U.S. Department of the Interior
National Park Service

Great Lakes Network Office
Ashland, Wisconsin

Strategic Approach to Problem Identification and Monitoring of Aquatic Invasive Species Within the Great Lakes Inventory and Monitoring Network Park Units

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Great Lakes Network Report GLKN/2007/08 **September 2007**

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September 27, 2007

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Ashland Fishery Resources Office
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Final report to the National Park Service, Great Lakes Network Office
Interagency Agreement Number: F2105-04-0029

Great Lakes Network Report
GLKN/2007/08

Suggested citation:
Quinlan, H., M. Dryer, G. Czypinski, J. Pyatskowit, and J. Krajniak. 2007. Strategic approach to problem identification and monitoring of aquatic invasive species within Great Lakes Inventory and Monitoring Network park units. U.S. Fish and Wildlife Service, Ashland Fishery Resources Office, Ashland, Wisconsin, for National Park Service, Great Lakes Inventory and Monitoring Network, Ashland, Wisconsin. GLKN/2007/08.

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Acknowledgements

Information was compiled from many sources in the preparation of this document. The following individuals provided valuable information or steered us toward resources that greatly helped with this project; Steve Yancho (Sleeping Bear Dunes National Lakeshore); Julie Van Stappen (Apostle Islands National Lakeshore); Lora Loope (Pictured Rocks National Lakeshore); Brad Welch, Brenda Lafrancois, and Jay Glase (National Park Service); Ron Martin, Steve Schram, Terry Margenau, Scott Toshner, and Nancy Nate (Wisconsin Department of Natural Resources); Steve Geving, Kevin Peterson, Dave Zappetillo, and Roger Hugil (Minnesota Department of Natural Resources); Amy Benson (USGS Florida Integrated Science Center); Miles Falk and Dara Olson (Great Lakes Indian Fish and Wildlife Commission); Mark Edlund (St. Croix Watershed Research Station); Gary Kohlhepp and Bill Taft (Michigan Department of Environmental Quality); Phil Schneeberger and Shawn Sitar (Michigan Department of Natural Resources); Gary Cholwek (USGS Lake Superior Biological Station); Seth Moore and Margaret Watkins (Grand Portage Band of Lake Superior Chippewa); Dr. Jill Leonard (Northern Michigan University); Matt Heiman (Leelanau Conservancy); Mike Litch (Glen Lake Association); Brian Biebert (Indiana Department of Natural Resources); Dr. Stacy Daniels (Crystal Lake Watershed, Michigan); Vanlandschoot’s Fishery, Munising, Michigan; and staff from the Great Lakes Fishery Commission. Thanks to Joan Elias and Ulf Gafvert of the National Park Service Great Lakes Inventory and Monitory Network. Ms. Elias provided guidance, review and comments that improved the quality and organization of this document and Mr. Gafvert provided consultation and assistance with GIS and map components of the project. Finally, the authors thank the National Park Service - Great Lakes Inventory and Monitoring Network for funding this project.

Introduction

Great Lakes Inventory and Monitoring Network (hereafter referred to as GLKN) park units are situated in one of the most water-rich locations in the world, and aquatic habitats in the parks are substantial in area and diversity. The GLKN includes portions of three major watersheds (Great Lakes, Mississippi River, Hudson Bay), and comprises nine park units. The geographic scope for this project is shown in Figure 1, hereafter referred to as the project area.

Figure 1. Location of National Parks in the Great Lakes Inventory and Monitoring Network and geographic area included in this strategic approach to aquatic invasive species monitoring.

Great Lakes Inventory and Monitoring Network park units on Lake Superior are Apostle Islands National Lakeshore (APIS), Grand Portage National Monument (GRPO), Pictured Rocks National Lakeshore (PIRO), and Isle Royale National Park (ISRO). On Lake Michigan, park units are Indiana Dunes National Lakeshore (INDU) and Sleeping Bear Dunes National Lakeshore (SLBE). In the Mississippi River watershed, park units are Saint Croix National Scenic Riverway (SACN) located along the border of Wisconsin and Minnesota, and the Mississippi National River and Recreation Area (MISS) located near Minneapolis/St. Paul. Voyageurs National Park (VOYA), located in northern Minnesota adjoining the U.S. border with Canada, is in the Hudson Bay watershed. Description and characteristics of park units are summarized in Appendix 1.

Exotic plant and animal species ranked highest of all GLKN vital signs (Route 2004). Of the exotic plant and animal species known to occur within the GLKN project area, aquatic invasive species (AIS) represent perhaps the most significant and imminent biological threat to aquatic resources. Changes or shifts in animal and plant community composition brought about by the introduction of AIS could have significant implications for native species, most notably through competition for food and habitat, or direct predation. It is therefore vital to monitor water bodies for the presence of these species (Lafrancois and Glase 2005). The intent of vital signs monitoring is to obtain scientifically sound trend information that will have multiple applications for management decision-making, research, education, and promoting public understanding of park resources. We have chosen to focus attention on 12 target aquatic invasive animal and plant species (Table 1).

Table 1. Target aquatic invasive species in Great Lakes Network park units.

Fishes
- ruffe (*Gymnocephalus cernuus*)
- round goby (*Neogobius melanostomus*)
- sea lamprey (*Petromyzon marinus*)
- threespine stickleback (*Gasterosteus aculeatus*)
- white perch (*Morone americana*)

Invertebrates
- fish-hook waterflea (*Cercopagis pengoi*)
- quagga mussel (*Dreissena bugensis*)
- rusty crayfish (*Orconectes rusticus*)
- spiny waterflea (*Bythotrephes longimanus*)
- zebra mussel (*Dreissena polymorpha*)

Plants
- curly leaf pondweed (*Potamogeton crispus*)
- Eurasian water milfoil (*Myriophyllum spicatum*)

Project Objectives

The objectives of this project are to:

- Document current distribution and rate of range expansion for 12 target AIS.
- Identify entities conducting monitoring activities capable of detecting presence of AIS.
- Summarize life history information and impacts of invasion for target AIS.
- Propose three to five AIS to monitor and recommend monitoring methods that:
    - provide early warning of AIS in GLKN park units, and
    - indicate status and trends of select AIS in GLKN park units.

This information will assist the GLKN park units in prioritizing species for monitoring, determining feasibility of control efforts, and in educating park visitors on ways to avoid spreading AIS.

Distribution and Relative Importance of Target AIS

We have summarized recent information on the taxonomy, description, distribution, status, invasion impacts, and control options, and provided reference lists for each of the 12 target species (Appendix 2). All of the target AIS are native to Europe or Asia. Some were introduced decades ago and are now widespread. Others are relative newcomers. All are commonly considered invasive to natural communities. Many are also a nuisance and economically costly to society. None of them were introduced intentionally for recreational, commercial or ecological value.

There continues to be a need to coordinate and communicate with other agencies regarding AIS presence, the importance of monitoring the status and trends of aquatic species in and adjacent to National Park units, the benefit of early warning or detection of AIS in park units, and the need for education and outreach efforts. Since this project was initiated, four AIS previously undetected in most of the Great Lakes have been detected within the project area. These are the Chinese mitten crab (*Eriocheir sinensis*), a freshwater shrimp (*Hemimysis anomala*), a diatom known as Didymo (*Didymosphenia geminata*) (Spaulding and Elwell 2007), and a rhabdovirus known as the viral hemorrhagic septicemiaus virus (Office International of Epizooties 1963). These newest AIS will not be addressed in this report.

Existing AIS Monitoring Programs

Numerous government and non-government entities monitor population levels and distribution of animals and plants within the project area by a variety of methods, and for various reasons. Equally variable are the levels of effort, quality control, and data analysis. Some monitoring is specifically dedicated to determining presence, absence, or trends of AIS, but most of it is not dedicated to AIS-related information. To the extent possible, we provide notification of ongoing aquatic surveys conducted within or extending 7.5 miles (5 nautical miles) outside of GLKN park unit boundaries that are capable of capturing one or more of the 12 target AIS of concern to the GLKN (Appendix 3). Appendix 3 displays agencies and organizations conducting the surveys; contact person(s); survey purpose, frequency, locations, and sampling methods; how the data are archived; and whether the data are subject to quality assurance procedures (QAP). This information was obtained from agency and organization contacts whose names were provided to us by individual park resource specialists.

Below, we provide a description of known survey work conducted outside park unit boundaries, but still within the GLKN watersheds. This information and Appendix 3 established the foundation for our recommendations on which AIS the GLKN park units should monitor, and methods for monitoring those species.

Lake Michigan Watershed

In the fall of 2003, the Great Lakes Commission (GLC) distributed a 25-question survey to 127 entities and/or programs that had the capability to detect and monitor invasive species in high risk areas on Lake Michigan. Analysis of survey results from 46 entities and/or programs (response rate of 36%) has helped in assessing the potential for existing monitoring programs to detect new AIS invasions, as well as monitor their potential spread (Great Lakes Commission 2004).

Only 10 (22%) of the 46 respondents said the purpose of their program was to monitor the introduction, spread, range expansion, distribution, and/or abundance of specific AIS.

Because industrial ports are high risk locations for introduction of AIS due to the large volume of shipping traffic, many respondents are focusing their monitoring efforts in these areas. All of the major ports are being monitored to some degree, including Burns Harbor, Chicago, Green Bay, Ludington, Menominee, Milwaukee, Muskegon, Escanaba, Port of Indiana, Gladstone, Sturgeon Bay, Calumet River, Benton Harbor, Manistee, and Grand Traverse Bay.

The frequency of monitoring varies widely from daily to annually. Some sampling is not conducted routinely, but rather as “discoveries are made,” “as time allows,” “depending on the weather,” “depends on the site,” or “as needed for program implementation.” Survey results indicate that there is a need to coordinate related monitoring temporally in order to make better comparisons and use of the data.

Respondents were asked if some type of a quality assurance plan or procedure is in place that requires elements such as performance/measurement criteria for information collected, description and justification of sample design strategy, and equipment calibration. Although 39% of respondents indicated that they do have such plans in place, an almost equal amount (35%), do not.

The majority of monitoring data (70%) are reported to state agencies. Additionally, reporting monitoring data to the general public was a practice of nearly half of survey respondents (46%), while reporting to a federal agency was roughly the same (43%). Because it is important to inform the general public in a timely manner and to enlist their assistance in preventing the spread of AIS, more sampling programs should report monitoring data to the general public.

Thus, results indicate the need for a more coordinated, consistent approach to the reporting of AIS monitoring data.

The majority of monitoring programs (63%) are collecting fish-related data. Results also indicated that many respondents (39%) collect information on aquatic invertebrates. Nearly one-third of survey respondents (33%) indicated that their monitoring programs are collecting AIS information. Other areas where there are potential linkages with AIS monitoring include those collecting water quality information such as physical (30%) and chemical data (26%), and collection of vegetation data (20%). There were “other” responses indicating specific collection of phytoplankton, zooplankton, and algae.

The following eight AIS were identified as those being actively monitored by one or more entities in the Lake Michigan basin. The five species noted with an “*” are also on the list of target AIS species for this report:

- sea lamprey*
- white perch*
- gizzard shad
- alewife
- carp
- Eurasian water milfoil*
- zebra mussel*
- quagga mussel

Because many sampling methods do not discriminate, several respondents reported that their program is capable of detecting and monitoring for a variety of different species. Sampling equipment such as plankton nets, trawling gear, and an array of other nets are non-species specific, allowing for open-ended collection of AIS. Several of the programs that monitor for new introductions of invasive species are programs that monitor for ecological and species composition changes.

