep the soil cool and help suppress germination (Cornell University 2012). Further control methods may be needed.

Chemical

There is a high amount of genetic variation among *E. crus-galli* communities. Managers who choose a chemical control method may need to adjust the compound/application rate to fit the needs of the site (Altop and Mennan 2011). Mature plants show little sensitivity to herbicides applications. Herbicides applied pre-emergence or shortly after emergence typically exhibit the most effective control (Ahmadi et al. 1980, Maun and Barrett 1986, OLA and MAFF 2002).

Barnyard Grass is susceptible to sulfometuron methyl (Oust XP®, Spyder®), clethodim (Select MAX®, Intensity®), glyphosate (Accord®, Foresters' Glypro®, Roundup®, Cornerstone®, Razor®), imazapyr (Aresenal AC®, Habitat®, Chopper®), linuron (Linex 4L®, Loroz DF®), norflurazon (Predict®), sethoxydim (Sethoxydim E-Pro®, Poast Plus®). It is also susceptible to simazine (Simazine 4L®, Simazine 90 DF®), fluazifop (Fusilade DX®), hexainon (Velpar®), pendimethalin (Pendulum 3.3 EC®); however it should be noted that these herbicides cannot be used in Forest Sustainability Certified Areas (Keely and Thullen 1991, WIDNR 2011).

Ahmadi et al. (1980) found that when applied to Barnyard Grass five cm in height, glyphosate, terbuthryn, paraquat, atrazine and buthidazole all resulted in 100% control. The efficacy of herbicides containing glyphosate was increased when applied when soil was moist, because it allowed for better translocation through the entire plant (Ahmadi et al. 1980). Glyphosate plus 2,4-D will also control *E. crus-galli*; it is most effective when applied six days after the last irrigation or rainfall (Wicks and Hanson 1995).

Haloxifop, fluazifop, and sethoxydim offer effective control of (Balyan and Malik 1989). Seedling growth of *E. crus-galli* can be effectively reduced by applying fluazifop and haloxifop (separately) to the soil (Kells et al. 1986).

Ammonium salt of imazapic (Plateau®) is effective at controlling Barnyard Grass (BASF Corporation 2011).

Diclofop was effective at controlling Barnyard Grass, especially when applied postemergence to plants with less than four leaves (West et al. 1980).

2,4-D sodium (Hormicide®) applied pre-emergence will help prevent barnyard from growing. Young plants can be controlled with paraquat, where as more mature plants can be controlled with 2,2-DPA. In Australia *E. crus-galli* plants treated with F-34 (3,4 - dichloropropionanilide) 2-3 weeks post-emergence were successfully controlled (FAO 2012).

Bispyribac is a postemergent herbicide that is registered for grasses in rice fields and offers effective control of *E. crus-galli*. Its efficacy may be increased when used in combination with a spray adjuvant and/or urea ammonium nitrate (Koger et al. 2007).

Postemergent treatment with propanil and pendimethalin offers good control of Barnyard Grass (Setre Chemical Company 1986). Propanil is most effective against Barnyard Grass applied before the plants have three leaves (Snipes and Street 1987). However, Barnyard Grass can become resistant to propanil and resistant communities have been reported in Arkansas (Gealy et al. 2003).

DPX-79406 (1:1 premix of nicosulfuron and rimsulfuron) and rimsulfuron are registered for use in Ontario and offer good control of Barnyard Grass growing intermingled with corn (Bosnic and Swanton 1997). When applied post-emergence, Rimsulfuron plus thifensulfuron resulted in 97% control (Krausz et al. 2000).

Cyhalofop-butyl offers good control of *E. crus-galli* when applied early postemergence. When applied to Barnyard Grass growing within rice, this compound had only slight effects on rice quantity and quality (Ntanos et al. 2000). The ethyl ester of fenoxaprop will control *E. crus-galli* and most varieties of rice are tolerant (Snipes and Street 1987).

For more specific information on chemical control methods, please visit:

[Cornell University's Pesticide Management Education Program](#), [Pacific Northwest Weed Management Handbook](#), and [The Rice Knowledge Bank](#).

Other

Ongoing research indicates that *E. crus-galli* may be susceptible to various natural, biodegradable herbicides derived from microorganisms and from other plant species (Malik 1997, Khanh et al. 2006, Kato-Nogucki et al. 2012, Li et al. 2012a).

***Epilobium hirsutum* L.**

Regulations (pertaining to the Great Lakes region)

Great Hairy Willow Herb is ranked as having “moderate environmental invasiveness” by the New York State Office of Invasive Species and is considered “well-established” by the Ohio Department of Natural Resources (Ohio Division of Natural Areas and Preserves and Nature

Conservancy 2000, NYISC 2010). The transportation, translocation, or introduction of *E. hirsutum* is prohibited in Wisconsin, except in Kenosha County (Bureau of Plant Industry 2012). *Epilobium hirsutum* is listed as an introduced species in Ontario (Canadensis 2012). Regionally, the Great Lakes Indian Fish & Wildlife Commission (GLIFWC) classifies this species as capable of causing moderate to severe ecological impacts and/or having limited effective control options available.

Control

Given the invasive nature of *E. hirsutum*, control methods need to be applied and monitored for several years to be effective (King County 2008). For localized infestations, the GLIFWC recommends *E. hirsutum* “be controlled immediately upon detection before it becomes established and spreads” (Falck and Garske 2003).

Biological

Elephant moth (*Deilephila elpenor*) feeds on *Epilobium*, but is not a native to the Great Lakes (Hoskins 2012, Pittaway 2012). Genetic material extracted from *E. hirsutum* individuals displaying phyllody of flowers and/or plant yellowing revealed infection by *Epilobium* phyllody (EpPh) phytoplasma, an obligate, parasitic bacteria that attach to phloem tissue (Alm et al. 2002). The ability of this phytoplasma to act as a biocontrol agent is still unknown. Additional insects that feed on and use *E. hirsutum* for part of their life cycle are listed on the [website of J. Lindsey](#).

Physical

Small populations of *E. hirsutum* can be hand dug, placed into plastic bags, and disposed of in the trash (King County 2008). When hand digging, one should be sure to remove as much of the root pieces as possible because rhizomes left in the soil can generate new plants (King County 2008, Campbell et al. 2010). Mowing or cutting of mature plants will not kill the plant, but flowering stems can be cut in late summer or early fall to prevent seed production and dispersal (King County 2008).

After 18 weeks in water logged and flooded conditions, Lenseen et al. (2000) found *E. hirsutum* populations only achieved 82% and 54%, respectively, of the mean biomass growth as populations in drained conditions. Furthermore, it was determined that flooded individuals experienced reduced rhizomal growth in terms of numbers, size, and biomass (Lenseen et al. 2000). Growth of water logged plants was further limited by pruning adventitious roots. This procedure reduced the depth of the plant’s primary root system and made individuals more susceptible to uprooting by various environmental conditions (flooding, wind, etc.) (Etherington 1984). Both of these experimental insights suggest that combined water level manipulation and root pruning may be beneficial to the control of *E. hirsutum*.

Due to the regenerative nature of rhizomes, composting plant material off-site is not recommended (King County 2008).

Chemical

Epilobium hirsutum populations treated with Patron 170 are typically susceptible to severe injury or death. It should be noted that this pesticide is currently prohibited by from use on Forest

Stewardship Council (FSC) land (WIDNR 2011). Under dry conditions, 2,4-D will control great hairy willow-herb (Evans et al. 2003). In moist or aquatic locations, glyphosate (Rodeo®) will stress or kill above-ground portions of the plant, but the root system will remain intact and plants will recover (Evans et al. 2003).

***Frangula alnus* P. Mill.**

Regulations (pertaining to the Great Lakes region)

The New York Invasive Species Council assessed *F. alnus* as having a high risk of causing ecological harm and recommended that its use be prohibited (NYISC 2010). This species is restricted in Wisconsin; it may not be transported, transferred, or introduced into any ecosystem (Bureau of Plant Industry 2012). *Frangula alnus* is considered an exotic weed by the Illinois Department of Natural Resources; sale of this species within the state is not allowed (Bureau of Environmental Programs 2009). It is listed as a restricted noxious weed in Minnesota and the importation, sale, or transport this plant is illegal (MNDNR 2009).

This species is not widespread in the ceded territories governed by the Great Lakes Indian Fish & Wildlife Commission (GLIFWC). The GLIFWC recommends that Glossy Buckthorn be controlled immediately upon being found. In areas where *F. alnus* is already present, the GLIFWC categorizes it as capable of having severe ecological impacts and recommends that small, peripheral populations be controlled upon detection and center populations be monitored (Falck and Garske 2003).

Control

When treating a large infestation and/or working with limited resources, priority should be given to the largest trees bearing blooms or fruits (Thompson and Luthin 2004). It is important to have a disposal method in place for the portions of the Glossy Buckthorn that contain fruit. Stems and branches with berries can be destroyed by burning; those without fruit can be left on site to decompose (USDA NRCS 2007). If burning is not an option, fruit should be disposed of off-site (PADCNr n.d.).

Biological

Currently, there are no specific biological control agents for this species, but research on more generalized herbivores is ongoing (Chandler et al. 2010).

Physical

Cutting alone will not control this species because it will resprout, regardless of what time of year it was cut (Brock 2012). When using a physical control method, effort should be made to limit soil disturbance so as not to cause erosion (Larson 2009).

Individual plants less than 0.5 inch in diameter can be removed by hand. Removing manually is easiest when the soil is sandy or is moist (MNFI 2012). Care must be taken to disturb the soil as little as possible (Buenzow 2010). Plants that are 0.5 inch to 1.5 inch in diameter can be physically removed with a mechanical device- care should be taken to disturb as little of the soil as possible (Buenzow 2010).

Glossy Buckthorn can also be controlled through girdling. In this method, cuts are made to the trunk or main stem just above the base, the bark is removed (including the green, cambium layer

beneath the bark) (USDA NRCS 2007). The cut should be large enough, about an inch long, to prevent the tree from healing. The Illinois Natural History Survey recommends making two parallel cut 4-5 inches apart when girdling (Heidorn 2011). This method is most effective when in the summer after the leaves have fully developed or after the leaves have dropped off in the early winter (USDA NRCS 2007). This method is less effective on plants that have many main trunks/main stems.

If Glossy Buckthorn is growing in a grassland or savanna ecosystem, controlled burns may offer long-term control. This method needs to be repeated ever 2-3 years (MNDNR 2009). Burns are most effective from April through June and from September through November (Hanson et al. 2012).

Repeated mowing in open areas has been reported to be effective in prevented seedling establishment (Ohio EPA 2001). This method is most effective for plants less than two years old. Mowing in early spring and again in fall will help deplete the energy reserves in the root system, deplete the seed bank, and will not interfere with any birds that may use Glossy Buckthorn for nesting (USDA NRCS 2007).

If Glossy Buckthorn is present in a managed wetland with a lowered water level, returning the level to its original depth may flood and kill the plants. The impact a changed water level will have on the whole ecosystem should be determined in advance (Roman 2007).

Chemical

Adding dye to herbicides prior to application, will help distinguishes between plants that have and have not been treated (MNFI 2012).

One method is to spray foliage with herbicides such as glyphosate (Accord®, Foresters' Glypro®, Roundup®, Cornerstone®, Razor®), triclopyr (Garlon 3A®, Garlon 4®, Tahoe 4E®), fluazifop (Fusilade II®), imazapyr, metsulfuron-methyl, 2,4-D, or picloaram to control Glossy Buckthorn (WIDNR 2011, Hanson et al. 2012). Glyphosate will kill any vegetation it comes in contact with and triclopyr will kill broadleaf plants, but will not harm grass if applied properly (MNDNR 2009). The best time to use the foliage spray method is between May and November (Hanson et al. 2012).

For plants with stems less than six inches in diameter, basal steam treatment, in which an oil-based herbicide is applied directly the bark from the root collar up 12-18 inches, can be used without having to cut down the plant (MNDNR 2009, Buenzow 2010). Glyphosate and triclopyr can also be used for this technique (Hanson et al. 2012). Herbicide can be applied with a low-pressure backpack sprayer. Herbicide can be applied any time of year, providing there is access to the ground line; although late fall and winter are preferred because plants are dormant. Glossy Buckthorn may leaf out in the spring after a fall or winter herbicide application, but the leaves should senesce as the chemicals are translocated throughout the plant (Buenzow 2010). Basal spraying is the most cost-effective method of controlling populations of Glossy Buckthorn (Thompson and Luthin 2004).

Both the ester and amine formulations of triclopyr can be used to treat Glossy Buckthorn; the amine form is safe for use in most wetlands (Larson 2009, MNFI 2012).

Other

Larger trees can be cut near soil level in late summer or early fall. The stumps should then be treated within two hours of being cut with herbicides containing triclopyr. Only the cut surface needs to be treated when using a water-soluble herbicide, but when using an oil-based herbicide, treat the cut surface and the remaining bark (MNDNR 2009).