The US Fish and Wildlife Service’s Ludington Biological Station (Sea Lamprey Control Program), in cooperation with the Grand Traverse Band of Ottawa and Chippewa Indians (GTB), the Little River Band of Ottawa Indians (LRBOI), and private contractors, annually traps adult sea lamprey in 12-20 Lake Michigan tributaries to assess abundance. The Ludington Biological Station also conducts annual assessments for detecting, evaluating, estimating, and/or delineating larval sea lamprey populations in 70-90 tributaries to Lake Michigan using back pack electrofishing equipment, portable assessment traps, mechanical traps, and fyke nets.

Lake Superior Watershed

On Lake Superior, the US Fish and Wildlife Service’s Ashland Fishery Resources Office (Ashland FRO) conducts annual assessments to locate new populations of ruffe and describe their age and/or size composition. The assessments are conducted in tributary estuaries and embayments on the periphery of the known distribution range for ruffe, and in or near shipping ports where ruffe could be introduced by ballast water from inter- and intra-lake shipping. A 4.9 m-wide bottom trawl is towed spring and fall at each sample site. In addition to trawling, other gear types including gill nets, seines, fyke nets, and experimental perch traps (called modified Windermere traps) (Edwards et al. 1998) are used.

The Ashland FRO also conducts bottom trawling and seining in four south shore tributaries of Lake Superior (Amnicon, Iron, Flag, and Ontonagon rivers) as part of a long-term effort to monitor the relative abundance of ruffe and native species. This monitoring consists of three cycles (spring, summer, fall), and the number of tows and seine hauls varies to cover the lower area of the rivers from the mouth up to 3 km upriver.

The US Fish and Wildlife Service's Marquette Biological Station (Sea Lamprey Control Program) in cooperation with the Great Lakes Indian Fish and Wildlife Commission annually trap adult sea lamprey in 15-20 Lake Superior tributaries to assess abundance. The Marquette Biological Station also conducts annual assessments for detecting, evaluating, estimating, and/or delineating larval sea lamprey populations in 40-60 tributaries to Lake Superior using back pack electrofishing equipment, portable assessment traps, mechanical traps, and fyke nets.

The Lake Superior Biological Station of the US Geological Survey (LSBS) conducted bottom trawling in the St. Louis River estuary from 1988 through 2004 as part of a long-term effort to monitor the relative abundance (number per hectare (ha)) of ruffe and native species. The primary gear used was a 4.9 m-wide bottom trawl, towed spring, summer, and fall for five minutes at 40 randomly selected stations.

Since 1978, the LSBS has annually conducted bottom trawling during May and June at 88-94 stations established systematically around the coastal waters of Lake Superior, including the United States and Canada, to assess spring fish community abundance. The primary gear is an 11.9 m-wide bottom trawl with a 6 mm square mesh cod end. Over the period of 1989-2004, zooplankton samples for Lake Superior were typically taken annually at a subset of trawling stations. Zooplankton were sampled with a 50 cm diameter, 63 µm mesh conical plankton net, which was towed once vertically from approximately 1 m off the bottom to the surface at the deep end of a trawl station. Water column depth varied from 30 to >140 m.

In 2005, six agencies from the U.S. and Canada coordinated efforts and shared resources to collect samples of organisms from microscopic plankton to fish in an effort to gain a thorough understanding of Lake Superior's lower food web. Sampling took place during spring, summer and fall. The sampling stations were dispersed across large geographic regions and will thus capture variability at this scale in addition to nearshore/offshore gradients. Objectives of the sampling were to describe seasonal biomass and abundance densities of phytoplankton, zooplankton, *Mysis*, (a free swimming crustacean) and *Diporeia* (a bottom-dwelling crustacean) across the lake, and to determine the production of these trophic levels, if possible. Nearly 1,500 samples were collected including 776 for zooplankton (animal plankton), 298 targeting *Mysis*, and 411 bottom samples. Sampling for zooplankton takes place during the daytime, whereas *Mysis* samples are collected at night. Information from this sampling will guide future efforts to establish a long-term cooperative monitoring program.

Upper Mississippi River

Fish, macroinvertebrates, and vegetation in the commercially navigable reaches of the Upper Mississippi River have been monitored for 10 or more years under the protocols of the Long-Term Resource Monitoring Program (LTRMP). The LTRMP was authorized under the Water Resources Development Act of 1986 (Public Law 99-662). The long-term goals of the LTRMP are to understand the system, determine resource trends and impacts, develop management alternatives, manage information, and develop useful products for decision makers to maintain the Upper Mississippi River System as a sustainable large river ecosystem. Detection of AIS is not the focus of the LTRMP, but the sampling protocols may incidentally detect AIS invasions.

The LTRMP is being implemented by the Environmental Management Technical Center, a U.S. Geological Survey Science Center, in cooperation with the five Upper Mississippi River System (UMRS) States of Illinois, Iowa, Minnesota, Missouri, and Wisconsin. The U.S. Army Corps of Engineers provides guidance and has overall Program responsibility.

Fish monitoring activities focus on single species and community status and trends. Some sampling gear targets specific species, while other sampling gear captures particular community types (e.g., benthivores, forage fish, etc.). LTRMP fish data provide critical information on the occurrence, establishment, distribution, and abundance of non-native fish species (Gutreuter et al. 1995). For example, silver carp (*Hypophthalmichthys molitrix*) and bighead carp (*H. nobilis*), referred to generically as Asian carp (*Hypophthalmichthys* spp.), were introduced as the result of aquaculture activities in the Mississippi River basin, and have recently established themselves in the lower reaches of the UMRS. Data from LTRMP have been critical in documenting the spread of these prolific species within the UMRS and towards the Great Lakes drainage.

Macroinvertebrates such as mayflies (Ephemeridae), fingernail clams (Sphaeriidae), midges (Chironomidae), the non-native Asiatic clam (*Corbicula* spp.), and the zebra mussel (*Dreissena polymorpha*), are monitored for their ecological significance in the food web or because they are recent non-native invaders to the UMRS. Rather than taking a community approach, macroinvertebrate monitoring for the LTRMP is designed to focus on abundance trends in select UMRS macroinvertebrates, largely because of the sampling logistics, and funding levels. Mayflies, fingernail clams, and midges were chosen because they play an important ecological role in the UMRS. The Asiatic clam and the zebra mussel were chosen for sampling because of their possible detrimental effects on the economy and biology of the UMRS (Thiel and Sauer 1999).

Long-term monitoring of aquatic vegetation was initiated under the LTRMP in 1991 with the primary objective of determining trends in submerged and rooted floating-leaf vegetation in the UMRS. Aquatic vegetation is monitored by point collection procedures and recorded by species. Data collected by these procedures can be used to quantify the abundance of individual species at each site, as well as over large areas where many sites have been surveyed at random (Yin et al. 2000).

In 1998, a stratified random sampling protocol was initiated for aquatic vegetation (Yin et al. 2000) to allow for estimation of poolwide means. After 3-year concurrent sampling periods, wherein both protocols were followed, transect sampling was discontinued and only stratified random sampling has been conducted since 2001.

Other monitoring programs for animals, plants and/or invertebrates have been in existence at various levels of thoroughness by federal, state, tribal and local government agencies; industries; conservation organizations; citizen groups; educational institutions; and others over various time periods and geographic areas.

St. Croix River

Professionals from the Wisconsin Department of Natural Resources, the University of Wisconsin, counties, high school, and interest groups are working with citizens to monitor the health of the St. Croix River in Polk County. Six parameters (temperature, dissolved oxygen, stream flow, turbidity, habitat, and biotic index) are included as part of the monitoring program. The biotic index is generally assessed twice a year, spring and fall. Citizens collect macroinvertebrates from the stream and separate them into groups of similar-looking organisms. Some AIS, such as zebra mussels and rusty crayfish, could be detected in this monitoring, although additional training of volunteer collectors on species identification would be necessary.

Monitoring of freshwater mussel communities of the Saint Croix National Scenic Riverway began in 1988 when five locations on the St. Croix and Namekagon rivers were sampled (Heath and Rasmussen 1990). During 1995 and 1996, four of the five monitoring sites were re-sampled (Doolittle and Heath 1997, Doolittle et al.1995). In 1995, 1996, and 1998, researchers randomly placed 1 m^2 quadrat plots, counted living and dead unionids, margaritiferids, Asian clams (*Corbicula fluminea)*, and zebra mussels, measured and aged them, and determined gravidity. In addition to quadrat sampling, researchers randomly collected larger numbers of mussels (relative abundance collections) to complement comparisons of relative abundance, and age and total length distributions, between years and sites.

As part of the baseline monitoring strategy for non-wadeable rivers in Wisconsin, the Wisconsin Department of Natural Resources (WIDNR) sampled fish in the lower St. Croix River during the 1999 and 2000 field seasons. The purpose of this survey was to develop a baseline inventory of the existing fishery resources in the lower St. Croix River and to make recommendations for future fisheries management activities. In addition, the study was used to develop standardized methods and procedures for monitoring non-wadeable rivers in Wisconsin (Benike and Michalek 2001). Two stations were established and sampled in 1999 and 2000. One station near Marine on St. Croix, Minn. (RM 34.0) and the other near St. Croix Falls (RM 52.0).

Large fish were collected using two-pulsed DC mini-boomshockers during daylight hours. Small fish were collected using a DC mini-streamshocker and three 50-foot seine hauls. The WIDNR proposes to continue long-term trend monitoring on the lower St. Croix fish community. Trend information will allow local management staff to determine if the native fish community is stable, improving, or decreasing through time following the non-wadeable baseline monitoring protocol.

Description of Monitoring Approach

Species to Monitor

We developed a ranking and evaluation method to identify three to five AIS that should be monitored within individual or multiple GLKN park units. These target species are currently expanding their ranges and have the potential to invade or be introduced into park unit waters. Based on the current rate of new introductions of aquatic organisms to the Great Lakes and the potential for additional AIS, we expect that species targeted for monitoring will change over time.

For example, New Zealand mud snails have been collected in Thunder Bay and the Duluth-Superior harbor in Lake Superior. New Zealand mud snails are small (3 mm) and susceptible to human assisted spreading by attachment to chest waders and hip boots. Voyageurs and Isle Royale Nat'l. Parks, Grand Portage Natl. Monument, Apostle Islands Natl. Lakeshore, and possibly the St. Croix Natl. Scenic Riverway are at risk for invasion by New Zealand mud snails. Benthic invertebrate sampling gear used to monitor other species should be capable of capturing this species. Analysts examining substrate samples collected from the park units at risk should be aware of the potential presence of New Zealand mud snails.

Our ranking and evaluation method involved five questions and assignment of a value from 1-10 for each question. Each of the 12 AIS addressed in this report was put through the five questions for each park unit. Within each park unit we summed the values for each question to arrive at a total species score. Our questions considered likelihood of invasion, magnitude of ecological impact, and cost to obtain meaningful monitoring data within each GLKN park unit. Independent of the species ranking process we also asked two "yes or no" questions of each species to reveal which ones are currently being monitored to adequately detect presence and ecological impact.

The following five questions were used to rank the 12 AIS and determine the three to five priority species for each park unit.

1. How great is the likelihood that a subject species will invade a subject GLKN park unit based on the habitat available and rate of current spread and distribution, if not now present? 1 = Low, 10 = Great. If currently present, criterion value = 0.
2. After invasion, what is the likelihood of significant ecological impact to native plant and animal communities? 1 = Low, 10 = Likely. If currently present and stable, criterion value = 0.
3. How difficult will it be to monitor for early detection, if not now present? 1 = Difficult, 10 = Easy.
4. How difficult will it be to monitor ecological impacts of invasions upon native plant and animal communities? 1 = Difficult, 10 = Easy.
5. How expensive, relative to current budgets, might an early detection or monitoring program be? 1 = Prohibitive, 10 = Manageable.

These two additional questions relating to current monitoring and knowledge of ecological impacts were also asked.

1. Is sufficient monitoring currently underway to determine early detection or ecological impacts within a subject GLKN park unit? Yes or No.

2. Is sufficient monitoring currently underway elsewhere to assume early detection or ecological impacts within a subject GLKN park unit? Yes or No.

The values assigned to each AIS based on its potential to invade a GLKN park unit and rate of spread (Question 1) were gleaned from mapping the range and current distribution into a spatial (GIS) database. The database was then queried to develop data layers for the years 1985, 1995, and 2005, for each species for which data were available. Examples of time series maps for curly leaf pondweed and ruffe, and accompanying metadata are displayed in Appendix 4. Information on current abundance within the project area, and significance of invasion impact on native species and habitats is described in the species profiles in Appendix 2. Values depicting relative cost and ability to monitor were subjectively predicted based on our professional knowledge of the species habits and occurrence, sampling methods, and current funding levels.

The priority AIS to be monitored are signified by the letter P in Table 2. The same gear and sampling methods used to monitor priority (P) species/target AIS will incidentally capture other biota noted in Table 2.

Our concluding recommendation for which AIS to monitor within which park unit (Table 2) is not based entirely on the numeric score from the criteria questions because in some instances our professional judgment drew us to a different conclusion. Species' scores and rationale are detailed in Appendix 5. Other factors and assumptions follow.

Table 2. Priority (P) species to monitor in each GLKN park unit.

	Lake Superior Units				Lake Michigan Units		Inland Units		
	APIS	GRPO	ISRO	PIRO	SLBE	INDU	SACN	MISS	VOYA
Fishes									
ruffe		P	P	P	P				
round goby	P			P		P			
sea lamprey									
threespine stickleback					P				
white perch		P	P		P	P	P	P	
Invertebrates									
rusty crayfish	P	P	P	P	P	P	P	P	P
quagga mussel	P	P	P				P	P	P
zebra mussel	P	P	P	P					P
fish hook waterflea									
spiny waterflea									
Plants									
curly leaf pondweed									
Eurasian water milfoil									P

General Rating Factors

- The ecological impact of the spiny and fishhook waterfleas is not well documented and their abundance tends to be inconsistent. Because they are difficult and expensive to monitor, they tend to rank lower in priority than the other AIS.
- Threespine stickleback is either present or possesses a high potential to invade all GLKN park units, but its ecological impact is not well documented; it is assumed that it will compete for food and space with native sticklebacks. Therefore, threespine stickleback tends to rank in the medium priority range. It should be noted that baited trap nets set for ruffe and rusty crayfish are also capable of capturing threespine stickleback if the netting is constructed of a small (<6 mm) stretch mesh.
- Round goby has only been confirmed in three locations in Lake Superior; in two of those locations, only one specimen has been captured. In addition, there may be a link between distribution and abundance of round goby and the distribution and abundance of zebra mussel. In Lake Superior, zebra mussels are only established in the Duluth-Superior harbor. Although abundant in coastal Lake Michigan and a known egg consumer, the ecological impacts of round goby are not consistent in all locations in Lake Michigan. Therefore, round goby tends to rank medium in priority.
- Ruffe and white perch were considered equal in terms of impact and significance. Ruffe and white perch can be captured with the same gear (trawls, traps, gill nets), which are in the medium range of expense. Ruffe and white perch can co-inhabit the same coastal sites and have intermittent distributions in the Great Lakes; both are abundant in some locations. Therefore, ruffe and white perch tend to rank between medium and high in the priority range.
- The two invasive plants, curly leaf pondweed and Eurasian water milfoil, are widely distributed. Both species are known to have negative ecological impacts and have high potential to expand their distribution. Curly leaf pondweed can survive under ice, but Eurasian water milfoil cannot and has to regenerate when the water warms. Therefore, curly leaf pondweed has a growth advantage over Eurasian water milfoil, and tends to score higher in priority than Eurasian water milfoil in some of the GLKN park units. Both curly leaf pondweed and Eurasian water milfoil are either present within some of the GLKN park units or likely to invade some of the parks. Cost of monitoring may vary depending on the extent of invasion. Curly leaf pondweed and Eurasian water milfoil tend to rank from medium to high in priority, and this range in ranking was established primarily by risk of invasion.
- Zebra mussel, quagga mussel, sea lamprey and rusty crayfish have broad distributions in the Great Lakes Basin. Their ecological impacts are significant and recognizable. Generally, they are easy and inexpensive to monitor. The ranking score represents risk of invasion. These species tend to rank from medium to high in the priority range.
- Sea lamprey completely colonized the Great Lakes more than four score years ago. Since that time it has had a significant ecological impact. Because extensive monitoring and control is currently being conducted, sea lamprey was not chosen for monitoring.

Monitoring Principles and Assumptions

We will define field techniques and species to monitor based on the following concepts and assumptions:

- Field sampling techniques used to monitor ecological and/or species composition changes will also provide data on priority AIS species.
- Field sampling techniques used for priority species may also collect data on other AIS species.
- A monitoring technique that collects useful information on a number of species will yield a more complete portrait of an area's status than does information for a single species.
- Additional sampling with different gear can often be conducted simultaneously with little additional effort and cost.
- The majority (75-80%) of sample sites will be at locations where early invasions are most likely to be detected.
- The remainder (20-25%) will be randomly selected.
- Our reported occurrence information on AIS is often based on incidental catch, meaning that we probably do not have good data on actual occurrence across the project area.
- Other than for sea lamprey, no monitoring programs are currently being conducted that will detect statistically significant change over time for specific AIS in GLKN park units.
- Some monitoring currently underway by others can fill data needs or gaps.
- Monitoring design needs periodic evaluation and needs to be adaptive to new measurement technologies and evaluation methods.
- Counts of adult and juvenile animals will reveal if AIS are reproducing.
- Sampling methods and types of data may or may not vary by species group (Table 3).

Table 3. Common sampling methods and data products for different species groups.

Species Group (species)	Potential Data Collected	Data Collection Method
Fishes (ruffe, threespine stickleback, white perch, round goby, sea lamprey)	Presence/absence, proportion of area occupied (PAO), catch rates (CPE), age class	Trawl, seine, trap, gillnet, electrofishing
Zooplankton (spiny waterflea, fishhook waterflea)	Presence/absence, proportion of area occupied (PAO), density per volume filtered	Plankton tow
Mussels (zebra mussel, quagga mussel)	Presence/absence, proportion of area occupied (PAO), density per m^2 bottom area, age class, density per volume filtered (veligers)	Plankton tow, plate samplers, underwater visual census (SCUBA)
Crayfish (rusty crayfish)	Presence/absence, proportion of area occupied (PAO), density per m^2 bottom area	Underwater visual census (SCUBA), crayfish traps, trawl
Aquatic vegetation (curly leaf pondweed, Eurasian watermilfoil)	Presence/absence, proportion of area occupied (PAO), density per m^2 bottom area	Underwater (SCUBA) and surface water visual census, rake

Implementation Strategy

Procedures for Monitoring Current Status and Early Detection of AIS

The following sections describe recommendations for monitoring, sampling design, and requirements for the species identified as “priority species” in each of the Great Lakes Parks: ruffe, white perch, round goby, threespine stickleback, rusty crayfish, zebra and quagga mussels (together referred to as *Dreissena*), and Eurasian water milfoil.

The monitoring designs focus on early detection - detecting the presence of the species and its relative abundance in terms of the sampling gear employed.

Monitoring Target

Ruffe, white perch, round goby, and threespine stickleback with potential to capture rusty crayfish, *Dreissena*, and Eurasian water milfoil on mud/sand substrate with minimal bottom obstructions.

Gear: The use of a bottom trawl, commercially referred to as a semi-ballon trawl, and commonly referred to as a shrimp trawl is recommended. Trawling is not recommended to target rusty crayfish, *Dreissen*a, and Eurasian water milfoil specifically, but is capable of collecting these species while targeting the priority fish species. The trawl is towed by a small vessel no longer than 6 m for ease in navigating narrow streams and shallow waters. For ease of handling, the trawl headrope should not exceed 5.0 m in length. Other trawl parameters include a 3.8 cm stretch-mesh body, a 31.8 mm stretch-mesh cod end, and a 6 mm or less stretch-mesh inner liner to hold small specimens. The bottom trawl can be deployed and retrieved manually, or deployed with a winch in free-spool mode and retrieved hydraulically. If trawl deployment and retrieval is mechanical, then the towing cable can consist of a 4.7 mm diameter stranded steel cable terminated by two 3.9 mm diameter stranded steel cables (called “bridle cables”), 7.6 m to 15 m in length. Each bridle cable is attached to a 375 mm x 750 mm spreader (commonly referred to as a “door”). The two spreaders are attached to the lead ropes of the trawl. The towing line is guided to the winch by a boom that subtends over the cage (metal cover that protects the outboard engine(s) and directs the towing line a safe distance from the propellers of the outboard engines. If trawling manually, the tow line and bridle lines should be a soft variety rope, at least 15 mm in diameter for ease of handling. Also for ease of handling, the doors can be downsized to 300 mm x 600 mm, but avoid using smaller doors at depths greater than 3 m.

Procedure: Bottom trawling is normally conducted during daylight hours. Plan a sufficient number of tows to adequately sample a location, usually a minimum of three**.** Target time for duration of tow is 5 min, but may vary depending on the size of the area to be trawled, the presence of submerged obstacles, and numbers of fish captured. The minimum time for a valid tow is considered to be 2 min. Tow speed is maintained at approximately 3 km/hour, and monitored by engine tachometer readings, speedometer, or global position system (GPS) devices. Start and end location coordinates are recorded for each tow. Bottom water temperature is recorded prior to each established tow, except when consecutive tows are conducted in close proximity to each other. Depth is recorded at the start and finish of individual tows and then averaged to determine the mean depth for each tow. Tows are directed along or across contours, with the majority along contour. Catches of fish are sorted by species and counted, and the total length of up to 50 specimens of each species is measured to the nearest millimeter. All captured species are released alive, except AIS or unidentified species. Captured AIS are destroyed, preserved in 95% ethyl alcohol (EtOH), or frozen for later laboratory analysis. One to three specimens of any unidentified species are retained for identification in the laboratory.

Where to Monitor: Monitoring should be concentrated in deep channels and pools of river estuaries, embayments, canals, sloughs, and river runs within 3 km of a river mouth. Ruffe especially prefer waters that are 3 to 10 m deep, lack vegetation, are cloudy, turbid, or stained with little light penetration. Shallow river runs (<3 m) and channels adjacent to heavy vegetation should also be monitored.