Glyceria maxima

Regulations (pertaining to the Great Lakes region)

Glyceria maxima poses a high ecological threat to ecosystems; therefore, the New York Invasive Species Council recommends that this species be prohibited (NYISC 2010). The New Invaders Watch Program lists *G. maxima* on its “watch list” for Illinois (Maurer 2009). *G. maxima* is prohibited from transport, transfer, or introduction in Wisconsin; however, there are exceptions made for several counties (Bureau of Plant Industry 2012).

The Great Lakes Indian Fish & Wildlife Commission (GLIFWC) has not detected *G. maxima* within its territories. To keep it from invading, the GLIFWC recommends controlling any individuals of this species immediately (Falck and Garske 2003).

Control

G. maxima is a perennial species; therefore, populations may require treatment for two to three years for complete control (USACE 2012b).

Biological

There are no known biological control methods for this species.

Physical

Small infestations of *G. maxima* can be dug up; care should be taken to remove all parts of the roots and rhizomes (Forest Health Staff 2006e). Subsequent removal of seedlings germinated from the seed bank or missed rhizomes pieces may be necessary (King County 2012). Small, dense communities of Reed Mannagrass can also be controlled by being covered with black plastic for five or six weeks during the growing season (Forest Health Staff 2006e).

The vegetative spread of larger populations can be controlled by repeated mowing, cutting, harvesting, roto-tilling, or rotovating (Sundblad and Robertson 1988, USACE 2012b). Where applicable, these treatment methods can be supplemented with artificially created flood conditions (Hroudová and Zákrevský 1999). Mowing or cutting two to three times a summer may deplete the energy reserves in the roots and rhizomes. This may reduce *G. maxima*'s ability to compete and allow other vegetation to expand into the site (King County 2012).

Chemical

A foliar spray of glyphosate (3% solution) applied early to late summer will control populations of *G. maxima* (King County 2012, USACE 2012b). Rhizomes may survive after initial spraying

(USACE 2012b). Braverman (1996) found that glyphosate at two kg ai/ha and dalapon (2,2 dichloropropanoic acid) at 10 kg ai/ha controlled *G. maxima*. Imazapyr is most effective on Reed Mannagrass when applied in summer or early fall and when water levels are low and plant stems are not submerged (King County 2012, USACE 2012b).

In floating fens in the Netherlands, sulfate was experimentally added to the soil. This caused the free sulfide concentration to increase and resulted in a decrease in the growth of *G. maxima* (Loeb et al. 2007).

Other

For large populations, herbicide treatment will be an effective option. If the decaying plant material falls into a nearby body of water and decomposes, the dissolved oxygen levels could decrease. To avoid this, dead plant material should be removed two to four weeks after herbicides have been applied (King County 2012).

***Hydrocharis morsus-ranae* L.**

Regulations (pertaining to the Great Lakes region)

Prohibited in Illinois, Michigan, Minnesota, and Wisconsin (GLPANS 2008). In Minnesota, possession, import, purchase, transport, or introduction of *H. morsus-ranae* will result in a misdemeanor (MN DNR 2013b).

The New York Invasive Species Council ranks this species as posing a very high ecological threat and recommends that it be regulated (NYISC 2010).

The Great Lakes Indian Fish & Wildlife Commission have not found *H. morsus-ranae* in their ceded territories, but recommend immediate control upon detection (Falck and Garske 2003).

Control

Biological

Grass Carp, *Ctenopharyngodon idella*, feeds on *H. morsus-ranae*. However, this introduction of this species may also have a negative effect on native vegetation, which might outweigh the benefits of *H. morsus-ranae* control (Mikulyuk and Nault 2011).

Physical

Mowing does not control *H. morsus-ranae* population (Sager and Clerc 2006). Removing manual harvesting may provide temporary control (IL DNR 2009a, WIDNR 2012c). To improve efficacy of this method, harvesting should occur in the spring; after the turions have begun growing, but before dense mats form (Catling et al. 2003).

Another possible method for small water bodies would be to have a water draw-down after turions have germinated, but before extensive growth occurs (Catling et al. 2003).

Chemical

Diquat, imazapyr, penoxsulam, and imazamox offer excellent control of *H. morsus-ranae* (AERF 2013).

Impatiens glandulifera

Regulations (pertaining to the Great Lakes region)

Impatiens glandulifera is ranked as having “low environmental invasiveness” by the New York State Office of Invasive Species and is therefore unregulated in that state. Regionally, the Great Lakes Indian Fish & Wildlife Commission (GLIFWC) classify this species as capable of causing moderate to severe ecological impacts and/or having limited effective control options available (Falck and Garske 2003).

Control

Regardless of method, all control methods should be adaptive and involving monitoring of the site for several years to ensure plants do not germinate from the seed bank (King County 2010a).

It should also be noted that in experiments conducted by Wadsworth et al. (2000), *I. glandulifera* was so prolific that even scenarios with 99% control efficiency were as ineffective as scenarios with no management action.

Biological

To date, no specific biological control methods are available for *I. glandulifera* (Sheppard et al. 2006). However, allowing cattle or sheep access to areas infested with *I. glandulifera* will control the population and the spread of the species either by direct grazing or by trampling of young seedlings (CEH 2004b).

The species *Aphis fabae*, *Impatiens balsamines*, and *Deilephila elpenor* are known to feed on Ornamental Jewelweed, but their capacity to act as biological control agents is still unknown (Beerling and Perrins 1993). Although an initial experiment by Tanner (2011) indicated that *Deilephila elpenor* exhibited lower biomass and survivorship when raised on *I. glandulifera*.

In its native range, *I. glandulifera* has been known to harbor *Puccinia komarovii* (a rust pathogen) which is currently undergoing research as a control agent (Tanner 2011).

Physical

A good control method for small infestations is the removal of Ornamental Jewelweed by pulling or digging. Efforts should be concentrated prior to seed-set in the spring, while the soil is still moist (King County 2010a).

Hartmann et al. (1995) found that mowing, mulching or soil cultivation were successful in controlling *I. glandulifera* populations in Germany. Mowing infestations of *I. glandulifera* also causes less soil erosion than hand-pulling or digging (King County 2010a). Ornamental Jewelweed should be cut close to the ground, preferable below the lowest node to prevent regrowth (CEH 2004b). If the vegetative parts are to be left on-site to decompose, plant material should be allowed to dry out completely or the stems should be crushed (by walking or jumping on them) to prevent regrowth. Flower heads and seed capsules should not be left on site (King County 2010a).

Chemical

Herbicides are more effective on populations of *I. glandulifera* that are large or inaccessible to other equipment (King County 2007a). Application of any herbicide needs to be carefully timed. Seedlings need to be large enough that they will be covered by the herbicide, but before flowers are produced (King County 2010a). Glyphosate (Round-up® or Aquamaster™) is most effective if applied to growing leaves. Care should be taken not to get the herbicide on desirable plants because it is non-selective and will damage any foliage it comes in contact with (King County 2007a). Herbicides containing triclopyr (Renovate3), 2,4-D, or metsulfuron are more selective and will not harm most grass species (King County 2007a). These herbicides may be preferable if *I. glandulifera* infestations are in mixed communities and/or near water bodies (Centre for Plant Aquatic Management 2004).

When using herbicides, do not cut or mow treated plants until they have died completely, which may take two weeks (King County 2007a).

Iris pseudacorus

Regulations (pertaining to the Great Lakes region)

The New York Invasive Species Council ranked *Iris pseudacorus* as posing a high ecological risk and recommends that it be prohibited (NYISC 2010). The Ohio Department of Natural Resources lists Yellow Iris as a “well-established invasive” (Ohio Division of Natural Areas and Preserves and Nature Conservancy 2000). Under the [Michigan Public Acts 74-80 of 2005](#), *I. pseudacorus* is a prohibited aquatic plant species: a person cannot have any purebred or hybrid variant of this species, or fragments or seed unless they are being collected for identification, and/or the person is in the process of legally removing/eradicating the species (Latimore et al. 2011). In Illinois, *I. pseudacorus* cannot be possessed, propagated, bought, sold, bartered, transported, transferred, or loaned with a permit (GLPANS 2008). *I. pseudacorus* is established and considered a “moderate threat” to local ecosystems in Minnesota. This has led it to be classified as a restricted species that cannot be planted/released with a permit (GLPANS 2008, Minnesota Invasive Species Advisory Council 2009).

In 2001, the Great Lakes Indian Fish & Wildlife Commission (GLIFWC) considered *I. pseudacorus* capable of severe ecological impacts even though it tends to occur in a few small populations and has a wide array of control options (Falck and Garske 2003). Yellow Iris was still listed as a “high priority” invasive species in 2009, 2010, and 2011 (Falck et al. 2010, 2011, and 2012).

Control

For large infestations, it is best to start in the areas with the lowest concentration of Yellow Irises and progressing towards high density areas (i.e. periphery toward the center, upstream toward downward) (King County 2009).

The glycosides in *I. pseudacorus* leaves and rhizomes cause skin irritation, so care should be taken to protect skin from contact regardless of the control method (Cooper and Johnson 1984, Forest Health Staff 2006f, Nature Conservancy n.d.).

Biological

Insects and animals do not exert grazing pressure on *I. pseudacorus* in its native range (Forest Health Staff 2006f). Although numerous pathogens and insects attack Yellow Iris, there are currently no known biological control agents (Tu 2003).

Physical

Small infestations of *Iris pseudacorus* seedlings can be easily pulled or dug up by hand, especially in damp or wet soil (King County 2009). This method is also feasible for small stands of mature plants; however, tools (pickaxes, saws, etc.) may be needed to remove the rhizomes (King County 2009). Care should be taken to remove all parts of the rhizomes to prevent resprouting (Forest Health Staff 2006f). For populations of Yellow Iris growing in standing water, removal of the leaves and stems above water before flowering can result in good control and reduced spread (Simon 2008 in Noxious Weed Control Program 2009). Sites should be monitored for the emergence of new plants from the seed bank or from rhizome sections that were not removed after control measures are completed (King County 2009). This control method may need to be repeated for three or four years to be effective.

Repeat mowing or removal of seed pods can control the spread of larger infestations (Forest Health Staff 2006f). Repeated removal of the aboveground portions of Yellow Iris may also deplete the plant's energy reserves and may eventually kill it (Tu 2003).

Mechanical removal in sensitive areas, such as shallow streambeds, can be expected to cause extensive disturbance to the substrate and permit the establishment of other unwanted plants (Jacono 2001). Plant material should not be composted on-site because rhizomes can continue to grow for up to three months without water (Sutherland 1990).

Burning is not recommended for control because of this plant's strong tendency to resprout from rhizomes (Clark et al. 1998, Sutherland 1990).

Chemical

Applications with herbicides such as glyphosate (Rodeo™ or Aquamaster™) or imazapyr (Habitat™) can provide control of larger infestations (Forest Health Staff 2006f, King County 2009, MN DNR 2012a). If Yellow Iris is mixed with desirable plant species, targeted control can involve cutting the stems of *I. pseudacorus* and applying the herbicide directly to the cut area (IISCTC 2007). Areas should not be mowed for several weeks after herbicide application to allow the treatment to be effective (King County 2009).

Other

Cutting followed by herbicide (glyphosate) treatment with a dripless wick may be the best method for controlling plants in sensitive sites.

***Juncus compressus* Jacq.**

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

Biological

Aphids may occasionally feed on *Juncus* spp., but most rushes are fairly resilient to extensive damage from insect or diseases (Stevens and Hoag 2003). Cattle, horses, and sheep graze on *Juncus* spp., but the extent of control gained from grazing is unknown (CEH 2004a, Cosyns et al. 2005).

Physical

There are no known physical control methods for this species.

The rhizome matrix of *Juncus* spp. enables it to withstand periods of drought and flooding; water level fluctuation is not recommended as a physical control method (Stevens and Hoag 2003).

Chemical

Sethoxydim (Vantage®) will target most grass species and should not affect nearby broadleaf herbs, sedges, or woody plants (Brock 2012). Glyphosate and ammonium salt of imazapyr (Plateau®) will control *J. compressus*, but are non-selective and should be applied carefully (CEH 2004a) Glyphosate should be sprayed directly onto foliage in mid-late summer (CEH 2004a).

***Juncus gerardii* Loisel.**

Regulations (*pertaining to the Great Lakes region*)

There are no known regulations for this species.

Control

Juncus gerardii seedlings that emerge in late winter or early spring are taller and fertile and can be distinguished from the seedlings that emerge at the end of spring or beginning of summer and are shorter and infertile (Bouzillé et al. 1997).

Biological

Aphids may occasionally feed on *Juncus* spp., but most rushes are fairly resilient to extensive damage from insect or diseases (Stevens and Hoag 2003). Cattle, horses, and sheep graze on *Juncus* spp., but the extent of control gained from grazing is unknown (CEH 2004a, Cosyns et al. 2005).

Physical

There are no known physical control methods for this species.

The rhizome matrix of *Juncus* spp. enables it to withstand periods of drought and flooding; water level fluctuation is not recommended as a physical control method (Stevens and Hoag 2003).

Efforts made to graze an area with *J. gerardii* may increase the size of its seed bank (Jutila 1998, Jutila 1999).