Other habitat should be monitored as follows: Round Goby **-** include shallow flats (<3 m). Threespine Stickleback **-** include light to moderate areas of vegetation in channels and flats, especially areas holding native stickleback (ninespine and brook stickleback). Expect a large amount of vegetation to accumulate just forward of the trawl cod end, as well as within the cod end. This vegetation can trap and hold the target species as well as native species. If disruption to vegetation is a concern, a trawl may not be the best approach.

When to Monitor: Ruffe, White Perch, and Threespine Stickleback **-** monitor in the spring (April-May) just prior to spawning, and in the fall (late September-early October) when the water temperature has cooled to 10-15˚C and the young-of-the-year (YOY) are easily recruited to the gear (YOY are large enough to be trapped and held by the codend netting). Round Goby - begin monitoring when water temperature rises above 5˚C.

Monitoring Target

Ruffe, white perch, round goby, and threespine stickleback with potential to capture rusty crayfish on substrate with rocks, cobble, or large woody debris.

Gear: Use commercially available minnow traps or custom made experimental perch traps called modified Windermere traps (MWT). The minnow traps are available in two lengths but the shorter length with 63 mm entrance holes is recommended. Modified Windermere trap measurements are 0.6 m high by 1.2 m long with netting consisting of a 6.35 mm bar mesh. The diameter of the trap entrance holes measures 5.08 to 6.35 cm. For placement of these traps, see section "Where to Sample".

Procedure: For each sampling location, set traps in groups of three with each trap baited with night crawlers and/or fish spawn. Set traps during daylight hours and lift during daylight hours the following day. Traps can then be moved to another location, or set back if additional effort is desired for a given location. Location coordinates are recorded for each trap. Bottom water temperature is recorded during set and lift. Depth is recorded for each trap within a group, and then averaged to determine the mean depth for that grouping. Recording of catch data is the same as for bottom trawling.

If rock bass or other predators are abundant, they may reduce the effectiveness of minnow traps and modified Windermere traps to attract and catch the target fish. In this circumstance, or to just supplement the minnow and Windermere traps, set commercially available mini-fyke nets along the shoreline in proximity to the traps. The mini fyke nets consist of 0.7 m x 1.0 m rectangular hoops interconnected with 6.35 mm mesh bar and a 15 m lead.

Commercially available gill nets and set lines are also effective in capturing ruffe, white perch, and round goby but due to bycatch mortality it is suggested that these only be used if it is suspected that trawling and/or trapping are not monitoring effectively. Ruffe and white perch can be captured in gill net mesh sizes ranging from 25 to 63 mm stretch mesh, but optimum is 38 mm stretch mesh. Gill net bycatch mortality can be minimized by reducing the net length and width from 30 m x 1.22 m to 15 m x 0.61 m. For set lines, use number 6 or 8 barbless hooks to minimize bycatch mortality along a 15 m line. Gill net and set line bycatch mortality can also be minimized by fishing for a shorter (3-4 hour) period rather than for a typical 12-24 hour period.

Where and When to Monitor: Same as monitoring on mud/sand substrate with minimal obstructions.

Monitoring Target
Rusty Crayfish

Procedure: The Notre Dame University has conducted extensive research on rusty crayfish. Their procedure for capturing rusty crayfish is as follows (J. Murray, personal communication). Modify a commercial minnow trap by adjusting the hole diameters to 38-50 mm and bait it with 120 grams of beef liver (one standard slice in a package holding 4 slices, available at most food markets).

Where to Monitor: Sampling should target areas of rock, cobble, and large woody debris. Set traps in water depths 1.5-2.0 m and a maximum of 26 m apart. If sampling a lake with no rock or woody structure set traps around the lake in water 1.5-2.0 m deep, 50 m to 1 km apart, or a minimum of 20 traps total sampling for the lake. Allow traps to fish over night before lifting.

When to Monitor: The preferred time period to monitor is during early August. Males have just completed their final molt for the season, the water is uniformly warm and they are most active during this time. A second potential sampling period is from late June to early July. This time period is typically an intermolt period. However, the onset of this period may fluctuate due to varying water temperature.

Monitoring Target
Dreissena (zebra and quagga mussels)

The information provided in this section can be found in a sampling protocol developed by Wisconsin Department of Natural Resources and University of Wisconsin Extension (2006) and attached as Appendix 6.

Procedures: If calcium and magnesium concentrations are less than 20 ppm, *Dreissena* are unlikely to develop a shell and survive. If adult *Dreissena* are detected in areas of low conductivity waters they were likely introduced by detaching from a vessel or floating debris. Consider monitoring the water chemistry for calcium and magnesium, targeting waterways with high levels of these chemicals first. Water temperature or more specifically, ice cover, for a significant duration (3-5 months) may reduce the likelihood of establishment as well.

Dreissena can be sampled for the presence of veligers (larvae), post-veligers, or adults. Veliger sampling involves collecting water samples by plankton net, preserving the water samples, and shipping samples to a laboratory for analysis. While veliger sampling is the most likely chance of early detection of dreissenids, the approach is labor intensive, time consuming, and can be expensive. In addition, collection of veligers can be unreliable, and the presence of low numbers of veligers does not in itself constitute establishment of a reproducing population.

We recommend sampling of adults because it is less expensive and simpler to conduct. If adults are detected the gonads can be analyzed in the laboratory to determine maturation and the potential for reproduction. However, maturity and the potential for reproduction does not confirm that offspring will survive as water chemistry, water temperature, or other parameters may not be suitable for completion of the life cycle.

The presence of multiple size classes of adults can be used to infer the presence of multiple year classes and thereby, successful reproduction. Likewise, the presence of adults in different locations within a waterbody can be considered evidence of reproduction and classification as an infestation (Wisconsin Department of Natural Resources and University of Wisconsin Extension 2006). A comprehensive sampling protocol for veligers and adult that can be implemented by volunteers is provided in Appendix 6.

Monitoring Target
Adult/Post-Veliger *Dreissena*

The information provided in this section can be found in a sampling protocol developed by the Wisconsin Department of Natural Resources and University of Wisconsin Extension (2006) and attached as Appendix 6.

Procedures: Substrate monitoring can be conducted with an adult *Dreissena* substrate sampler consisting of four plastic square plates of different sizes assembled in series. It is recommended to suspend two samplers, one on top of the other at mid-depth or 2 m (6 ft), whichever is less. The top sampler is removed, replaced with a clean sampler, and analyzed once per month. The bottom sampler is left for the entire season and removed and analyzed at the end of the season. A cinder block can also be substituted as an adult substrate sampler. No matter what is used as a substrate sampler, measure the total surface area of the sampler, so that the density of detected *Dreissena* can be expressed in terms of number per square meter. Use a chain or steel cable to suspend samplers at the depth prescribed by WIDNR (2 m (6 ft) or mid-depth whichever is less). Try to attach samplers to dock posts, pilings, and other large permanent shoreline structure without the use of buoys (visible markers) as markers may invite vandalism. We also recommend that samplers be inspected at least once per month, in order to facilitate early detection. Construct a map showing placement of samplers in relation to prominent physical features, and record GPS coordinates for each sampler.

Where to Survey:
Shoreline Survey: A shoreline survey can cover a large area of substrate in a short time and eliminates the necessity to tend and maintain sampling equipment. Standard equipment consists of a boat and a pocket magnifying glass to facilitate the identification of juvenile *Dreissena*.

Any hard surface can hold adult and post-veliger *Dreissena*; especially dock pilings, rock, and woody debris. Surveys should target areas of shipping or boating activity, including public boat launches, popular fishing areas, and resort and campground shorelines.

Fall Marina Survey: If a public or private marina is nearby, coordinate with the marina manager in the fall to survey boat hulls shortly after the vessels are removed from the water and placed in dry dock for winter storage. Only one person is required to perform this survey and hundreds of square feet of hard surface can be surveyed in a short time. In most instances, these hard surfaces have been in the water since the water temperature reached 10˚C in the spring. Use a pocket magnifying glass to detect small juvenile *Dreissena*.

Substrate: Samplers should be set in the target locations, and at the time described in "Shoreline Surveys" above.

When to Survey: The WIDNR recommends surveys at 2-week intervals for each water body monitored, starting at ice-out and terminating at ice formation. If funding is limited, we recommend surveys at 1-month intervals, starting when the water temperature climbs to 10˚C in the spring, and terminating when the water temperature declines below 10˚C in the fall. A dock survey conducted in the fall by visual examination of removed dock posts and pilings can be an effective tool for detection of adults.

Documentation: Develop a spreadsheet for recording sampler location, date inspected, number of *Dreissena* detected (if any), and a comments block at the end of each line entry for recording water temperature, chemistry, and other miscellaneous information. In oligotrophic waters such as Lake Superior, it may be helpful to monitor the calcium and magnesium content of the water. On the back of the spreadsheet, include a length/frequency form for recording lengths of any *Dreissena* detected. If adult *Dreissena* are detected initially, preserve in rubbing alcohol, and suggest contacting a biological laboratory to have the gonads of the specimen analyzed for reproduction. Dr. Mary Balsar, Director of Biological Research, University of Wisconsin-Superior, is experienced conducting this procedure.

Monitoring Target
Eurasian Water Milfoil

The Wisconsin Department of Natural Resources (2006) Eurasian water-milfoil protocol (Appendix 7) and GLIFWC Invasive Species Survey Structure (D. Olsen, personal communication) (Appendix 8) were used as references for the following information.

Procedure: Start in shallow water first, where the plants can be observed growing on the bottom. Drag a garden rake suspended by a rope across the substrate to collect observed suspicious plants for identification. After shallow waters have been surveyed where bottom plants can be observed visually, move into deep areas where plants cannot be observed from the surface. Continue towing a garden rake in deep water to cover a few meters of substrate per tow depending on resistance felt, and then pull up and inspect the rake. Do not return any collected plants or plant fragments into the water; dispose properly onshore. If Eurasian water milfoil is detected, try to determine and map the total area of infestation using a global positioning system (GPS).

Where to Monitor: Eurasian water milfoil can grow and survive under a variety of conditions, but grows best in eutrophic waters on silt substrate at a depth range of 2-5 m. However, monitoring should also include mesotrophic and oligotrophic waters, rock and sand substrate, and depths from 0.3 to 6 m. GLIFWC targets areas of greater risk of infestation first (e.g., boat launches), looking at rooted plants and floating plant fragments. Another starting point is the downwind shoreline area of the prevailing wind. From this point, expand the survey to cover the entire potential Eurasian water milfoil habitat within the water body.

When to Monitor: Monitor from late spring to mid-summer when plant growth is greatest.

Documentation: Develop a spreadsheet for each water body monitored. Include a line entry for each rake tow and whether the substrate is visible or not. Record GPS coordinates for the beginning and end of each tow, the survey date, water temperature, and substrate type. Retain unidentified plants for later identification, referencing the date and location collected. Mark tow locations on a map in order to facilitate total water body coverage over the course of the growing season. If Eurasian water milfoil is detected, try to determine and map the outer boundary of the plant bed, or the center of the plant bed.

Detecting Status and Trends

How will change be statistically detected over time?

The following guidance was paraphrased from Droege and Moore (2004). It is basic, but applicable in approach if interested in Percent of Area Occupied (PAO).