Chemical

Sethoxydim (Vantage®) will target most grass species and should not affect nearby broadleaf herbs, sedges, or woody plants (Brock 2012). Glyphosate and ammonium salt of imazapyr (Plateau®) will control *J. gerardii*, but are non-selective and should be applied carefully (CEH 2004a). Glyphosate should be sprayed directly onto foliage in mid-late summer (CEH 2004a).

Other

Charpentier et al. (1998) found that a water depth of 10 cm and salinity level of 2 g/L NaCl caused *J. gerardii* to stop shoot and root growth and decrease seed production. A constant water level and a low salinity level may limit the growth and expansion of *J. gerardii* populations (Charpentier et al. 1998).

***Juncus inflexus* L.**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

Aphids may occasionally feed on *Juncus* spp., but most rushes are fairly resilient to extensive damage from insect or diseases (Stevens and Hoag 2003). Cattle, horses, and sheep graze on *Juncus* spp., but the extent of control gained from grazing is unknown (Centre for Aquatic Plant Management 2004a, Cosyns et al. 2005).

Physical

Juncus inflexus will regrow if it is cut or mowed; therefore, these methods will only provide control if done repeatedly (Missouri Botanical Garden 2012).

The rhizome matrix of *Juncus* spp. enables them to withstand periods of drought and flooding; water level fluctuation is not recommended as a physical control method (Stevens and Hoag 2003).

Chemical

Sethoxydim (Vantage®) will target most grass species and should not affect nearby broadleaf herbs, sedges, or woody plants (Brock 2012). Glyphosate and ammonium salt of imazapyr (Plateau®) will control *J. inflexus*, but are non-selective and should be applied carefully (CEH 2004a). Glyphosate should be sprayed directly onto foliage in mid-late summer (CEH 2004a).

Lupinus polyphyllus

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Lupines usually require ecological disturbance to persist. Control is generally unnecessary in undisturbed sites.

Biological

There are no known biological control methods for this species. Lupines may be toxic and populations often increase in grazed (pasture) systems. Several native insects feed on Lupines, but are considered insufficient for control (DiTomaso 2013).

Physical

Hand pulling, tillage, and digging are effective for controlling established plants, but the disturbance from these methods can promote new recruitment. The root system should be severed below the thickened crown. Mowing is not effective unless done frequently enough to prevent seed production. Fire is not an effective control as this promotes germination (DiTomaso 2013).

Chemical

Several alternatives are available for chemical control – most are most effective when applied post-emergence and before flowering. 2,4-D and/or dicamba can be applied at temperatures less than 26.6°C. Glyphosate is effective for spot treatment where reseeding (with natives) is planned as it will not injure seedlings. Chlorsulfuron and metsulfuron are also effective (DiTomaso 2013).

Lycopus asper

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods specific to this species.

***Lycopus europaeus* L.**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods specific to this species.

Lysimachia nummularia

Regulations (pertaining to the Great Lakes region)

New York Invasive Species Council determined that this species has a very high ecological impact and recommends that this species be prohibited within the state (NYISC 2010).

Control

Biological

There are no known biological control methods for this species.

Physical

Small populations can be pulled up by hand; all plant fragments should be removed to prevent resprouting (Kennay and Fell 2011, MISIN and MNFI 2013d, PADCNr n.d.).

A prescribed burning in the spring, when *L. nummularia* is green and native plants are still dormant, can be an effective means of control (Kennay and Fell 2011). This control method may need to be repeated for several years for complete control (Tu et al. 2001).

Given the growth structure of Moneywort, mowing will not control this species (Kennay and Fell 2011, PADCNr n.d.). Planting native grasses after any physical control method could shade out any potential regrowth from Moneywort (MISIN and MNFI 2013d, PADCNr n.d.).

Chemical

Rodeo® or Rodeo® will be effective at controlling *L. nummularia* (Kennay and Fell 2011, PADCNr n.d.)

Lysimachia vulgaris

Regulations (pertaining to the Great Lakes region)

The New York Invasive Species Council determined that this species poses a high ecological threat and recommends that it be prohibited within the state (NYISC 2010).

The Great Lakes Indian Fish & Wildlife Commission considers this species capable of causing severe ecological impacts and recommends it be controlled within their ceded territories (Falck and Garske 2003).

Control

Biological

There are no known biological control agents for this species (King County 2010b).

Physical

Hand-pulling is effective for small infestations and/or for young plants (King County 2010b, MISIN and MNFI 2013c). Care should be taken to dig out and remove all plant fragments, especially the rhizomes, to prevent regrowth (King County 2010b). All seed heads and rhizomes pieces should be disposed of in plastic bags and removed from the site (King County 2010b).

Repeated mowing of *Lysimachia vulgaris* may contain the existing population, but it will not eradicate it (King County 2010b).

Chemical

To control larger infestations, treatment with herbicides containing glyphosate (Rodeo®, AquaMaster®), imazapyr (Habitat®), or triclopyr (Garlon 3A®, Renovate 3®) may be necessary (King County 2010b). It is important to note that glyphosate and imazapyr are non-selective and will harm any plant it comes in contact with (King County 2010b). Triclopyr will not harm grasses, sedges, or cattails, and may be more appropriate to use in diverse plant communities (King County 2010b). Physical control methods should not be used on populations treated with herbicides until several weeks after application (King County 2010b).

Lythrum salicaria

Regulations (pertaining to the Great Lakes region)

Lythrum salicaria is listed as an exotic weed in Illinois ([525 Illinois Compiled Statutes § 10/3 and 10/4](#)) making it illegal to buy, sell or distribute plants, its seeds, or any part without a permit. Planting, sale, or other distribution without a permit is also prohibited in Indiana ([312 IN Administrative Code § 14-24-12](#)). Purple Loosestrife – including all cultivars – is a prohibited invasive species in Minnesota ([MN Administrative Rules § 6216.0250](#)). The species is restricted in Michigan, with an exemption for sterile cultivars ([MI NREPA 451 § Section 324.41301](#)). Planting or sale of the species without a permit is prohibited in Ohio ([Ohio Revised Code § 927.682](#)), though the director may exempt varieties ‘demonstrated not to be a threat to the environment’. Pennsylvania has designated all nonnative *Lythrum* species and their cultivars as noxious weeds ([7 PA Code § 110.1](#)). Purple Loosestrife is designated both as a restricted species ([Wisconsin Statutes NR § 40.05](#)) and as an invasive aquatic plant ([Wisconsin NR 109.07 \(2\)](#)) in Wisconsin.

Control

Biological

Biological control agents do not eliminate the target weed, but when successful, can suppress weed populations to a nonsignificant level (Rees et al. 1996). Five species of beetles have been approved for the biocontrol of *L. salicaria* (Blossey et al. 1994ab).

Galerucella californiensis and *G. pusilla* are both leaf-feeding chrysomelids. These beetles defoliate and attack the terminal bud area, drastically reducing seed production. The mortality rate to Purple Loosestrife seedlings is high. Evidence of *Galerucella* spp. damage are round holes in the leaves. Four to six eggs are laid on the stems, axils, or leaf underside. The larvae feed constantly on the leaf underside, leaving only the thin cuticle layer on the top of the leaf. Initial introductions in eastern North America occurred in Virginia, Maryland, Pennsylvania,

New York, Minnesota, and southern Ontario in August 1992 (Hight et al. 1995). In 1992, these three beetles were released in Washington. *Galerucella* spp. populations visibly impacted Purple Loosestrife stands by 1996 (Washington State Department of Ecology 2012). In the Great Lakes region, Sea Grant conducted an extensive, multi-state program involving youth in raising and releasing *Galerucella* beetles for control of Purple Loosestrife (Michigan Sea Grant 2001).

Hylobius transversovittatus is a root-mining weevil that also eats leaves. This beetle eats from the leaf margins, working inward. The female crawls to the lower 2-3 inches of the stem then bores a hole to the pithy area of the stem where 1-3 eggs are laid daily from July to September. Or, the female will dig through the soil to the root, and lay eggs in the soil near the root. The larvae then work their way to the root. *H. transversovittatus* damage is done when xylem and phloem tissue are severed, and the carbohydrate reserves in the root are depleted. Plant size is greatly reduced because of these depleted energy reserves in the root. The larvae evidence is the zig-zag patterns in the root.

Nanophyes marmoratus and *N. brevis* are seed eating beetles. Young adults feed on new leaves on shoot tips, later feeding on the flowers and closed flower buds. Sixty to one hundred eggs are laid in the immature flower bud. Seed production is reduced by 60%. There were two test sites releases in 1996. Approval to introduce *N. marmoratus* was granted followed by introductions in New York and Minnesota in 1994. Additional releases occurred in New Jersey in 1996. *N. marmoratus* has also been released in Ohio (Ohio EPA 2001). Release of *N. brevis* planned for 1994 was delayed due to contamination of the original shipment with a parasitic nematode (Piper, 1997). This infection appeared benign for *N. brevis*; however, due to the potential for non-target effects of the nematode after introduction into North America, only disease free specimens should be introduced, which, at present, effectively precludes the introduction of *N. brevis* (Blossey 2002a).

Bayeriola salicariae, a gall midge, was studied and screened between 1990 and 1992 (Blossey and Schroeder 1995). Based on results indicating a potential wider host range, the gall midge *B. salicariae* was not proposed for introduction (Blossey and Schroeder 1995).

Targetted grazing by sheep has also been used as a biocontrol (Kleppel and LaBarge 2011).

Revegetation of disturbed riparian sites can be used to prevent Purple Loosestrife establishment and to reduce re-establishment after control procedures are applied. Fowl Mannagrass (*Glyceria striata*), Foxtail Sedge (*Carex alopecoidea*), and Reed Canarygrass (*Phalaris arundinacea*) have achieved dominance and prevented re-invasion in plots where Purple Loosestrife was experimentally removed. Smartweed (*Polygonum lapathifolium*) is reported to out-compete Purple Loosestrife during its first year of growth. Seeding Japanese Millet (*Echinochloa frumentacea*, also called billion-dollar grass) at 30 pounds/acre on exposed moist soil after drawdown and before Purple Loosestrife seedlings began to grow provided control. Japanese Millet is considered an exceptional wildlife plant (Jacobs 2008).

Physical

Most mechanical and cultural attempts to control Purple Loosestrife are ineffective. A single known exception is cutting followed by flooding.

For small infestations and isolated plants, hand pulling may be effective. Pull individual loosestrife plants by hand before seed is set. The entire root system must be removed, but do not dig out roots because soil disturbance may release seeds buried in the soil and break off plant parts, which then reproduce. Instead, a cultivator may be used to tease roots from the soil. All plant parts should be bagged to prevent dispersal or resprouting and preferably burned. Follow-up treatments are recommended for at least three years.

Frequent cutting of the stems at ground level is effective but must be continued for several years (Courtney 1997). Mowing is generally not effective as it exposes the seed bank.

Flooding is generally ineffective at controlling Purple Loosestrife, though some success has been reported for control of seedlings when using flooding regimes in excess 30 cm for over seven weeks (Balogh 1986).

Prescribed burning is not an effective management tool for Purple Loosestrife. The dead upright stems do not carry fire well and the fine fuels are often lacking. The growing points of the root crown are about two cm (0.8 inch) below the soil surface, so surface fires are not likely to inflict much damage. Purple Loosestrife begins spring growth about a week or 10 days after broadleaved cattails, so a fire of sufficient intensity to damage Purple Loosestrife could also damage desirable native species (IL DNR 2009a).

Chemical

Only herbicides permitted for wetland use may be used to control Purple Loosestrife. There are four chemicals that can be used to manage Purple Loosestrife on sites with standing or moving water typical of where it invades. Triclopyr and glyphosate are used most commonly. However, 2,4-D, and imazapyr are also formulated for aquatic applications. Widespread elimination of standing biomass may result in the exposure and sprouting of the immense Purple Loosestrife seed bank. Thus, broadleaf-specific herbicides which do not harm monocot species (such as common wetland grasses and sedges) are preferred. The most species specific way to apply herbicide is by cutting and treating the stems. Foliar spray can be used by applying herbicide after the period of peak bloom, in late August. Any control method should be followed up on a yearly basis to catch any missed plants or new sprouts (Ohio EPA 2001)

Marsilea quadrifolia

Regulations (pertaining to the Great Lakes region)

Marsilea quadrifolia is a prohibited plant species in Illinois (GLPANS 2008).

The Great Lakes Indian Fish & Wildlife Commission have not found *M. quadrifolia* within their ceded territories, but recommend that it should be controlled immediately if found (Falck and Garske 2003).

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

In studies conducted in Japan, *M. quadrifolia* was susceptible to the herbicide bensulfuron methyl (Aida et al. 2004, Luo and Ikeda 2007).

***Mentha aquatica* L.**

Regulations (pertaining to the Great Lakes region)

The Great Lakes Indian Fish & Wildlife Commission considers this species capable of causing severe ecological impacts and recommends it be controlled within their ceded territories (Falck and Garske 2003).

Control

Biological

Chrysolina herbacea feeds on *M. aquatic*, despite the deterrents this species produces to minimize damage caused by herbivores (Atsbaha Zebelo et al. 2011).

Physical

Hand-pulling may control small populations of *Mentha* spp. (MISIN and MNFI 2013d).

Chemical

General herbicides, such as glyphosate, are effective at controlling *Mentha* spp. (MISIN and MNFI 2013d).

***Mentha gracilis* Sole (pro sp.)**

Regulations (pertaining to the Great Lakes region)

In 2011, the Great Lakes Indian Fish & Wildlife Commission ranked *Mentha gracilis* to be a lower priority for regulation and management (Falck et al. 2012).

Control

Biological

Production of Gingermint can be significantly reduced by diseases such as rhizome rot (*Rhizoctonia solani* Kuhn.) (Skotland 1979, Skotland and Traquair 1982), septoria leaf spot (*Septoria menthae* Oudem.) (Green 1961) and verticillium wilt (*Verticillium dahliae* Kleb.), and insects such as mint flea beetle (*Longitarsus waterhousei* Kutschera) and mint bud mite (*Tarsonemus* spp.). Further, *Mentha gracilis* is susceptible to both races of mint rusts that affect its parent mint species (Johnson et al. 1999). However, none of these have not been evaluated for biocontrol of invasive populations (Poovaiah et al. 2006).

Physical

Hand-pulling may control small populations of *Mentha* spp. (MISIN and MNFI 2013d).

Chemical

General herbicides, such as glyphosate, are effective at controlling *Mentha* spp. (MISIN and MNFI 2013d).

Mentha spicata

Regulations (pertaining to the Great Lakes region)

In 2011, the Great Lakes Indian Fish & Wildlife Commission ranked *M. spicata* to be a lower priority for regulation and management (Falck et al. 2012).

Control

Biological

Several diseases of mints are well characterized as crop pests – e.g., Mint Rusts – and activity of these pests appears to be quite host specific (Johnson 2013) but the potential for use of mint diseases for biocontrol has not been the subject of direct research.

Physical

Hand-pulling may control small populations of *Mentha* spp. (MISIN and MNFI 2013d).

Soil barriers may be used to restrain rhizomatous spread if plants are grown in borders or other areas where spread is unwanted. Removal of flower spikes will stimulate new vegetative growth (Missouri Botanical Garden 2013).

Chemical

General herbicides, such as glyphosate, are effective at controlling *Mentha* spp. (MISIN and MNFI 2013d).

***Myosotis scorpioides* L.**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species. **Wisconsin** has a proposal to list this species as restricted.

Control

Control options have not been very well documented. This species is likely very difficult to control due to abundant seed production and spread via stolons.

Biological

There are no known biological control methods for this species.

Physical

This plant cannot survive exposure to temperatures below -33°F (USDA NCRS 2012).

***Myosoton aquaticum* (L.) Moench**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Control options have not been very well documented.

Biological

There are no known biological control methods for this species.

Physical

Mow before seeds form, reseed heavily infested areas with competitive forage species (Doll et al. 2004).

Chemical

Dicamba or glyphosate are considered effective controls (Doll et al. 2004).

***Myriophyllum spicatum* L.**

Regulations (pertaining to the Great Lakes region)

Myriophyllum spicatum is a prohibited species in Illinois and Michigan; its hybrids and variants are also prohibited in Minnesota and Wisconsin (GLPANS 2008). In Michigan, a person cannot knowingly possess a live organism (Latimore et al. 2011). In Minnesota, it is illegal to possess, import, purchase, sell, propagate, transport or introduce Eurasian Watermilfoil (Falck et al. 2011).

The Great Lakes Indian Fish & Wildlife Commission listed this species as a “high priority” for control within their ceded territories (Falck et al. 2012).

Control

Due to decades of university, state and federal research and experience with *M. spicatum* in the United States and Canada, several methods have been developed to help in its management.

The best way to minimize the spread of *M. spicatum* is to remove any visible plant fragments and rinse all equipment; allow them to dry completely before using them in another waterbody (IL DNR 2009a).

Biological

Since 1963, the Grass Carp, *Ctenopharyngodon idella* has been released to suppress Eurasian Watermilfoil and other nuisance aquatic plants in numerous sites within North America (Julien and Griffiths 1998, CEH 2004d). It has been found that Grass Carp may only eat Eurasian Watermilfoil after native plants have been consumed (IL DNR 2009a). To achieve control of Eurasian Watermilfoil generally means the total removal of more palatable native aquatic species before the Grass Carp will consume Eurasian Watermilfoil. In situations where Eurasian Watermilfoil is the only aquatic plant species in the lake, this may be acceptable. However, generally Grass Carp are not recommended for Eurasian Watermilfoil control (Washington State Department of Ecology 2013).

Laboratory research has shown that the fungus *Mycoleptodiscus terrestris* reduces the biomass of *M. spicatum* significantly and may be a possible biocontrol (biopesticide) agent (IL DNR 2009a).

A North American weevil, *Euhrychiopsis lecontei*, may be associated with natural declines at northern lakes (Sheldon 1994, Creed and Sheldon 1995). *Euhrychiopsis lecontei* feeds on the new growth of *M. spicatum* and can help keep populations under control; it is common for the populations of for *E. lecontei* and *M. spicatum* to exhibit the classic predator-prey cycles (Creed and Sheldon 1995, Michigan Sea Grant 2012b). Studies have found the herbivorous weevil to cause significant damage to Eurasian water-milfoil while having little impact on native species, suggesting the insect as a potential biocontrol agent (Creed and Sheldon 1995). Female weevils have great fecundity when raised the on *M. spicatum* as opposed to native *M. sibiricum* (Solarz and Newman 1996, Creed 1998, TNC Vermont 1998, Sheldon and Jones 2001).

Physical

Because this plant spreads readily through fragmentation, mechanical controls such as cutting, harvesting, and rotoation (underwater rototilling) should be used only when the extent of the infestation is such that all available niches have been filled. Using mechanical controls while the plant is still invading will tend to enhance its rate of spread.

Mechanical harvesting has been widely used in the Midwest (RICRMC 2007). Small populations of Eurasian Watermilfoil, such as those around docks or in swimming areas, can be removed by hand-pulling and/or the use of a sturdy handrake (Bargeron et al. 2003). Multiple harvests within the same growing season will yield the best results (Bargeron et al. 2003). If multiple harvests are not possible, the single harvest should happen before peak biomass, in early summer, otherwise regrowth will occur (Bargeron et al. 2003, WI DNR 2012b). Large equipment exists to mechanically remove milfoil in larger areas (Bargeron et al. 2003). Dredging is also effective method of removal (CEH 2004d). Care should be taken to remove all fragments to prevent regrowth or deoxygenation from plant decomposition (CEH 2004d, MISIN and MNFI 2013b). Plant fragments can be disposed of by burning, burying, composting (away from the water), or by trash disposal (IL DNR 2009a). In Okanagan Lake, British Columbia, authorities have apparently successfully experimented with management by simultaneously rototilling plants and roots and underwater vacuuming (Newroth 1988).

Where possible, Eurasian Watermilfoil can be drowned or dehydrated by water level manipulation (Bates et al. 1985, Bargeron et al. 2003, WIDNR 2012b). Water drawdowns are most effective when the plants are exposed to several weeks of drying time and root crowns are exposed to sub-freezing temperatures (IL DNR 2009a). This method could have serious effects on other aquatic life (IL DNR 2009a).

Water level manipulation is often used conjunction with herbicides and/or shade barriers (Swearingen et al. 2002, Bargeron et al. 2003). Localized control (in swimming areas and around docks) can be achieved by covering the sediment with an opaque fabric which blocks light from the plants (bottom barriers or screens).

Myriophyllum spicatum is also susceptible to ultrasound pulses and this could prove to be a more selective physical method of control (USACE 2012b).

Chemical

Numerous chemicals will have an effect on *M. spicatum*: amine salts of Endothall (Hyrothol 191®), and Dipotassium Salts of Endothall (Aquathol K®), Diquat dibromide (Reward®), Komeen®, cooper, and confentrazone (Water Bureau 2005, RICRMC 2007).

Myriophyllum spicatum is also susceptible to terbutryn (Clarosan 1FG®) and dichlobenil (Midstream GSR®, Casoron G® and Luxan®). Treatment with either chemical is most effective when applied in spring before the plant is fully grown and in still water (CEH 2004). Herbicides containing dichlobenil will also affects rooted submerged plants and some rushes (CEH 2004d).

The amine formulations of 2,4-D granules (Navigate®, Aquakleen®, Aquacide®) are effective on controlling Eurasian Watermilfoil and will not damage grasses (Lembi 2003, Water Bureau 2005, IL DNR 2009a). The liquid formulation of 2,4-D can be used in ponds and lakes at concentrations less than 2.0 parts per million (Barger et al. 2003). This herbicide method is not appropriate for large unmanageable areas of milfoil (Barger et al. 2003).

One lose-dose application (10 µg/ L) of fluridone (Sonar® and Avast!®) applied in the early stages of growth can result in season long control of Eurasian Watermilfoil (Water Bureau 2005,USACE 2012b, WIDNR 2012b). This application rate resulted in >93% control for a year post-treatment in seven out of eight test lakes in Michigan (USACE 2012b). However, the Minnesota Department of Natural Resources found that the application rate of 10 parts per billions would cause unavoidable damage to native vegetation (Welling 2013).

Liquid triclopyr (Renovate 3® and Renovate® OTF) will provide effective control of Eurasian Watermilfoil and is safe to use around grasses and cattails (Lembi 2003, IL DNR 2009a). A concentration of 0.75 parts per million of triclopyr was used to control Eurasian Watermilfoil in Loon Lake, New York (Miller 2013).

Light attenuating dyes (Aquashade® and Admiral®) also exhibit effective control of this species (USACE 2011b).

***Najas marina* L.**

Regulations (pertaining to the Great Lakes)

Listed as a “species of special concern” in Minnesota; meaning it is extremely uncommon and deserves careful monitoring of its status (MN DNR 2013b).

Control

Biological

While waterfowl and fish may consume parts of *N. marina*; there are no known biological control methods for this species (Agami and Waisel 1986, Tarver et al. 1986, Agami and Waisel 1988).

Physical

There are no known physical control methods for this species.

Chemical

Herbicides containing Endothall (Aquathol K® Liquid, Aquathol Super K® granular), Diquat (Reward®), or Fluridone (Sonar®, Avast!®) are effective against *N. marina* (Lembi 2003).

***Najas minor* All.**

Regulations (pertaining to the Great Lakes region)

Najas minor is prohibited in Minnesota, Wisconsin, and Illinois (GLPANS 2008). In Minnesota it is illegal to possess, import, purchase, sell, propagate, transport, or introduce *N. minor* or any related varieties or hybrids (Invasive Species Program 2011). The New York Invasive Species Council ranks this species moderate ecological risk and recommends that the species be regulated (NYISC 2010).

The Great Lakes Indian Fish & Wildlife Commission ranked this species as a “low priority” for control in 2011, because it was not detected in their ceded territories (Falck et al. 2012).

Control

Biological

There are no known biological control methods for this species (Ohio EPA 2001).

Physical

Manual removal may provide short-term relief by reducing the biomass of *N. najas*, however, small plant fragments may break off and create new plants/infestations (Ohio EPA 2001, Robinson 2004, Office of Water Resources 2010).

Benthic barriers (which restrict light and upward growth of submerged plants) may be effective in controlling *N. minor* in high traffic areas: boating lanes, docks, and swimming beaches (Robinson 2004). These structures need to be anchored to the sediment and regularly maintained, which may impact other benthic and/or plant organisms (Robinson 2004).

Chemical

Herbicides may be most effective for controlling large populations of Brittle Waternymph (Office of Water Resources 2010). Herbicides containing amine salts of Endothall (Hydrothol 191®), dipotassium salt of Endothall (Aquathol K® Liquid, Aquathol Super K® granular), Diquat dibromide (Reward®), or Fluridone (Sonar®, Avast!®) will control *N. minor* (Robinson 2004, Water Bureau 2005). Cutrine®, Komeen®, Nautique®, and Weedtrine®, will also provide effective control of Brittle Waternymph (Robinson 2004, Water Bureau 2005).

Nasturtium officinale

Regulations (pertaining to the Great Lakes region)

Nasturtium officinale is prohibited in Illinois (GLPANS 2008). Even though it is not restricted or prohibited, the Wisconsin Department of Natural Resources acknowledges this species as being highly invasive and recommends its eradication upon detection (Robert W. Freckman Herbarium 2012).

In 2001, the Great Lakes Indian Fish & Wildlife Commission determined that this species has low to moderate ecological impacts, and is difficult to control given the widespread populations and/or limited effective control options (Falck and Garske 2003).

Control

Biological

There are no known biological control methods for this species.

Physical

Manual removal of *N. officinale* offers control for small populations (WIDNR 2010).

Chemical

The non-selective compound glyphosate will provide some control; however it will not be effective in flowing water and will harm other plant species if it comes in contact (WIDNR 2010b).