Rather than monitoring the number of individuals of a target species (abundance) in an area, it may be advantageous to monitor what fraction of the area is occupied by the species, i.e., the distribution of the species across a landscape. Sometimes this is referred to as the frequency of occurrence, although Droege and Moore (2004) use the phrase "proportion of area occupied" (PAO). The logic being that estimating abundance can be expensive and difficult in some situations (individuals need to be identifiable and counted accurately), but that changes in abundance will likely correspond in a change in the PAO by the species (which may be more easily measured with presence/absence surveys), hence it may be suitable as a coarse surrogate.

Droege and Moore (2004) suggest PAO may be most appropriate in mid-level monitoring programs where a lower resolution measure could provide adequate information about changes *in species distributions without consuming resources that might be better used in more intensive monitoring (i.e., abundance estimation) of more "valuable" species.*

The term "area" refers to the region or collection of units that are of primary interest to the monitoring program. This could be a Park unit or a collection of discrete habitat units such as an inland lake, stream or estuary. A number of monitoring sites are selected from within the area of interest (using a rigorous study design), and the presence or absence of the species at the sites determined through any appropriate field techniques.

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Appendices

Appendix 1. Description and characteristics of Great Lakes Inventory and Monitoring Network (GLKN) park units, from Lafrancois and Glase (2005).

Apostle Islands National Lakeshore

Apostle Islands National Lakeshore (APIS), established in 1970, is an island archipelago consisting of 21 islands located off northern Wisconsin's Bayfield Peninsula in Lake Superior. The Lakeshore also features a 19 km (12 mile) mainland unit along Lake Superior. Together, the islands and mainland unit protect 257 km (160 miles) of Lake Superior shoreline. Apostle Islands National Lakeshore jurisdiction in Lake Superior waters near the islands is limited to the meniscus; that is, jurisdiction does not extend below the surface through the water column, but the surface area under Park jurisdiction covers nearly 11,000 ha (27,000 acres). The mainland unit and several islands contain notable inland water resources that have received some attention over the past several decades. The Sand River runs through the mainland unit, and small perennial and intermittent streams are found on several of the islands. Unlike other parks, APIS features very few named streams and lakes. However, it features more kilometers of intermittent streams than any other Great Lakes area park. Unique lagoon ecosystems are found on the mainland unit as well as Stockton, Outer, and Michigan Islands. Bogs, beaver ponds, and wetlands occur on many of the islands.

Grand Portage National Monument

Grand Portage National Monument (GRPO) was designated in 1951 as Grand Portage National Historic Site and re-designated a national monument in 1958. Situated just south of the Canadian border along Grand Portage Bay of Lake Superior, GRPO lies entirely within the Grand Portage Band of Minnesota Chippewa Reservation. It remains chiefly a historical monument, preserving a vital center of centuries-old fur trade activity and Anishinaabeg (Ojibwe) heritage. Unlike other Great Lakes area parks, GRPO contains very few water resources. Lake Superior and the Pigeon River are the most prominent water resources in the area, but these are beyond GRPO jurisdiction. Local parts of Lake Superior are managed jointly by the Grand Portage Band of Minnesota Chippewa Reservation and the Minnesota Pollution Control Agency, in keeping with a novel cooperative agreement approved by the Environmental Protection Agency in 1996. Grand Portage National Monument's 14 km (8.5 mile) portage corridor between Lake Superior and Fort Charlotte intersects the watersheds of three streams, namely Grand Portage Creek, Poplar Creek, and Snow Creek. All three drain portions of the rugged Grand Portage Highlands.

Isle Royale National Park

Isle Royale National Park (ISRO) is a remote island archipelago situated in the northwestern portion of Lake Superior. Isle Royale National Park was designated in 1931 and preserves a total land area of over 220,000 ha (965 square miles), including submerged land, which extends four and a half miles out into Lake Superior. The Park consists of one large island surrounded by about 400 smaller barrier islands, which together protect 543 km (338 miles) of coastline, more than any other Great Lakes park. Much of the Park was designated as Wilderness in 1976, and its relatively pristine condition has made it an ideal natural laboratory and a United Nations Biosphere Reserve. Prominent water resources include Lake Superior, including an expansive 165,182 ha (408,173 acres) of bays, nearshore waters, and offshore waters. Additionally, ISRO features more named inland lakes than any other Great Lakes area park, several perennial streams (e.g., Washington, Grace, and Tobin Creeks, and Big Siskiwit, Little Siskiwit, and

Siskiwit Rivers), many kilometers of un-named perennial and intermittent streams, and many inland wetlands associated with lake littoral zones, beaver activity, and the pronounced ridge-valley topography.

Pictured Rocks National Lakeshore

Pictured Rocks National Lakeshore (PIRO) preserves 62 km (39 miles) of Lake Superior shoreline in Michigan's Upper Peninsula. The Lakeshore's namesakes are the colorful sandstone cliffs extending along the shoreline, but the Lakeshore also protects a variety of aquatic resources. Lentic water resources include extensive coastal habitats, over 2,400 ha (6,000 acres) of Lake Superior surface waters, 14 named inland lakes, and several beaver ponds and wetlands. Prominent inland lakes include Grand Sable, Beaver, Little Beaver, Chapel, Little Chapel, Miners, Trappers, Legion, Kingston, and the Shoe Lakes. Lotic water resources at PIRO are unique and more plentiful than at many Great Lakes area parks (excluding the river parks). Pictured Rocks National Lakeshore features 19 named streams, of which Miner's River is the longest and largest. In general, PIRO streams are short and drain directly to Lake Superior. Two PIRO streams (Beaver and Grand Sable Creeks) originate in lakes, although the lakes themselves have tributary streams. In addition to the federally-owned shoreline zone, PIRO also includes a second layer of protection via the non-federally-owned Inland Buffer Zone (IBZ), consisting of state forest land, private commercial forests, and small private holdings. At one time, the IBZ also contained national forest land but this land was turned over to Lakeshore ownership several years ago. The designation of the IBZ acknowledged the importance of watershed processes to the protection of PIRO's inland waters.

Sleeping Bear Dunes National Lakeshore

Sleeping Bear Dunes National Lakeshore (SLBE) was established in 1970 and preserves over 105 km (65 miles) of Lake Michigan shoreline, including the 120 m (400 feet) tall Sleeping Bear Dunes and the Manitou Islands. In addition to these unique scenic qualities, SLBE also protects a variety of water resources, all of which have been designated outstanding state resource waters (OSRW) by the State of Michigan. Sleeping Bear Dunes National Lakeshore waters include 18 named inland lakes of varying size and character, four sizable streams (all of Otter Creek and parts of the Platte River, Crystal River, and Shalda Creek), and many bogs, springs, and interdunal wetlands. In general, SLBE surface waters are characterized by significant groundwater contributions and feature relatively stable hydrographs.

Indiana Dunes National Lakeshore

Indiana Dunes National Lakeshore (INDU) was designated in 1966 and protects 19 km (12 miles) of Lake Michigan shoreline between Gary, Indiana and Michigan City, Indiana. Prominent aquatic resources include Lake Michigan, 241 ha (596 acres) of which are under INDU jurisdiction, extensive emergent wetlands, forested wetlands, and bogs, the Grand Calumet Lagoons, and streams. In addition to natural streams, INDU also features 19 km (12 miles) of ditched streams, some of which have been named and studied intensively. Lentic water resources are not strongly represented at INDU, with only two named lakes (Lake George and Long Lake) and one named bog (Pinhook Bog) noted in our analysis. Indiana Dunes National Lakeshore water resources are exposed to a complex mixture of adjacent land uses, ranging from heavily industrial to residential and agricultural.

St. Croix National Scenic Riverway

One of eight rivers granted protection under the original Wild and Scenic Rivers Act, St. Croix National Scenic Riverway (SACN) was designated in 1968, with the Lower Riverway designated in 1972. The Riverway stretches 420 km (261 miles) from the forested headwaters of the St. Croix and Namekagon Rivers in northern Wisconsin to the confluence of the St. Croix with the Mississippi River near the Twin Cities Metropolitan Area. The Riverway is recognized for its outstanding scenic qualities and its diverse and rare biological resources. It is home to over 100 species of fish and 40 species of unionid mussels, two of which are federally endangered. Upper reaches of the Riverway flow through forests, marshes, and peatlands. At Taylors Falls, Minnesota, the river passes through a hydroelectric impoundment and a deep gorge. The river below the dam is characterized by islands, sandbars, and sloughs, and becomes lake-like for its final 41 km (26 miles), due to its impoundment by a 9,500 year-old delta of the Mississippi River. Various aquatic habitats can be found within and along the Riverway, including tributary streams, wetlands, floodplain forests, backwaters, ponds, and spring and cliff seeps. Due to the nature of the Park, more kilometers of perennial and intermittent streams are found at SACN than at any other Great Lakes area park. Since the federal designation affords only a thin corridor of protection to the St. Croix River, SACN resource managers coordinate their activities with those of other resource management agencies throughout the watershed. This cooperation is facilitated by the St. Croix Basin Water Resources Planning Team (hereafter referred to as the Basin Team), created in 1994 via an interagency memorandum of understanding.

Mississippi National River and Recreation Area

Mississippi National River and Recreation Area (MISS) comprises a 123 km (77 mile) stretch of the Mississippi River that passes through the Minneapolis - St. Paul Metropolitan area. Lands held by the National Park Service are limited to a handful of floodplain islands in the Mississippi River, but MISS boundaries encompass a complex mix of privately owned lands and public lands administered by local governments, organizations, and state and federal agencies. The primary role of MISS is to support and coordinate activities that protect natural and cultural resources, provide diverse recreational opportunities, and contribute to the economy. Primary water resources include parts of the Mississippi, Minnesota, and Vermillion Rivers, a number of perennial and intermittent tributary streams, and significant floodplain wetlands and standing backwaters. Water resource information pertinent to MISS is available from a variety of sources. The Metropolitan Council and U.S. Geological Survey represent two primary sources for information cited in this document. Information collected by the U.S. Army Corps of Engineers, the Minnesota Department of Natural Resources, and local parks may be available at the respective agency offices, but is not presented here.

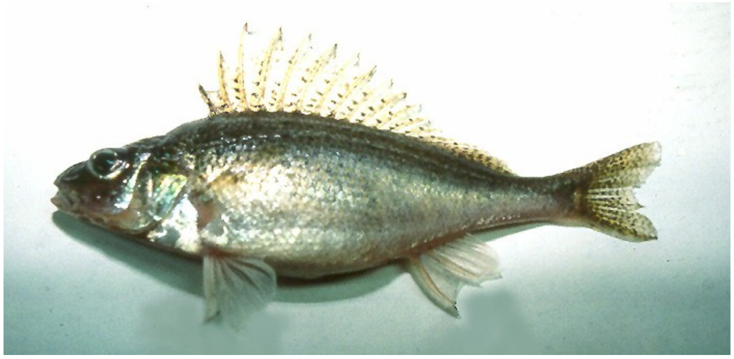
Voyageurs National Park

Voyageurs National Park (VOYA) protects over 88,000 ha (217,000 acres) of the watery U.S.-Canada borderlands in northern Minnesota. The Park is situated along the southern reaches of the Canadian Shield and is characterized by Precambrian granite bedrock geology and expansive boreal forests. Voyageurs National Park is the only Great Lakes Network park within the Hudson Bay drainage basin. Water is central to the Park's history and its present day ecology, and aquatic habitats cover nearly half of the Park's area. These aquatic habitats include four large lakes (Rainy, Namakan, Kabetogama, and Sand Point), 29 named interior lakes, and countless wetlands and beaver ponds. Relative to its lentic water resources, VOYA features relatively few kilometers of perennial and intermittent streams. It does, however, contain a greater area of wetlands and inland lakes than any other Great Lakes Network park, including some 275 un-named lakes. Lake levels in the Park's large lakes have been regulated by a hydroelectric dam on Rainy Lake and regulatory dams on Namakan Lake since the early 1900s. Because these waters are shared by the United States and Canada, the International Joint Commission oversees water level management in the area. Reservoir operations were modified in 2000 to more closely approximate natural water level fluctuations in the large lakes at VOYA.