Nymphoides peltata

Regulations (pertaining to the Great Lakes region)

Nymphoides peltata is prohibited in Illinois, Michigan, and Wisconsin (GLPANS 2008). The New York Invasive Species Council ranks this species as posing a high ecological risk, and recommends that it be prohibited within the state (NYISC 2010).

The Great Lakes Indian Fish & Wildlife Commission has not found *N. peltata* in their ceded territories, but recommend immediate control upon detection (Falck and Garske 2003).

Control

Biological

There are no known biological control methods for this species (CEH 2004c).

Physical

Hand-pulling and mechanical removal is possible because the stems are easily cut by hand tools (CEH 2004c, MISIN 2013). Hand raking or using a rope and grapnel is effective when the bottom sediments are loose (CEH 2004c). All plants pieces should be removed because new plants can grow from broken fragments and/or the decomposing plant material could decrease the oxygen levels in the water (CEH 2004c, Kelly and Maguire 2009). Booms or nets can be used to catch and remove drifting plant materials (Kelly and Maguire 2009). Even with multiple harvests, 100% control is unlikely (CEH 2004c).

Chemical

Herbicides containing dichlobenil (Midstream GSR®, Casoron G®, Luxan dichlobenil®) are effective at controlling *N. peltata* (CEH 2004c). Granules should be sprinkled evenly over this species in early spring, after growth has started, but before the leaves reach the water surface (CEH 2004c). To ensure eradication, plants may need to be cut back and subjected to a second application and/or be treated with another application the following spring (CEH 2004c).

Glyphosate can be used to control *N. peltata*, but it is less effective than dichlobenil and does not offer long-term control (CEH 2004c).

***Pluchea odorata succulenta* (Fern.) Cronq.**

Regulations (pertaining to the Great Lakes region)

In Pennsylvania, *Pluchea odorata* is listed as an endangered species (USDA NRCS 2012).

Control

Biological

The fruit fly *Acinia picturata* has been known to use *P. odorata* as a host, but it is unknown if this species could be used a biological control agent (Stegmaier 1967).

Physical

There are no known physical control methods for this species.

Chemical

There are no known chemical control methods for this species

***Pluchea odorata odorata* (L.) Cass.**

Regulations (pertaining to the Great Lakes region)

In Pennsylvania, *P. odorata* is listed as an endangered species (USDA NRCS 2012).

Control

Biological

The fruit fly *Acinia picturata* has been known to use *P. odorata* as a host, but it is unknown if this species could be used a biological control agent (Stegmaier 1967).

Physical

There are no known physical control methods for this species.

Chemical

There are no known chemical control methods for this species.

***Poa trivialis* L.**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Poa trivialis is used as a turf grass, often overseeded with Bermuda grass. Most research on management of this species focuses on promotion of the plants rather than control. Its tendency to yellow in the heat of summer makes it undesirable in some turf applications, and most information on control is based on these applications.

Biological

Combining herbicides with overseeding an alternative desired grass will help discourage the regrowth of surviving *P. trivialis* and improve overall success of control (Morton and Reicher 2007).

Physical

There are no known physical control methods for this species.

Chemical

Non selective control with glyphosate (Roundup) followed by reseeding has been the only option for control. A herbicide called sulfosulfuron (Certainty®) is currently being labeled by Monsanto for selective control of *Poa trivialis* in Creeping Bentgrass (Street and Sherratt 2013).

Other

This species has low tolerance for high temperatures and drought.

***Polygonum persicaria* L.**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

Although *P. persicaria* plants are susceptible to Arabis Mosaic Virus, no research has been undertaken on the development of biological control agents, whether viral or fungal. No arthropods known to attack this species specifically have been identified (Plantwise 2013).

Physical

When handling Smartweed, do not place hands close to eyes – it contains a chemical which burns.

Management of this species in agricultural settings focuses on elimination of seed as a contaminant and cultivation to prevent seedling establishment. Tillage and cultivation which disrupts seedlings can be effective. Repeated mowing to prevent seed production can also reduce populations over time (Plantwise 2013).

Solarizing black plastic, burning and acetic acid treatments are all effective at killing seeds.

Chemical

P. persicaria seems relatively quick to develop resistance to herbicides – most resources discourage use of chemical control or encourage rotation of herbicides with differing modes of action (Plantwise 2013).

Pre-emergent herbicides, especially with the chemical Dichlobenil, are effective. Use on-going for up to one year. Systemic herbicides to kill the fibrous roots can also work well. Other options for herbicide control are non-selective, contact herbicides. Products containing dicamba work best over others containing 2,4-D and glyphosate (Easy Butterfly Garden 2013).

Potamogeton crispus

Regulations (pertaining to the Great Lakes region)

Potamogeton crispus is prohibited in Illinois and Minnesota; restricted in Michigan and Wisconsin (GLPANS 2008). The New York Invasive Species Council ranks this species as posing a “high” ecological risk, and recommends that it be prohibited within the state (NYISC 2010).

As of 2011, the Great Lakes Indian Fish & Wildlife Commission lists this species a high priority species and recommends it be controlled within their ceded territories (Falck et al. 2012).

Control

Biological

The herbivorous Grass Carp, *Ctenpharyngodon idella*, will provide effective control of *P. crispus*, but may feed on native plants (CEH 2004e). Grass Carp is illegal in some Great Lake states (GLIFWC 2006).

Other bottom feeding fish, such as Common Carp, do not feed on *P. crispus*, but they create turbid water conditions and may prevent the growth of this plant species (CEH 2004e).

Physical

Small infestations can be removed manually by cutting, raking, or digging up plants (IISCTC 2007). The optimal timing for cutting is debated. Some agencies claim that plants should be cut in early spring and as close to the sediment surface as possible to prevent turion formation (MISIN and MNFI 2013a, WIDNR 2012a). Other organizations claim that cutting should not be carried out until mid-to later summer to prevent regrowth (CEH 2004e). Regardless when cutting/raking occurs, it is important to remove as many plant fragments as possible to limit new populations of Curlyleaf Pondweed (PADCNr n.d.).

When removing this species via digging, root crowns should also be removed from the soil; this removal method can be enhanced by the use of a suction apparatus (ENSR International 2005).

The use of equipment such as dredges, underwater rototillers, or hydrorakes are more effective for populations in deep waters (ENSR International 2005, USACE 2012b). These physical methods are indiscriminate and should only be used on monoculture populations of *P. crispus* (ENSR International 2005). Plant material should be removed after it is cut to prevent regrowth or decreases in oxygen concentration due to plant decomposition (ENSR International 2005).

Another option would be to use blankets or other benthic barriers to block sunlight from reaching *P. crispus* (ENSR International 2005). This method will eliminate all vegetation, including native species, in 30 – 60 days (ENSR International 2005, GLIFWC 2006).

In some waterbodies, water draw-down may be an option. All plants, including natives, will be exposed to drying or freezing (ENSR International 2005). A water draw-down in autumn may kill *P. crispus* turions and increase the efficacy of this control method (MISIN and MNFI 2013a).

Chemical

P. crispus plants dieback completely in early summer; in order for effective control, herbicides should be applied before dieback occurs (MISIN and MNFI 2013a).

P. crispus was effectively controlled by fluridone (Sonar®, Avast!®) in test site lakes in Michigan (Getsinger et al. 2001). Control can be obtained with a dose of 6 - 15 ppb with an exposure time of 60 – 120 day (ENSR International 2005). This method is only appropriate for whole lake applications (IL DNR 2009b).

Endothall (Aquathol K® Liquid, Aquathol Super K® granular) and diquat (Reward®) may offer effective control if applied to *P. crispus* before turion production; typically in April and May (ENSR International 2005, WIDNR 2012a). Plants may still continue to grow, but their reproductive ability will be greatly reduced (ENSR). Application of either of these chemicals is most effective when the water temperature is between 50° – 55° F (IL DNR 2009b). Reapplication of diquat in subsequent years may be necessary for complete control (Bugbee 2009).

The compounds dichlobenil (Midstream GSR®, Casoron G®, Luxan Dichlobenil® Granules) or terbutryn (Clarosan®) will also provide effective control if applied in early spring just as growth starts (CEH 2004e)

Herbicides containing 2,4-D will be rapidly taken up by *P. crispus*, but complete control is unlikely (ENSR International 2005).

***Puccinellia distans* (Jacq.) Parl.**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Chemical

Alkali grasses are extremely tolerant of salinity and outcompete other grasses in most brackish conditions. Thus management efforts in the freshwater Great Lakes regions targeted at reducing salt contamination may benefit efforts to control *P. distans*.

***Rorippa sylvestris* (L.) Bess.**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Once established, this plant is very difficult to control.

Physical

Readily re-grows from rhizome fragments – most attempts at physical control only exacerbate the problem when small fragments are missed and regrow.

Chemical

Pre-emergent herbicides suppress top growth but do not kill rhizomes which quickly produce new plantlets in response to the loss of the top. Glyphosate is marginally effective. Selective post-emergent herbicides are not available (NC State 2012)

Preemergence control was excellent with dichlobenil granules at 3 or 6 lb/A, isoxaben at 1 or 2 lb/A and the geotextile/herbicide (Biobarrier). The geotextile (Tympar) fitted as collars alone were not effective. Trifluralin incorporated into the surface two inches at 2 lb/A was effective but did allow some emergence. Trifluralin plus isoxaben or oryzalin plus isoxaben were also effective at rates of 2 plus 0.5, 4 plus 1, or 6 plus 1.5 lb/A, or 3 plus 1, 4.5 plus 1.5, or 6 plus 2 lb/A, respectively, of the two herbicide combinations. Metolachlor at 3, 4.5, or 6 lb/A was ineffective for preemergence control of three cm rhizome pieces. Post emergence control was not commercial with 2,4-D, triclopyr, clopyralid or a combination of the latter two, when treated in the 6 to 8 leaf stage with 0.25% or 0.5% solutions (Elmore et al. 1996)

***Rumex longifolius* DC.**

Regulations (pertaining to the Great Lakes region)

There are no federal or state (within the Great Lakes region) regulations for this species.

Control

Biological

Docks are grazed by cattle, sheep, goats, and deer but not by horses.

Physical

Repeated cultivation is recommended for control of young (seedling) populations.

Mowing has little effect on established docks, but will prevent seed production. However, frequent cutting encourages taproot growth, branching shoots and may aid seedling development (from previous year's seed bank) and so is not recommended. In a pasture heavily infested with docks the best option may be to plough and reseed with grass but not immediately. The docks are likely to regenerate both vegetatively and from seed, and a period of fallowing or arable cropping may help to reduce re-establishment.

Chemical

Many chemical controls are available for dock species. However, very few are approved for use in or near water. Repeated treatments are usually needed to control re-growth.

Dicamba (benzoic acid) is effective on Curly Dock (*Rumex crispus*) but not on Broadleaf Dock (*R. obtusifolius*). Picloram (pyridine) is effective on most *Rumex* species. 2,4-DB amine or 2,4-D ester are effective when applied before the flower stalk elongates, but require a 30 day withdrawal before feeding as forage. Aminopyralid can be applied to actively growing plants before the bud stage. Chlorsulfuron and metsulfuron can be used with young, actively growing plants, but should not be used on powdery, dry, or light sandy soils. Sulfoteturon has similar use,

but should not be applied to cropland. Glyphosate can be used at early heading (Pacific Northwest Extension 2013).

Rumex obtusifolius

Regulations (pertaining to the Great Lakes region)

The Great Lakes Indian Fish & Wildlife Commission ranked *Rumex obtusifolius* as capable of low to moderate ecological impacts and does not consider it a priority for control within their ceded territories (Falcke and Garske 2003). There are no federal (or state within the Great Lakes region) regulations for this species.

Control

Biological

Bitter Dock is avoided by rabbits, but it appeared to be a favorite food plant of deer (Amphlett and Rea 1909). Docks are grazed by cattle, sheep, and goats, but not by horses.

Cavers and Harper (1964) list a range of fungi and insects that attack, feed on or occur on docks but this not an indication that of their efficacy as control agents. The use of the stem boring larvae of the weevils *Apion violaceum* and *A. miniatum* for controlling *R. obtusifolius* has been investigated (Hopkins 1980, Freese 1995). In the United Kingdom and elsewhere, there has been research on the chrysomelid beetle (*Gastrophysa viridula*) as a biocontrol agent for both *R. obtusifolius* and *R. crispus* (Bentley et al. 1980). Larvae of the Leaf-Mining Fly *Pegomya nigratarsis* cause blotch mines on leaves of *R. obtusifolius* (Whittaker 1994). *R. obtusifolius* is the preferred host plant of *Coreus marginatus* and has been shown to moderately reduce its seed viability (Hruskova et al. 2005). The Leaf Spot Fungus *Ramularia rubella* causes red spots to develop on dock leaves but has no major effect on plant survival. The Rust fungus *Uromyces rumicis* is also non-systemic but has been shown to have some potential as a biological control agent (Inman 1971, Schubiger et al. 1986). Dock species are also an alternate host for number of viruses, fungus (Dal Bello and Carranza 1995), and nematodes (Townshend and Davidson 1962, Edwards and Taylor 1963).