Appendix 2. Profiles for 12 target aquatic invasive species.

This section provides descriptive information on the twelve aquatic invasive species that are the focus of this report. For each species addressed we provide a description of the organism, life history characteristics, status and distribution, invasive impacts, and control. In an attempt to make each species account easier to read and comprehend, the references used to develop these species profiles are provided at the end of species account rather than cited within the text. We encourage readers to familiarize themselves with the references at the end of the section if a more thorough understanding of a particular species is desired.

Life History and Invasion Impacts of the Ruffe



Kingdom	Animalia -- Animal, animals, animaux
Phylum	Chordata -- chordates, cordado, cordés
Subphylum	Vertebrata -- vertebrado, vertebrates, vertébrés
Superclass	Osteichthyes -- bony fishes, osteíceto, peixe ósseo, poissons osseux
Class	Actinopterygii -- poisson épineux, poissons à nageoires rayonnées, ray-finned fishes, spiny rayed fishes
Subclass	Neopterygii -- neopterygians
Infraclass	Teleostei
Superorder	Acanthopterygii
Order	Perciformes -- perch-like fishes
Suborder	Percoidei
Family	Percidae -- perches, perches, true perches
Genus	Gymnocephalus Bloch, 1793 -- European ruffes
Species	Gymnocephalus cernuus (Linnaeus, 1758) -- blacktail, pope, redfin darter, ruffe

Description

The Ruffe (pronounced ruff) (*Gymnocephalus cernuus*), is a small, slimy, spiny, bottom dwelling fish. Adults can exceed 250 mm (10 in.) in length, but in Lake Superior rarely exceed 200 mm (8 in.). The most common observed length of adult ruffe in Lake Superior is 125-150 mm (5-6 in.). Like yellow perch (*Perca flavescens*) and walleye (*Stizostedion vitreum*), ruffe are classified as spiny-rayed fish. The two dorsal fins are large and joined together; the anterior dorsal has 11-16 hard spines usually with rows of dark spots between the spines; the posterior dorsal is soft rayed. The pelvic fins have one anterior hard spine that is one-half the length of the posterior soft rays, and the anal fin has two anterior spines equal in length to the posterior soft rays. The gill flap (preoperculum), which is anterior to the gill opening, also has a few small spines (also referred to as "strongly serrated") around its posterior margin. The dorsal area of the ruffe's upper body is usually mottled with dark spots. Ruffe eyes are large and glassy similar to a walleye, and their caudal fin is forked. Young-of-the-year ruffe can easily be confused with trout-perch (*Percopsis omiscomaycus*), but are distinguished from trout-perch by the absence of an adipose fin (small fleshy fin separated from and posterior to the dorsal fin). Trout-perch have an adipose fin which is visible even on the smallest specimens.

Distribution and Status
The ruffe is native to Europe and Asia, and was first introduced into the St. Louis River Estuary (SLRE) (Duluth/Superior harbor), Minnesota/Wisconsin during the mid 1980s. The vector of introduction was probably the discharge of ballast water from an ocean-going ship. Ruffe migrated rapidly eastward along the south shore of Lake Superior reaching the Ontonagon River estuary, Michigan by 1994, 276 km (173 mi.) east of the Duluth/Superior harbor. As of 2004, ruffe occur in Lake Huron near Alpena, Michigan; northern Green Bay, Lake Michigan; and as far east as Thunder Bay Harbour, Ontario on the north shore of Lake Superior, and Marquette Harbor, Michigan on the south shore of Lake Superior. Ruffe have not been found in the Lower Great Lakes or in any inland lake or stream. Established ruffe densities in small tributary estuaries close to their origin (St. Louis River estuary) average 200/ha (81/acre) in trawls . In the large estuary of the St. Louis River (SLRE), ruffe densities peaked at 2,000/ha (810/acre) in trawls during the mid 1990s, but have since averaged 1,000/ha (405/acre). On the periphery of their range, ruffe are uncommon to common, except in the Kaministiquia River estuary, Ontario, where they are abundant.

Life History
Ruffe can live in a broad range of ecological and environmental conditions including fresh or brackish water, temperatures ranging from 0-30˚C, and oligotrophy to eutrophy. Ruffe prefer areas of turbid, slow-moving water with little light penetration, over soft substrate devoid of vegetation. These conditions often occur in river estuaries, embayments, canals, and shipping ports.

Ruffe may mature in one year, but females do not produce viable eggs until age 2 or 3. Spawning can occur from mid-April to July at a water temperature between 6˚C and 18˚C. In Lake Superior, spawning usually occurs from late April thru June (peaking during May) at a water temperature of 12-14˚C. Fecundity depends on the size of the female, and ranges from 7,000 to 80,000 eggs. Eggs are laid in batches on bare substrate and available submerged items, and are not guarded. Ruffe are benthic feeders; their diet consists of microcrustaceans, primarily Cladocera, during their first two months of life, and then transitions to macroinvertebrates, primarily Chironomids (midgefly larvae). Ruffe possess a highly developed cephalic lateral-line system, which allows them to detect the vibrations of their prey in turbid water. This attribute together with their spiny rays also helps them to avoid being preyed upon.

Invasion Impacts
Ruffe are usually very invasive (become abundant where introduced and spread rapidly). Due to potential competition for food and space, ruffe pose a threat to native fish populations. Experimental research conducted by the University of Minnesota-Duluth revealed that ruffe consumes a significant amount of benthic macroinvertebrate energy. Research also demonstrated significant declines in the growth of yellow perch, while in the presence of lesser densities of ruffe, as well as in the presence of equal or greater densities of ruffe. However, a statistical analysis conducted by the U.S. Geological Survey showed no significant relationship between an increasing ruffe population and declining native fish populations in the SLRE. In three Wisconsin tributaries just east of the SLRE, 1995-2002 trawl data suggests that yellow perch

abundance declines in years that ruffe abundance increases. Ruffe also prey on fish eggs, and have been implicated in decline of whitefish.

Nonindigenous parasite species were introduced into North America along with their host ruffe. These parasites are specific to Eurasian percids, but may pose a health threat to North American percids including yellow perch, walleye, and sauger (*Stizostedion canadense*).

Ruffe are also bait stealers, degrading the quality of sport fishing, and ruffe decrease commercial fishing efficiency by clogging nets as a nuisance bycatch. Their spiny rays make their removal from nets time consuming and injurious to commercial fishers.

Control

As a result of increasing abundance and expansion outside the SLRE and reports of potential impacts on native fish populations, the Aquatic Nuisance Species Task Force declared the ruffe to be a "nuisance species" in the spring of 1992. By authority of the Nonindigenous Aquatic Nuisance Prevention and Control Act of 1990, this designation authorized the formation of a control committee charged with the responsibility of designing and implementing a control plan. The *Ruffe Control Program* was drafted in 1995 with a revision in 1996 after ruffe were discovered in Lake Huron in 1995. The goal of the *Ruffe Control Program* is "to prevent or delay the spread of ruffe in the Great Lakes and inland waters." Of the eight objectives designed into the program to achieve this goal, surveillance, ballast water management, and education have shown to be the most effective. Surveillance facilitates early detection and response to implement more stringent regulations such as curtailment of commercial bait collection and ballast water exchange. Ballast water management refers to the voluntary ballast water exchange guidelines in the Great Lakes initiated by the Lake Carriers Association. Ships taking on ballast in ruffe infested harbors exchange that ballast over deep water beyond 5 nautical miles from shore. Education creates public awareness, which decreases the probability of accidental human assisted expansion, and increased incidental monitoring.

Formal surveillance efforts began in 1992 to detect pioneering populations of ruffe in the Upper Great Lakes. These efforts were initiated by the U.S. Fish and Wildlife Service-Ashland Fishery Resources Office and the Lake Superior Management Unit of the Ontario Ministry of Natural Resources.

Due to the spiny rays of ruffe, predators were slow to utilize ruffe as prey. Walleye, in particular, took several years before their predation on ruffe was observed. Predator enhancement initiated in the SLRE during the early 1990s by the Minnesota and Wisconsin Departments of Natural Resources failed to control that exploding ruffe population. Only when the availability of soft rayed forage fish declined in the SLRE, did predators begin to feed on ruffe. Presently, most predators, including salmonids, feed on ruffe. Due to the reproductive ability of ruffe, it is unlikely that biological control could be effective. In fact, predation of ruffe may have offsetting consequences. As a defense mechanism, ruffe flare their spiny fins and gill covers when threatened. An angler reported catching a brown trout with a ruffe spine protruding out from the stomach area, demonstrating that predation of ruffe may be occasionally injurious and fatal to predators.

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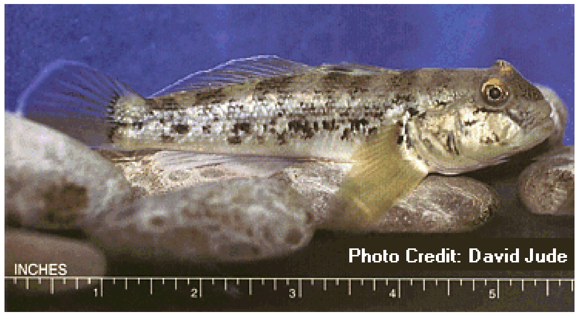
Life History and Invasion Impacts of the Round Goby



Kingdom	Animalia, -- Animal, animals, animaux
Phylum	Chordata -- chordates, cordado, cordés
Subphylum	Vertebrata -- vertebrado, vertebrates, vertébrés
Superclass	Osteichthyes -- bony fishes, osteíceto, peixe ósseo, poissons osseux
Class	Actinopterygii -- poisson épineux, poissons à nageoires rayonnées, ray-finned fishes, spiny rayed fishes
Subclass	Neopterygii -- neopterygians
Infraclass	Teleostei
Superorder	Acanthopterygii
Order	Perciformes -- perch-like fishes
Suborder	Gobioidei -- gobies, gobies
Family	Gobiidae -- gobies, gobies, true gobies
Genus	Neogobius Iljin, 1927 -- round gobies
Species	Neogobius melanostomus (Pallas, 1814) -- round goby

Description

The round goby is a small, bottom dwelling, fish that is morphologically similar to the native mottled sculpin (*Cottus bairdii*). The goby can grow larger than a sculpin, up to 300 mm (12 inches), but most Great Lakes gobies are less than 375 mm (7 inches) in length. Goby coloration is also similar to a mottled sculpin, mottled grey and brown in color, except that a spawning male goby is usually black. Goby eyes protrude like a frog's, near the top of their large heads, the remainder of their body being narrow and soft with fine scales. Like the mottled sculpin, goby fins are soft-rayed, but the goby pelvic fins are fused together to form a suction cup called a suctorial disk. The goby uses the suctorial disk to adhere to hard substrates in fast moving water. The pelvic fins on all sculpins are separated and distinctive. The goby's anterior dorsal fin also has a black spot near the base of the fin; this spot is absent on sculpin dorsal fins. The suctorial disk and the black spot at the base of the anterior dorsal fin are the two most distinguishing features separating the round goby from the mottled sculpin.