Physical

Caution should be used in physical removal as this plant can cause contact dermatitis.

Repeated cultivation is recommended for control of young (seedling) populations.

Mowing has little effect on established docks, but will prevent seed production. However, frequent cutting encourages taproot growth, branching shoots and may aid seedling development (from previous year's seed bank) and so is not recommended. In a pasture heavily infested with docks the best option may be to plough and reseed with grass but not immediately. The docks are likely to regenerate both vegetatively and from seed, and a period of fallowing or arable cropping may help to reduce re-establishment.

Chemical

Many chemical controls are available for dock species. However, very few are approved for use in or near water. Repeated treatments are usually needed to control re-growth.

Dicamba (benzoic acid) is effective on Curly Dock (*Rumex crispus*) but not on Broadleaf Dock (*R. obtusifolius*). Picloram (pyridine) is effective on most *Rumex* species. 2,4-DB amine or 2,4-D ester are effective when applied before the flower stalk elongates, but require a 30 day withdrawal before feeding as forage. Aminopyralid can be applied to actively growing plants before the bud stage. Chlorsulfuron and metsulfuron can be used with young, actively growing plants, but should not be used on powdery, dry, or light sandy soils. Sulfoteturon has similar use, but should not be applied to cropland. Glyphosate can be used at early heading (Pacific Northwest Extension 2013).

Salix alba

Regulations (pertaining to the Great Lakes region)

In 2011, the Great Lakes Indian Fish & Wildlife Commission ranked *S. alba* to be a low priority for regulation and control (Falck et al. 2012). There are no known federal or state (within the Great Lakes region) regulations for this species.

Control

Biological

To our knowledge no research into possible biological control organisms for White Willow has been attempted in North America or anywhere else. Candidates for insect control are generally not sufficiently specific to avoid damage to native willows.

Physical

Like most river trees willows resprout vigorously from cut stumps, and will usually grow back into a tree eventually. Repeated cutting of new stump shoots can eventually kill the trees. But given their large root systems, cutting would presumably be needed to be done several times per growing season for several years in order to starve the roots. Mowing or weed-whipping might be useful for seedlings (GLIFWC 2013). Small seedlings can be handpulled, Larger trees may require a weed wrench or machinery to remove the root systems.

Chemical

Given willows' tendency to resprout from the roots, multiple applications will likely be needed for control.

The nonselective herbicide glyphosate (available commercially as "Roundup" and "Rodeo") is commonly used for treating woody invasives such as Crack Willows. Triclopyr (Garlon 3A or equivalent amine formulation) is also effective against broadleaf and woody plants, and has the advantage of leaving grasses and sedges intact. Recently Roundup has been shown to be highly toxic to both adult frogs and toads and their tadpoles, probably due to the surfactant (polyethoxylated tallowamine, or POEA) in this glyphosate formulation (Relyea 2005). Because of this and other as yet unknown effects of various herbicide formulations on the environment, herbicide should be applied as precisely as possible and only when needed, using only the amount needed to get the job done. Any attempt to control Crack Willows or other invasive plants in aquatic habitats must be done using Rodeo or other herbicides formulated for use over

water. Permits are required for herbicide application over water in many states, including Wisconsin, Michigan and Minnesota (GLIFWC 2013).

Triclopyr amine is best used on young willows (seedlings) that are actively growing. 2,4-D LV ester can be applied when leaves are fully developed and growing (amenable to aerial application). Metsulfuron is used on fully leafed-out brush.

Salix fragilis

Regulations (pertaining to the Great Lakes region)

In 2011, the Great Lakes Indian Fish & Wildlife Commission ranked *Salix fragilis* to be a low priority for regulation and control (Falck et al. 2012). There are no known regulations for this species.

Control

Biological

To our knowledge no research into possible biological control organisms for Crack Willows has been attempted in North America or anywhere else. Candidates for insect control are generally not sufficiently specific to avoid damage to native willows.

Physical

Like most river trees willows resprout vigorously from cut stumps, and will usually grow back into a tree eventually. Repeated cutting of new stump shoots can eventually kill the trees. But given their large root systems, cutting would presumably be needed to be done several times per growing season for several years in order to starve the roots. Mowing or weed-whipping might be useful for seedlings (GLIFWC 2013). Small seedlings can be handpulled. Larger trees may require a weed wrench or machinery to remove the root systems.

Chemical

Given willows' tendency to resprout from the roots, multiple applications will likely be needed for control.

The nonselective herbicide glyphosate (available commercially as "Roundup" and "Rodeo") is commonly used for treating woody invasives such as Crack Willows. Triclopyr (Garlon 3A or equivalent amine formulation) is also effective against broadleaf and woody plants, and has the advantage of leaving grasses and sedges intact. Recently Roundup has been shown to be highly toxic to both adult frogs and toads and their tadpoles, probably due to the surfactant (polyethoxylated tallowamine, or POEA) in this glyphosate formulation (Relyea 2005). Because of this and other as yet unknown effects of various herbicide formulations on the environment, herbicide should be applied as precisely as possible and only when needed, using only the amount needed to get the job done. Any attempt to control Crack Willows or other invasive plants in aquatic habitats must be done using Rodeo or other herbicides formulated for use over water. Permits are required for herbicide application over water in many states, including Wisconsin, Michigan and Minnesota (GLIFWC 2013).

Triclopyr amine is best used on young willows (seedlings) that are actively growing. 2,4-D LV ester can be applied when leaves are fully developed and growing (amendable to aerial application). Metsulfuron is used on fully leafed-out brush.

Salix purpurea

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Cultivars of *Salix purpurea* are often used for erosion control and most species-specific management information stems from this use.

Biological

To our knowledge no research into possible biological control organisms for willows has been attempted in North America or anywhere else. Candidates for insect control are generally not sufficiently specific to avoid damage to native willows.

Gypsy moths can defoliate Purple Willow and Willow Midges can cause significant (though rarely fatal) damage. Willow blight is a serious pest to plantings, particularly plants that have been damaged by storms. Beaver can have huge impact on this species and are capable of eradicating entire stands (USDA NRCS 2002)

Physical

Like most river trees willows resprout vigorously from cut stumps, and will usually grow back into a tree eventually. Repeated cutting of new stump shoots can eventually kill the trees. But given their large root systems, cutting would presumably be needed to be done several times per growing season for several years in order to starve the roots. Mowing or weed-whipping might be useful for seedlings (GLIFWC 2013). Small seedlings can be handpulled, Larger trees may require a weed wrench or machinery to remove the root systems.

Chemical

Given willows' tendency to resprout from the roots, multiple applications will likely be needed for control.

The nonselective herbicide glyphosate (available commercially as "Roundup" and "Rodeo") is commonly used for treating woody invasives such as Crack Willows. Triclopyr (Garlon 3A or equivalent amine formulation) is also effective against broadleaf and woody plants, and has the advantage of leaving grasses and sedges intact. Recently, Roundup has been shown to be highly toxic to both adult frogs and toads and their tadpoles, probably due to the surfactant (polyethoxylated tallowamine, or POEA) in this glyphosate formulation (Relyea 2005). Because of this and other as yet unknown effects of various herbicide formulations on the environment, herbicide should be applied as precisely as possible and only when needed, using only the amount needed to get the job done. Any attempt to control Crack Willows or other invasive plants in aquatic habitats must be done using Rodeo or other herbicides formulated for use over water. Permits are required for herbicide application over water in many states, including Wisconsin, Michigan and Minnesota (GLIFWC 2013).

Triclopyr amine is best used on young willows (seedlings) that are actively growing. 2,4-D LV ester can be applied when leaves are fully developed and growing (amenable to aerial application). Metsulfuron is used on fully leafed-out brush.

Solanum dulcamara

Regulations (pertaining to the Great Lakes region)

After determining that *Solanum dulcamara* poses a low ecological impact, the Nature Conservancy gave this species a “low” national priority ranking (NatureServe 2008). The New York Invasive Species Council determined that this species poses a moderate ecological risk, and therefore recommended that this species be regulated (NYISC 2010).

As of 2011, the Great Lakes Indian Fish & Wildlife Commission ranked *S. dulcamara* as a “lower priority” (Falck et al. 2012).

Control

Biological

There are no known biological control methods for this species (King County 2010c).

Physical

S. dulcamara may be controlled by manually digging up the roots; most effective with the plants are young and the soil is moist (Waggy 2009). All parts of the roots should be removed to prevent regrowth (King County 2010c). Gloves should be worn when handling this species to prevent skin irritation (King County 2010c).

Mowing is not a practical method for control because this species can resprout from the suckering roots and rhizomes; however it may be useful when manually removing the root system (King County 2007b). If mowing is the only option, it must be done several times during the growing season to be effective (Forest Health Staff 2006a). Another option would be to cover the cut plants with a heavy geotextile cloth for two years to prevent photosynthesis and regrowth (King County 2010c).

Chemical

Larger infestations may require the use of an herbicide (Forest Health Staff 2006a). The herbicide Clopyralid (Transline®, Stinger®, Reclaim®, and Curtail®) is effective on Solanaceae (Tu et al. 2001). Triclopyr is effectively taken up in the woody stems, roots, and leaves of Bittersweet Nightshade. It is also unlikely to harm nearby grasses, sedges, rushes, cattails, lilies, and irises (King County 2010c).

***Solidago sempervirens* L.**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Seaside Goldenrod is being actively planted for dune restoration and wildlife habitat on the east coast. Most of the available research-based management information focuses on growing the plant rather than control.

Biological

There are no known biological control methods for this species.

Physical

Spreads only slowly other than by seed, so deadheading or other cutting prior to seed-set can be effective for control.

Chemical

This is a salt-adapted species. Efforts to control salt-pollution in the freshwater Great Lakes region may help to control populations of this species.

***Sparganium glomeratum* (Laestad.) L. Neum.**

Regulations (pertaining to the Great Lakes region)

Under the synonym, *Sparganium erectum*, this species is federally listed as a noxious weed ([Code of Federal Regulations § Title 7, 360.200](#)). It is additionally listed as prohibited in Minnesota ([MN Administrative Rules § 6216.0250](#)).

Control

Little to no information is readily available on control of *S. glomeratum*.

Trapa natans

Regulations (pertaining to the Great Lakes region)

Trapa natans is prohibited in Illinois, Michigan, Minnesota, New York, and Wisconsin (GLPANS 2008).

The Great Lakes Indian Fish & Wildlife Commission have not found *T. natans* in their ceded territories, but recommended immediate control upon detection (Falck and Garske 2003).

Control

An integrated management plan that incorporates multiple methods of control will be most effective at controlling populations of *T. natans*. Invaded habitats should be monitored for up to 12 years after control measures are complete to ensure that the seed bank is exhausted (PADCNr n.d., Swearingen et al. 2002).

Biological

In its native range in China, the leaf beetle *Galerucella birmanica* has significant negative impacts on *T. natans* populations (Ding et al. 2006). However, this species has many other host species in the United States, making it unsuitable for use as a biocontrol agent (Maryland Sea Grant 2012).

Physical

Smaller populations can be controlled by hand harvesting or raking because the roots are easily uplifted from the sediment (Naylor 2003). Larger populations, including those thick enough to clog waterways, may require the use of a large aquatic plant harvester (PADCNR n.d.).

Harvesting methods should be conducted before plants set seeds-- typically in July (Maryland Sea Grant 2012). All plant fragments, especially those containing roots, should be removed to prevent the expansion of the *T. natans* population (Swearingen et al. 2002). Plant fragments should be disposed of far from the water, preferably in a plastic bag (PADCNR n.d.).

260,000 lbs. of Water Chestnut were removed by mechanical means and the help of over 60 volunteers from the Sassfras River (Maryland) during a three day harvest in 1999 (Naylor 2003). Mechanical removal methods have been used annually in Sodus Bay, New York since the 1960s, but the *T. natans* population persists (US EPA 2000). However, mechanical removal followed by an application(s) of 2,4-D was able to eradicate a population of *T. natans* in Maryland (Naylor 2003).

Laboratory and greenhouse studies by Wu and Wu (2007) demonstrated that ultrasonic waves of 20 kHz, aimed directly at Water Chestnut stems and petioles, for 10 seconds resulted in 100% plant death.

Chemical

Herbicides containing 2,4-D (both the amine and butoxy-ethyl ester formulations) have been effective in controlling *T. natans* (USACE 2012b, WIDNR 2012e) Applying 2,4-D just as plants are reaching the surface of the water, in early summer, will provide the best results (USACE 2012b). This compound causes minimal adverse effects on neighboring wildlife (Maryland Sea Grant 2012).

Herbicides containing triclopyr are also effective at controlling *T. natans*, but it is non-selective and may harm other plant life (USACE 2012b).

The growth and expansion of Water Chestnut populations can also be repressed if light attenuating dyes are applied prior to plant germination (USACE 2012b).

***Typha angustifolia* L.**

Regulations (pertaining to the Great Lakes region)

This species is restricted in **Wisconsin**; it may not be transported, transferred, or introduced into any ecosystem (Bureau of Plant Industry 2012).

Control

Biological

Muskrat (*Ondatra zibethicus*) populations can have a serious impact on *Typha* populations; however, large populations of muskrats can shift to other plants species and have a long-term detrimental effect on the vegetation community (Miklovic 2000).

The native boring-moth larvae (*Arzama* spp.) have been reported to cause damage to *Typha* stands, but their use as a species specific biological control is unknown (Miklovic 2000).

Heavy grazing will eliminate *Typha* spp. from riparian corridors; however, this technique might also affect other native species (Stevens and Hoag 2006).

Physical

Mowing during the growing season, once just before the flowers reach maturity and again about a month later (when new growth is 2-3 feet high), will kill at least 75% of Narrow-Leaved Cattails (Stevens and Hoag 2006).

Burning may also be effective at controlling *Typha*; however, it needs to be repeated several times. Unless the flames have access to the belowground portions of cattails, the rhizomes will resprout and grow new plants (Forest Health Staff 2006c, USDA Forest Service 2012). This treatment option might also be unfeasible in wet ecosystems or sensitive natural areas (Miklovic 2000).

Typha spp. are sensitive to the ethanol produced from anaerobic respiration. Flooding a wetland could trigger this reaction and help control *Typha* (Miklovic 2000). Manually digging up plants or cutting stems, followed by raising the water level by three inches above the plants will yield effective control, as well (Forest Health Staff 2006c).

Chemical

Typha spp. can be controlled by 2,4-D, glyphosate (Rodeo®, Eagle®, AquaNeat®, Pondmaster®, Aquapro®, Avocet®, Shore-Klear®, Touchdown Pro®), impazapyr (Arsenal AC®, Habitat®, Chopper®, Aquapier®, Gullwing Avocet®), and diquat (Harvester®, Redwing®, Reward®, Weedtrine D®) (Water Bureau 2005, Forest Health Staff 2006c, WIDNR Division of Forestry 2011, AERF 2012). Glyphosate can result in greater than 80% control (Thorsness et al. 1992).

Wick, broom, and/or foliar applications are appropriate techniques for these herbicides (Ohio EPA 2001, Forest Health Staff 2006c, Borland et al. 2009). Due to the energy reserves in the extensive root system, re-treatments may be necessary (Ohio EPA 2001).

Veronica beccabunga

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Noted by several sources as ‘spreading quickly but easy to control. However, we have not been able to track down details for recommended control methods.

A.14 Platyhelminthes

Bothriocephalus acheilognathi Yamaguti, 1934

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Bath treatments are effective control methods for *B. acheilognathi* infections. Baths should contain Droncit® (praziquantel), isopropyl alcohol, and water yielding a final mixture concentration ≥ 0.67 ppm praziquantel. Fish densities during treatment should be no greater than 60 mg fish/L and exposure should last 24 hours. After 24 hours, the treatment should be drained, worm parts discarded, and clean water added. After 72 hours, the treatment should be drained and worm parts discarded. Fish should then be transferred to a decontaminated container (Mitchell and Darwish 2009).

Bothriocephalus acheilognathi infections can be treated with chemically enhanced feed. Drugs should be mixed in oil and sprayed on feed at a rate of 1 L/70 kg dry weight. Effective chemicals and doses include dibutyltin oxide or dibutyltin dilurate (250 mg/kg fish) fed over three days (Mitchell and Hoffman 1980), Yomesan® (500 g/500 kg dry pellets) fed at 1.5% of body weight 2–3 times weekly, and Yomesan® (28 g/40 kg) fed for three days (Korting 1974, Mitchell and Hoffman 1980, Brandt et al. 1981).

Other

Bothriocephalus acheilognathi populations in aquaculture and ponds can be controlled by managing the intermediate host (i.e., copepods) population densities. Effective ectoparasiticides include Neguvon®, Masoten®, Dipterex®, Bromex®, and Naled® (Paperna 1996).

Dactylogyrus amphibothrium Wagener or Wegener, 1857

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

Implementation of Eurasian Ruffe management may potentially decrease *Dactylogyrus amphibothrium* prevalence due to host specificity. However, Ruffe management is considered by some (e.g., Ogle 1998) to be difficult and impractical given that the species has developed

several adaptations to compensate for high mortality rates (Lind 1977) and populations rebound quickly (Lelek 1987).

Physical

Establishment of quarantines may prevent *D. amphibothrium* transmission (Reed et al. 1996)

Chemical

D. amphibothrium-specific treatments are unknown. However, multiple chemicals are effective at treating monogenean fluke infections in aquaculture systems. Effective benzimidazoles include levamisole (Buchmann 1997) and praziquantel, which has high efficacy against *Dactylogyrus* spp. (Buchmann 1997, Schmahl and Mehlhorn 1985). Effective bath treatments include formaldehyde (30-100 ppm), sodium chloride, copper sulphate, hydrogen peroxide, sodium percarbonate (Buchmann and Kristensen 2003), formalin (25 mg/L for prolonged exposure or 150-250 mg/L for 30 minutes), and potassium permanganate (2 mg/L for prolonged exposure or 10 mg/L for 30 minutes) (Reed et al. 1996). Effective organophosphate bath treatments include metrifonate (0.25-0.5 ppm) and dichlorvos (0.25-0.5 ppm) (Sarig et al. 1965).

Pond infestations can be controlled with formalin (30 mg/L) or trichlorfon (Lepidex®; 0.5 mg/L) (Reed et al. 1996). However, monogenean eggs display chemical resilience and therefore the above chemical treatments are ineffective at destroying eggs (Reed et al. 1996, Rowland et al. 2007). Chemical toxicity varies considerably between monogeneans and fish species. Toxicology and tolerance tests are suggested prior to using anthelmintics (“dewormers”). Managers are encouraged to consider specific host drug tolerance, temperature, salinity, organic material content, and drug retention time prior to treatment (Buchmann and Bresciani 2006). Freshwater fish species can also be dipped in saltwater to minimize external parasite numbers prior to stocking (Reed et al. 1996).

Other

Lampricide TFM may effectively eliminate up to 97% of Ruffe, potential carriers of *D. amphibothrium*, with minimal non-target mortality (Crosier et al. 2012).

***Dactylogyrus hemiamphibothrium* Ergens, 1956**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

Implementation of Eurasian Ruffe management may potentially decrease *Dactylogyrus hemiamphibothrium* prevalence due to host specificity. However, Ruffe management is considered by some (e.g., Ogle 1998) to be difficult and impractical given that the species has developed several adaptations to compensate for high mortality rates (Lind 1977) and populations rebound quickly (Lelek 1987).

Physical

Establishment of quarantines may prevent monogenean fluke transmission (Reed et al. 1996).

Chemical

D. hemiamphibothrium specific treatments are unknown. However, multiple chemicals are effective at treating monogenean fluke infections in aquaculture systems. Effective benzimidazoles include levamisole (Buchmann 1997) and praziquantel, which has high efficacy against *Dactylogyra* spp. (Buchmann 1997, Schmahl and Mehlhorn 1985). Effective bath treatments include formaldehyde (30-100 ppm), sodium chloride, copper sulphate, hydrogen peroxide, sodium percarbonate (Buchmann and Kristensen 2003), formalin (25 mg/L for prolonged exposure or 150-250 mg/L for 30 minutes), and potassium permanganate (2 mg/L for prolonged exposure or 10 mg/L for 30 minutes) (Reed et al. 1996). Effective organophosphate bath treatments include metrifonate (0.25-0.5 ppm) and dichlorvos (0.25-0.5 ppm) (Sarig et al. 1965).

Pond infestations can be controlled with formalin (30 mg/L) or trichlorfon (Lepidex®; 0.5 mg/L) (Reed et al. 1996). However, monogenean eggs display chemical resilience and therefore the above chemical treatments are ineffective at destroying eggs (Reed et al. 1996, Rowland et al. 2007). Chemical toxicity varies considerably between monogeneans and fish species. Toxicology and tolerance tests are suggested prior to using anthelmintics (“dewormers”). Managers are encouraged to consider specific host drug tolerance, temperature, salinity, organic material content, and drug retention time prior to treatment (Buchmann and Bresciani 2006). Freshwater fish species can also be dipped in saltwater to minimize external parasite numbers prior to stocking (Reed et al. 1996).

Other

Lampicide TFM may effectively eliminate up to 97% of Ruffe, potential carriers of *D. hemiamphibothrium*, with minimal non-target mortality (Crosier et al. 2012).

***Dugesia polychroa* Schmidt, 1861**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

Ichthyocotylurus pileatus

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

Neascus brevicaudatus von Nordmann, 1832

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

Scolex pleuronectis Müller, 1788

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

***Timoniella* sp.**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

A.15 Protozoa

Acineta nitocrae

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

Glugea hertwigi Weissenberg, 1911

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

***Heterosporis* sp.**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

Biological

There are no known biological control methods for this species.

Physical

Effective physical control methods include complete desiccation of holding tanks and equipment for 24 hours, freezing at -20°C for 24 hours, and culling (Sutherland et al. 2006, GLFHC 2012b).

Chemical

Immersion of gear in a 2200 ppm bleach (0.7L of bleach per 20 L of water) solution for five minutes will destroy the parasite (IDNR 2005, Sutherland et al. 2006, GLFHC 2012b).

Other

Infected fish or fish parts should not be discarded back into the water body.

***Myxobolus cerebralis* Hofer, 1903**

Regulations (pertaining to the Great Lakes region)

The Great Lakes Fish Disease Control Policy and Model Program have prohibited stocking the Great Lakes and their tributaries with fish from whirling disease infected farms. Fish imported into the North Central Region states must be certified free of whirling disease in order to obtain import permits (Faisal and Garling 2004).

Ohio requires out of state source facilities to document annual salmonid fish, egg, and sperm health inspections for one year prior to importation. Source facilities outside the Great Lakes basin must document health inspections for the previous five years with no whirling disease occurrences prior to importing salmonids into the Lake Erie watershed (Baird 2005). Indiana requires source facilities within the Great Lakes basin to document they have been whirling disease free for three consecutive years prior to importing salmonid stock. Source facilities outside the basin must document salmonid stocks have been whirling disease free consecutively since 2002 (Baird 2005). Michigan requires source facilities to document salmonid stocks have been whirling disease free for two consecutive years prior to importation, while Wisconsin requires one. Illinois and Minnesota also require imported salmonid health inspections. Minnesota allows the importation of whirling disease infected eggs, if prior egg treatments are approved (Baird 2005). Ontario requires an import permit issued by the Canadian Food Inspection Agency (CFIA) prior to the importation of certain finfish. Under the Canadian Health of Animals Act, aquaculturists are required to report any whirling disease suspicions to the CFIA (CFIA 2012).

All eight Great Lakes states (New York, Pennsylvania, Ohio, Michigan, Indiana, Illinois, Wisconsin, and Minnesota) have instated similar baitfish regulations to control the spread of whirling disease and other fish pathogens. Those of New York include that bait harvested from inland waters for personal use is only permitted to be used within the same body of water from which it was taken and cannot be transported overland (with the exception of smelt, suckers,

Alewives, and Blueback Herring). Once transported, baitfish cannot be replaced to its original body of water (NYSDEC 2012a).

Live or frozen bait harvested from inland New York waters for commercial purposes is only permitted to be sold or possessed on the same body of water from which it was taken and cannot be transported over land unless under a permit and or accompanied by a fish health certification report. Bait that is preserved and packaged by any method other than freezing, such as salting, can be sold and used wherever the use of bait fish is legal as long as the package is labeled with the name of the packager-processor, the name of the fish species, the quantity of fish packaged, and the means of preservation (NYSDEC 2012a).

Certified bait may be sold for retail and transported overland as long as the consumer maintains a copy of a sales receipt that contains the name of the selling vendor, date sold, species of fish sold, and quantity of fish sold. Bait that has not been certified may still be sold but the consumer must maintain a sales receipt containing the body of water where the baitfish was collected and a warning that the bait cannot be transported by motor vehicle. Bait sold for resale require a fish health certification along with a receipt that contains the name of the selling vendor, date sold, species of fish sold, and quantity of fish sold, which must be kept for 30 days or until all bait is sold (NYSDEC 2012a).

In addition to baitfish protections, prior to placing fish in New York waters, a fish health certification report must document that the fish are whirling disease free.

Control

The following biological, physical and chemical controls only pertain to fish in captive or hatchery operations. There are no known control methods of whirling disease in wild populations (except for management of spread - see below).

Biological

Managing *Tubifex tubifex* populations can be implemented as a biocontrol of *M. cerebralis*. Maintaining water quality, reducing favorable habitat by preventing sediment accumulation in aquaculture (Crosier et al. 2012), and desiccating holding tanks, equipment, and intake pipes may help control *T. tubifex* (Kaster and Bushnell 1981). Lampricide TFM (3-trifluoromethyl-4-nitrophenol), administered at (4.2-14.0 mg/L) doses, is effective at destroying *T. tubifex* (Lieffers 1990). *Tubifex tubifex* can also be treated in 30°C water for four days, causing triactinomyxon (TAM) spore production to stop, thus preventing the next stage of the parasites life cycle (El-Matbouli et al. 1999). *T. tubifex* ability to support *M. cerebralis*' triactinomyxon (TAM) spore production may be due to genetic differences among *T. tubifex* populations. This variability may be an important factor in determining infection rates among fish (Baxa et al. 2006) and therefore might support certain management practices (Stromberg 2006).