Distribution and Status

The round goby is native to and abundant in the Black and Caspian seas, where it is commercially fished. Discovered in Lake St. Claire in 1990, round goby were soon found in all the Great Lakes including their tributaries and feeder streams. However, in Lake Superior the goby is only found in the harbors of Duluth-Superior and Thunder Bay, Ontario. The goby is believed to have arrived in the ballast water from a trans-oceanic ship, and its rapid expansion (5 years) throughout the Great Lakes was assisted by the ballast water discharge of intra-lake shipping. Gobies are most abundant in the nearshore waters and tributary estuaries of the Great Lakes during the warm season, and migrate into deeper waters during the cold season. As abundance increases, gobies migrate upstream from tributary estuaries. There is evidence to suggest that goby distribution and rate of expansion may be related to one of its preferred food items, the zebra mussel (*Dreissena polymorpha*). Since their discovery in the Duluth-Superior harbor in 1995, the goby has not expanded outside the harbor, except for the Amnicon River, 10 miles to the east. The only known reproducing population of zebra mussels in Lake Superior also remains confined to the Duluth-Superior harbor. The round goby is very invasive, increasing rapidly in abundance where introduced due to its aggressive feeding and defensive behavior, ability to survive in degraded water, and its prolific reproductive ability (multiple spawnings in one season). Goby densities vary depending on site with more gobies occurring on rocky sites than sandy sites. The largest documented density appears to be 90 per square meter (8.4 per square foot), which occurred in the St. Clair River near Sarnia, Ontario, Canada. Goby density in the Duluth-Superior harbor has increased to 0.008 per square meter (0.0007 per square foot) which includes a fourfold increase from 2003 to 2004. Initially, gobies only occupied the sand flats in the harbor. In 2004, gobies were found in all three harbor habitats including dredged and undredged channels. In Grant Calumet Harbor, Lake Michigan, goby density has been reported up to 40 per square meter (3.7 per square foot). The U.S. Fish and Wildlife Service (USFWS) conducts annual ruffe surveillance at 7 harbor locations in Lake Erie. From 2000 to 2004, round goby was either the most abundant or second most abundant species collected in trawls with relative abundance ranging from 10% to 54% of the total catches. The USFWS also conducts similar ruffe surveillance activity in 16 established locations (mostly river estuaries) in Lake Huron and the St. Marys River. During the same period, 2000-2004, round goby was either the most abundant or second most abundant species collected in trawls with relative abundance ranging from 17% to 83% of the total catch.

Life History

The Gobiidae are well-adapted and tolerate diverse conditions, including both fresh and salt water habitats. They can occur in both rivers and lakes, and prefer coarse gravel substrate, but they can also be found over sand and clay. Preferred depth varies by water body. They are found down to 20 m (6.1 feet) in the Black Sea and down to 70 m (21.3 feet) in the Caspian Sea. In the Great Lakes, gobies occupy the nearshore at depths less than 15 m (4.6 feet) during spring and summer, but move offshore into deeper water during winter. Spring inshore movement is triggered at a water temperature of 5-8˚C.

Gobies can spawn up to 6 times from April to September. Females mature at age one, and lay from 300 to 5000 eggs under and around rocks and logs. Males guard the nests, supplying oxygen to the eggs by fanning their tales. Males also turn black during spawning, and reportedly die after spawning. Although evidence is not well documented, authoritative observations confirm that male death post spawning does occur in the Great Lakes. However, some males have been aged at 5-6 years old, so male death post spawning may not always be the case. Round goby feed on amphipods, chironomids, polychaetes, small fish and fish eggs, but gobies larger than 100 mm (4 inches) feed predominantly on zebra mussels. One goby can consume as many as 80 zebra mussels per day. They have a sensory system which allows them to detect water movement and prey in complete darkness, and they tend to aggregate where abundant. Maximum length is reported to be 215-250 mm (8.6-10.0 inches), but a 275 mm (11.0 inches) goby has been captured in the Great Lakes.

Invasion Impacts

The round goby's aggressive behavior, larger size, and egg feeding appetite allow it to out-compete mottled sculpin (*Cottus bairdi*) and logperch (*Percina caprodes*) for spawning habitat. Significant declines in these two species and near extirpation have already been observed where they coexist with gobies. The implications of possible extirpation of these two forage species means a reduction in diversity of species and available prey for predators. Food chain bioaccumulation of PCBs occurs in the round goby through consumption of zebra mussels. Round goby are preyed upon by many sport fish. As the PCBs pass up the food chain, the chance of human exposure to PCB's increases. Large goby densities in Green Bay, Lake Michigan have reduced abundance of macroinvertebrates there. The round goby also preys upon the eggs of lake sturgeon, smallmouth bass, and other centrarchids, decreasing recruitment in these species. Implications include less availability for sport and commercial fishermen. Round goby also decrease the quality of angling through bait stealing.

Control

Round goby can only be controlled by preventing their further spread into inland lakes and streams. Due to their small size, soft finrays and bodies, and availability, gobies are attractive prey for all predators, but biological control is not feasible due to the goby's prolific reproduction. However, the goby's preference for zebra mussels may potentially limit their distribution. Round goby was discovered in the Duluth/Superior harbor in 1995, where zebra mussels are abundant. No reproducing population of zebra mussels has been detected in Lake Superior outside of the Duluth/Superior harbor, and the round goby has not migrated outside the harbor, since it was introduced there in 1995. The LaCrosse Fish Health Center of the USFWS evaluated several toxicants on round goby, but found none that were specific to goby. However, since the goby is a bottom dweller, time-release toxicants that toxify only the bottom strata may be useful in certain situations. The maximum swim speed of the round goby has been tested to be 75 cm/s. Where water velocities exceed 75 cm/s, the likelihood of a successful goby introduction would be remote.

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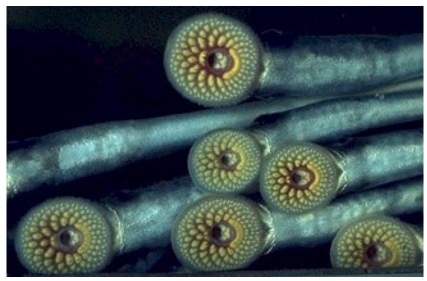
Life History and Invasion Impacts of the Sea Lamprey



Kingdom	Animalia -- Animal, animals, animaux
Phylum	Chordata -- chordates, cordado, cordés
Subphylum	Vertebrata -- vertebrado, vertebrates, vertébrés
Superclass	Agnatha -- agnato, cyclostomes, jawless fishes, peixe
Class	Cephalaspidomorphi -- lampreys
Order	Petromyzontiformes
Family	Petromyzontidae -- lampreys
Subfamily	Petromyzontinae
Genus	Petromyzon Linnaeus, 1758
Species	Petromyzon marinus Linnaeus, 1758 -- lamproie marine, sea lamprey

Description

The sea lamprey is an eel-like fish, which lacks several characteristics usually associated with fish including bones, jaws, scales, paired fins, a lateral line, and a swim bladder. The lamprey's body is supported by cartilage instead of bones. Its mouth lacks jaws, and contains sharp conical teeth with a rasping tongue. The sea lamprey usually grows to 300-500 mm (12-20 inches) in length, but it has been observed up to 900 mm (36 inches) in length. Sea lamprey eyes are small as is the tail fin, and there are two dorsal fins; the anterior is smaller than the posterior; and both dorsals are separated and set back beyond the midpoint of the body. The sea lamprey has 7 gill openings arranged in a lateral row on each side of the body, just posterior to the eyes. Dorsally, sea lamprey coloration is blue-brown to blue-grey with black patches, and becoming silver-white ventrally. Some yellow tones also develop during spawning.

Distribution and Status

The sea lamprey is native to the Atlantic Ocean. On the North American side of the Atlantic, the sea lamprey occurs from southwest Greenland to northern Florida. On the European side of the Atlantic, it ranges from Norway to the Mediterranean and Adriatic Seas, including the North and

Baltic Seas to Finland. The sea lamprey also occurs in Lake Champlain, and the inland lakes of northern and western New York State, It is considered nonindigenous in all of the Great Lakes except Lake Ontario and the St. Lawrence River which have always been open to the Atlantic. However, its origin in Lake Ontario is controversial, and some biologists consider it to be non-native there as well. Introduction from Lake Ontario into Lake Erie and the Upper Great Lakes occurred during the opening of the Welland Canal. In most areas of the Great Lakes, sea lamprey abundance is currently 10% of their peak abundance observed during 1961, due to the application of two lampricides called TFM (chemically identified as 3-trifluoromethyl-4-nitrophenal) and Bayer 73 (chemically identified as 2',5 dichloro-4'-nitrosalicylanilde).

Life History

In its native range the sea lamprey is anadromous (migrates from the sea to spawn in fresh water). In the Great Lakes, sea lampreys have adapted to complete their entire life cycle in fresh water. Parasitic phase sea lampreys spend 12-20 months in a lake or marine environment maturing and preying on fish. A sea lamprey feeds on fish body fluids and blood. This is accomplished by attaching its suctorial mouth to a fish, and rasping through the scale and muscle with its teeth. A sea lamprey will remain attached and feed until it is satisfied or the fish dies. Upon reaching sexual maturity, an adult sea lamprey migrates from the lake into a stream, and travels upstream until it finds suitable spawning habitat, a riffle consisting of gravel or small rocks roughly the size of a golf ball or baseball. Here it excavates a redd, a depression that serves as a nest, and mating occurs over the redd. One female can lay from 60,000-230,000 eggs, and both adults die after spawning. When the eggs hatch, the emerging larvae (ammocoetes) burrow into the sediment where they filter feed on debris and algae for 3-6 years. Between 3 and 6 years, the larvae metamorphose (transform) into the parasitic phase developing eyes and teeth and the ability to swim and parasitize fish. Metamorphosis generally occurs when sea lampreys are about 125-150 mm (5-6 inches) in length. After metamorphosing, the sea lamprey now known as transformers, migrate downstream to the lake or marine environment where they become parasitic and feed and mature, and the life cycle repeats. The total life cycle of a sea lamprey takes 5-8 years to complete.