It has been proposed that selective processes are yielding a surviving population of fish that is more resistant to *M. cerebralis* infection on the Madison River, Montana (Vincent 2006). The implications of this for management are still unclear. However, research is continuing to evaluate the possibility of a developing resistance within salmonid populations (Stromberg 2006).

Physical

Managers have observed that using concrete in aquaculture facilities can reduce the abundance of *T. tubifex* and thus limit the ability of *M. cerebralis* to reproduce (Mills et al. 1993, Ricciardi 2001).

The Colorado Division of Wildlife (CDW 2011) administers routine fish health sampling at hatchery sites to help slow the spread of *M. cerebralis* infections by early detection. At the Roaring Judy Hatchery, a project is underway to install an ultraviolet system that kills *M. cerebralis* spores (CDW 2011). Treating water with 2537Å UV at doses of 35mWs/cm² can be 86-100% effective at preventing whirling disease in Rainbow Trout fry (Hoffman 1974) and administering 1,300 mWs/cm² of UV under a static collimated beam, can inactivate 100% of the TAM spores present (Hedrick et al. 2000).

There is evidence that electricity (1,000 s exposure to low-level DC voltage for 48 hrs) can destroy *T. tubifex* in aquaculture (R. Ingraham and T. Claxton, pers. comm. in Wagner 2002). Electrical charges of 1-3 kV pulsed 1-25 times at 99 µsec/pulse is effective at killing large numbers of TAM spores (Wagner 2002). Exposing myxospores to 90°C water for 10 minutes is also effective at destroying the spores (Hoffman and Markiw 1977).

Experiments by Hoffman (1974) have demonstrated that filtration is not an effective method for removing TAM spores from water – due to the small spores size, the filter needed to remove them slows flow to rates unacceptable for most applications.

Chemical

Hatchery intake water treated with chlorine (0.5 ppm) administered at two hour intervals once a week can reduce infection rates in Rainbow Trout by 63-73% without causing harm to the fish (Markiw 1992). Supply water treated with calcium cyanide (488 g/m²) mixed with chlorine gas (300 ppm) can be very effective at destroying *M. cerebralis* spores (Hoffman and Dunbar 1961). Water treated with chlorine (130-260 ppm) for 10 minutes may kill 100% of TAM spores present (Wagner 2002), and treating with chlorine (5,000 ppm) for 10 minutes is sufficient enough to destroy both triactinomyxon and myxospore (E. MacConnell, pers. comm. in Wagner 2002). Treating fry with chlorine (10 ppm) for 30 minutes may prevent whirling disease infection (Hoffman and O'Grodnick 1977).

It has been demonstrated that feeding Rainbow Trout with pellets containing (0.1%) Fumagillin is effective at reducing whirling disease infection. Two groups of Rainbow Trout were administered pellets from days 14-64 and 30-160 post infection. Approximately 10-20% of the medicated fish harbored spores, whereas 73-100% of non-medicated fish harbored spores (El-Matbouli and Hoffman 1991).

Earthen pond substrate treated with quicklime (CaO) at concentrations >380 g/m² for two weeks prior to introducing fish can prevent whirling disease infection by destroying *M. cerebralis* spores (Hoffman and Hoffman 1972).

Other

Adherence to local laws regarding transportation of live fish between bodies of water, contacting

local agencies immediately upon noticing signs of whirling disease, properly disposing of fish and fish parts, and not transporting mud on boots and shoes between bodies of water are useful in controlling the transmission of *M. cerebralis* spores in the wild.

Psammonobiotus communis

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

Psammonobiotus dziwnowi

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

Psammonobiotus linearis

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

Sphaeromyxa sevastopoli

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

***Trypanosoma acerinae* Brumpt, 1906**

Regulations (pertaining to the Great Lakes region)

There are no known regulations for this species.

Control

There are no known biological, physical or chemical control methods for this species.

A.16 Viruses

***Novirhabdovirus* sp. genotype IV sublineage b**

Regulations (*pertaining to the Great Lakes region*)

Transportation of Viral Hemorrhagic Septicemia (VHS)-susceptible species out of New York, Pennsylvania, Ohio, Michigan, Indiana, Illinois, Wisconsin, Minnesota, Ontario, and Quebec is prohibited unless certain conditions are met (USDA APHIS 2008). International movement of VHS-susceptible species from the two infected Canadian provinces to the United States is permitted if the shipment meets certain requirements and is imported under an APHIS permit for direct slaughter, or during catch and release fishing activities (USDA APHIS 2008).

Movement of VHS-susceptible species between VHS-infected or at risk states is permitted as long as fish are sent directly to state-inspected slaughter facilities that discharge waste water to a municipal sewage system that includes disinfection, or discharge to a non-discharging pond or a settling pond that disinfects according to all applicable United States Environmental Protection Agency and state regulatory criteria, and are accompanied by a valid VS 1-27 ([Permit for Movement of Restricted Animals](#)) form issued by an APHIS area office. Remains from slaughter facilities must be rendered or composted (USDA APHIS 2008).

Interstate movement of VHS-susceptible fish from VHS-infected or at risk states to non-infected states is permitted as long as the fish are accompanied by appropriate state, tribal, or federal documentation stating the fish have tested negative for the virus (USDA APHIS 2008). Movement of VHS-susceptible species to state, federal, or tribal authorized research and diagnostic facilities is also permitted provided that the fish are accompanied by a valid VS 1-27 form issued by an APHIS area office and the remains are disposed of as medical waste adhering to all applicable United States Environmental Protection Agency and state regulatory criteria (USDA APHIS 2008).

New York, Pennsylvania, Ohio, Michigan, Indiana, Illinois, Wisconsin, and Minnesota have instated similar baitfish regulations to control the spread of VHS and other fish pathogens. Those of New York include that bait harvested from inland waters for personal use is only permitted to be used within the same body of water from which it was taken and cannot be transported overland (with the exception of smelt, suckers, Alewives, and Blueback Herring). Once transported, baitfish cannot be replaced to its original body of water (NYSDEC 2012a).

Live or frozen bait harvested from inland New York waters for commercial purposes is only permitted to be sold or possessed on the same body of water from which it was taken and cannot be transported over land unless under a permit and or accompanied by a fish health certification report. Bait that is preserved and packaged by any method other than freezing, such as salting, can be sold and used wherever the use of bait fish is legal as long as the package is labeled with the name of the packager-processor, the name of the fish species, the quantity of fish packaged, and the means of preservation (NYSDEC 2012a).

Certified bait may be sold for retail and transported overland as long as the consumer maintains a copy of a sales receipt that contains the name of the selling vendor, date sold, species of fish sold, and quantity of fish sold. Bait that has not been certified may still be sold but the consumer must maintain a sales receipt containing the body of water where the bait fish was collected and a warning that the bait cannot be transported by motor vehicle. Bait sold for resale require a fish health certification along with a receipt that contains the name of the selling vendor, date sold, species of fish sold, and quantity of fish sold, which must be kept for 30 days or until all bait is sold (NYSDEC 2012a).

In addition to baitfish protections, prior to placing fish in New York waters, a fish health certification report must document that the fish are VHS free.

Ontario has implemented management zones to help slow the spread of VHS. Commercial bait operators are prohibited from moving live baitfish out of the VHS Management Zone and live or dead bait in or out of the Lake Simcoe Management Zone. Salmon and trout eggs may be collected from virus-positive waters only if eggs are disinfected according to the Ministry's protocol. Walleye spawn collection is permitted as long as the fish are stocked back into virus-positive waters. Fish and eggs are permitted to be stocked in waters that are not virus-positive only if the facilities are certified VHS free (OMNR 2011).

Control

Biological

There are no known biological control methods for this species.

Physical

Multiple means of control are available to fish hatchery managers, including treatment of water with UV light subtype C (280-200nm wavelength) irradiation and heat (>15°C) (McAllister 1990), exposure to pH levels lower than 2.5 or higher than 12.2, desiccation of tanks and equipment (CFSPH 2007), minimization of stressors, cessation of water flow to adjacent waterways, and establishment of quarantines (Warren 1983, CFSPH 2007). Furthermore, exposure to VHS can be prevented through use of spring water, specific pathogen free (SPF) stock, and separate cultivation of salmonids and flatfish (CFSPH 2007).

As with other hitchhiking aquatic species, boaters and anglers are encouraged to clean and disinfect their gear (Bakal 2012), as well as to completely drain bilges and live wells before moving between bodies of water (OMNR 2012).

Chemical

The VHS virus is sensitive to ether, chloroform, glycerol, formalin, iodophor, sodium hydroxide, and sodium hypochlorite, which can be used as disinfectants (CFSPH 2007 McAllister 1990). No effective anti-viral agents or commercial vaccines exist (CFSPH 2007). Disinfection of live wells and other contaminated equipment can be accomplished with a 10% household bleach/water solution (e.g., 100 ml of household bleach to 900 ml of water). Waste water should be discarded away from any water body. Virkon® S is another widely available disinfectant (OMNR 2012).

Other

The United States Fish and Wildlife Service recommend implementation of the International Hazard Analysis and Critical Control Point (HACCP) planning standard to prevent the spread of VHS (Bakal 2012).

VHS should be reported to Area Veterinarians in Charge (AVIC) or state veterinarians immediately upon diagnosis or recognition of the disease. Fish health surveillance programs and fallowing are also useful methods of control (CFSPH 2007).

Ranavirus

Regulations (*pertaining to the Great Lakes region*)

Ohio requires out-of-state source facilities of live fish to provide health inspection and testing documentation prior to importation (NCRAC 2010ab). Michigan requires imported aquacultured fish to be accompanied by either an official interstate health certificate, official interstate certificate of veterinary inspection, or a fish disease inspection report. Importing aquacultured fish from source facilities with a record of an emergency disease within the past two years is prohibited. Fish imported from non-Michigan source facilities and intended for stocking in public waters must be certified free of Largemouth Bass Virus (LMBV) (NCRAC 2010ab). Illinois requires source facilities of any species of live fish, eggs, and sperm to document they are disease free prior to importation (NCRAC 2010ab). Wisconsin requires source facilities to document fish health inspections prior to importing live fish and eggs (NCRAC 2010ab).

Control

Biological

There are no known biological control methods for this species.

Physical

There are no known physical control methods for this species.

Chemical

Disinfection of live wells and other contaminated equipment can be accomplished with a 10% household bleach/water solution (i.e., 100 ml of household bleach to 900 ml of water). Waste water should be discarded away from any water body. Virkon® S is another widely available disinfectant (OMNR 2012).

Other

There is no known cure or effective treatment of LMBV infection (Syska et al. 2012).

Rhabdovirus carpio

Regulations (*pertaining to the Great Lakes region*)

Importation of live fish, fertilized eggs, and gametes of Spring Viremia of Carp Virus (SVCV)-susceptible species including Common Carp (*Cyprinus carpio*), Koi (*C. carpio koi*), Grass Carp (*Ctenopharyngodon idella*), Silver Carp (*Hypophthalmichthys molitrix*), Bighead Carp

(*Aristichthys nobilis*), Crucian Carp (*Carassius carassius*), Goldfish (*Carassius auratus*), Tench (*Tinca tinca*), Orfe (*Leuciscus idus*), and Sheatfish (*Silurus glanis*) is permitted, provided they are accompanied by a United States Department of Agriculture import permit and a veterinary health certificate (USDA APHIS 2012).

Control

Biological

Single-stranded and double-stranded RNA injections can provide *Rhabdovirus carpio* protection for up to three weeks (Alikien et al. 1996, Masycheva et al. 1995).

Physical

Establishment of quarantines, culling, and stock density reduction during the winter and spring are beneficial management practices to prevent the spread of Spring Viremia of Carp Virus (SVCV) (CFSPH 2007). *R. carpio* is inactivated by UV irradiation (254 nm), gamma irradiation (103 krad), heating to 60°C for 30 minutes, and exposure to pH 12 for 10 minutes, or pH 3 for three hours (CFSPH 2007, OIE 2009).

Chemical

Disinfection of facilities and equipment will prevent the spread of SVCV in aquaculture (CFSPH 2007). *R. carpio* is susceptible to oxidizing agents like sodium dodecyl sulphate, non-ionic detergents, and lipid solvents. The virus is inactivated by formalin (3%) for five minutes, chlorine (500 ppm), iodine (0.01%), NaOH (2%) for ten minutes, benzalkonium chloride (100 ppm for 20 minutes), alkyltoluene (350ppm for 20 minutes), chlorhexidine gluconate (100 ppm for 20 minutes), and cresol (200 ppm for 20 minutes) (Ahne and Held 1980, Ahne 1982, Fijan 1999, CFSPH 2007, Kiryu et al. 2007). Methisoprinol may be useful by inhibiting replication of SVCV in vitro. Further testing under culture conditions is necessary (Siwicki et al. 2003).