Invasion Impacts

The most significant impact of sea lamprey in the Great Lakes is the destruction of the large native predator fish populations which comprise the top levels of the food web in the Great Lakes. The repercussions from this destruction range from population explosions of lower trophic levels to the economic losses sustained by a $4 billion/ year commercial and recreational fishing industry. During its lifetime, one sea lamprey may kill up to 40 pounds of fish. In the Great Lakes, sea lamprey have been observed to prey on salmonids (trout and salmon), coregonids (whitefish), yellow perch (*Perca flavescens*), walleye (*Stizostedian vitreum*), northern pike (*Esox lucius*), muskellunge (*Esox masquinongy*), lake sturgeon (*Acipenser fulvescens*), and bass (*Micropterus sp*.). However, sea lampreys have inflicted their greatest impact with their predation on lake trout. Prior to the introduction of sea lamprey, the annual commercial and sport harvest of lake trout (*Salvelinus namaycush*) in Lakes Superior and Huron was 15 million pounds. At the peak of sea lamprey abundance (early 1960s) this annual harvest had declined to 300,000 pounds. As lake trout declined and became less available, sea lamprey increased predation on whitefish causing significant declines in those populations. Significant declines in lake trout led to the overabundance and subsequent die-off of the non-native alewife (*Alosa* *pseudoharengus*). Thousands of dead alewife washed up on Great Lakes shorelines making beaches unhealthy and decreasing recreational use. Adding to the decline of the recreational fish industry are the lamprey scars left on the large surviving fish which are sought by sport anglers for trophies as well as food. These large fish become undesirable for mounting due to the presence of the lamprey scars.

Pacific salmon, coho (*Oncorhynchus kisutch*) and Chinook (*Oncorhynchus tshawytscha*) were introduced to control increasing populations of alewife and provide a recreational fishery to replace the loss of lake trout. These salmon were chosen for introduction because they consume a large quantity of prey and grow much faster than lake trout. Pacific salmon grow to full size and mature at 3-4 years of age, while lake trout mature at 7-12 years and may life up to 30 years.

Control

The control of sea lamprey in the Great Lakes is the most successful account of aquatic invasive species control in North America. Since the implementation of the use of lampricides TFM and Bayer 73, the Lake Superior lake trout population is now reproducing at a sustained level; supplemental stocking of lake trout is no longer required in most areas of Lake Superior; and lake whitefish abundance is at an all-time high.

However, success of sea lamprey control cannot be attributed to the application of lampricides alone. Due to rising lampricide costs, concerns of lamprey developing immunity to the lampricides, and social acceptance of lampricides, an integrated approach to sea lamprey control has been adopted. The integrated control approach involved the use of lamprey barriers (dams, velocity and electric barriers), mechanical traps, and the stocking of sterile male sea lampreys. Sterile males compete with fertile males for females. When a female is stimulated by a sterile male to lay her eggs, the eggs come in contact with unviable sperm and never fertilize. In addition, research is currently being conducted on the use of pheromones (smell) attractants.

Research has discovered that adult sea lamprey return to streams based in part on the presence of chemicals produced by larval sea lamprey in the stream and that sex pheromones can be used to attract sea lampreys during spawning. If researchers can identify these smells or pheromones chemically, they might be synthesized and used to attract spawning phase sea lamprey into traps or to habitat unsuitable for spawning or larval development. Commercial harvest was also considered as a control tool, since lamprey is considered a delicacy in Portugal. However, this idea was suspended over concern for the amount of mercury contained in Great Lakes sea lamprey. Further research into alternative forms of sea lamprey control is ongoing.

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Life History and Invasion Impacts of the Threespine Stickleback

Kingdom	Animalia -- Animal, animals, animaux
Phylum	Chordata -- chordates, cordado, cordés
Subphylum	Vertebrata -- vertebrado, vertebrates, vertébrés
Superclass	Osteichthyes -- bony fishes, osteíceto, peixe ósseo, poissons osseux
Class	Actinopterygii -- poisson épineux, poissons à nageoires rayonnées, ray-finned fishes, spiny rayed fishes
Superorder	Ostariophysi
Order	Gasterosteiformes
Suborder	Gasterosteoidei
Family	Gasterosteidae -- sticklebacks
Genus	Gasterosteus Linnaeus, 1758 -- valid
Species	Gasterosteus aculeatus Linnaeus, 1758 -- valid

Description

The threespine stickleback is a small forage fish, consisting of several varieties or sub-species which occur throughout the world in freshwater, brackish water, and marine water. Morphologically, the threespine stickleback is separated into two forms, freshwater and anadromous. The distinguishing morphology between the two forms requires the use of a microscope, and will not be described here. The freshwater forms may grow to a maximum total length of 80 mm (3.2 inches), and the anadromous forms may grow up to 110 mm (4.4 inches) in total length. The freshwater forms can have three different colorations depending on location and environmental conditions. A typical coloration observed in Lake Superior is olive green on the dorsal area transitioning to silver or bronze ventrally. The other color variations of the freshwater forms include an overall bronze with grey mottling, a bronze/green, or a brassy color. The anadromous forms can also have three colorations including silver/green, blue/black, or all black. In both forms, freshwater and anadromous, the breast of the male may be red or orange during breeding. When distinguishing threespine from native sticklebacks, rely on the number of dorsal spines of which there are three to four anterior to the dorsal fin. Sometimes, only two dorsal spines may be visible to the naked eye. These are much more rigid, sharper, and stout than the 5 dorsal spines of the native brook stickleback, or the 9 dorsal spines of the native ninespine stickleback. Each pelvic fin of the threespine stickleback consists of one spine, and these are connected to a strong shell-like pelvic girdle. The anal fin also contains one spine, and together with the dorsal fin is set back on the body close to the caudal peduncle which is long and thin.

Distribution and Status

The threespine stickleback occurs throughout much of the northern hemisphere including most rivers in Europe, the Mediterranean and Black Seas, Iran in Asia, near Algiers in Africa, the Bering Sea to Korea and Mexico in the North Pacific, Hudson Bay to Chesapeake Bay in the North Atlantic, the states of Ohio, Wisconsin, Michigan, Massachusetts, California, and in the Great Lakes. They were introduced into the Great Lakes either by ballast water discharge from trans-oceanic ships, or they migrated south from Hudson Bay. Accidental bait bucket transfer is another possibility. They were first reported in Lake Huron in 1980. Since then, they have become widely dispersed throughout the Great Lakes, and more recently they have become present to common in several bays and river estuaries along the south shore of Lake Superior, and very abundant in southern Lake Michigan. Great Lakes dispersal was likely assisted by intra-lake shipping. In addition, during the early 1900s, stocking of threespine stickleback was advocated to control mosquitoes.

Life History

The threespine stickleback occurs in streams (especially estuaries), lakes, ponds, and the sea. In these waters, it is found over mud or sand with or without vegetation, but more so in association with vegetation. Threespine stickleback are multiple spawners with spawning occurring from March to October. Just prior to spawning, the male becomes brightly colored in orange or red, and builds a nest out of plant materials glued together by its renal secretions. The bright male coloration attracts a female and a courtship ritual takes place. Following the courtship ritual, the female usually lays 50-100 eggs in the nest, but is capable of laying as many as 300 eggs. The male then drives her away and fertilizes the eggs. One male can breed with several females concurrently. As many as 600 eggs have been observed in one nest at one time. The male guards the eggs and aerates them by disturbing the surface area of the nest and fanning his pectoral fins over the eggs. Spawning and hatching occur at a temperature range of 16-19˚C with hatching occurring in 7 days at the higher end of the temperature range. Just prior to egg hatch, the male scatters the eggs which enhances hatching success. The fry are guarded by the male until they are able to swim away. Threespine stickleback feed on a variety of fauna including zooplankton, oligochaetes (worms), macroinvertebrates (insect larvae), small fish, fish eggs, crustaceans, adult aquatic insects, and drowned aerial insects.

Invasion Impacts

Little is known about the potential impacts of the threespine stickleback, except that they compete with native sticklebacks for food and space, are bait stealers where abundant, and are known to prey on other fishes' eggs. Declines in trawl catches of native sticklebacks (mainly brook sticklebacks) have been observed in L'Anse Bay and Marquette Harbor, Michigan, while catches of threespine stickleback have increased in these two locations. However, there is no evidence to verify that the declines in native stickleback are due to the increases in the exotic stickleback. Angling quality has declined in southern Lake Michigan due in part to the large abundance of threespine stickleback.

Control

Little is known about controlling the threespine stickleback, nor does there appear to be interest in attempting control. Threespine stickleback are fished commercially in Scandinavia where they are processed into fishmeal and oil. Due to their extensive range and availability, they are also widely used as laboratory specimens for educational purposes. Their larger, sharp, rigid spines makes them less preferred prey than native sticklebacks, so predator enhancement would likely fail to control them and more likely impact native sticklebacks. They are unlikely to be used as bait, but they are very vulnerable to being accidentally captured by commercial bait collectors. Therefore, commercial bait collectors and retailers need to be able to recognize them and screen them out in order to prevent accidental availability in the public bait market.

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Life History and Invasion Impacts of the White Perch

Kingdom	Animalia -- Animal, animals, animaux
Phylum	Chordata -- chordates, cordado, cordés
Subphylum	Vertebrata -- vertebrado, vertebrates, vertébrés
Superclass	Osteichthyes -- bony fishes, osteíceto, peixe ósseo, poissons osseux
Class	Actinopterygii -- poisson épineux, poissons à nageoires rayonnées, ray-finned fishes, spiny rayed fishes
Subclass	Neopterygii -- neopterygians
Infraclass	Teleostei
Superorder	Acanthopterygii
Order	Perciformes -- perch-like fishes
Suborder	Percoidei
Family	Moronidae -- temperate basses
Genus	Morone Mitchill, 1814 -- striped basses
Species	Morone americana (Gmelin, 1789) -- baret, white perch

Description

The white perch is a perch-like fish in the family of temperate basses. Body length is usually 125 -175 mm (5-7 inches). The body shape is short horizontally and deep vertically. Edges of gill covers are slightly serrated. There are two dorsal fins; the anterior dorsal fin has rigid spines that increase in length to the third spine and then decrease; the posterior dorsal fin has softer spines, is the same height, but is shorter in length than the anterior dorsal fin. There is no gap between the two dorsal fins. Pectoral fins are rounded and soft. The lead spine on the pelvic fins is very rigid as are the first three spines on the anal fin, and both of these fins may be rose-colored. Scales are large. Color on upper body is dark gray-green to silver-gray fading to silver-green on sides, and to silver-white ventrally.

Distribution and Status

The native distribution of white perch includes the Atlantic seaboard from Maine to South Carolina, and the province of Quebec. The nonindigenous distribution of white perch includes the Great Lakes; Lake Champlain; the Missouri and Platte Rivers in Missouri, Kansas, Iowa, and

Nebraska; Smith Mountain Lake and Kerr Reservoir in Virginia; the upper Potomac River in West Virginia; the Illinois and Mississippi Rivers in Illinois; and the states of Colorado, Maine, Massachusetts, New Hampshire, New York, and Ohio. Introduction into some interior waterways of the U.S. was through intentional and accidental stockings. The likely vectors of introduction into the Great Lakes include natural migration through the Welland and Erie canals, and ballast water transfer from intra-lake shipping. White perch is classified as established in the Great Lakes, the Great Lakes states, and the states of Kentucky, Massachusetts, Vermont, Missouri, Nebraska, and New Hampshire However, with the exception of the Duluth-Superior harbor, abundance of white perch along the south shore of Lake Superior is rare, but more recently it is occurring in more locations.

Life History

White perch are bottom dwelling, semi-anadromous (live partly in rivers as well as larger lakes and bays) percids. They can live in freshwater or seawater, but prefer waters with less than 18‰ salinity. Habitat preference includes flats and channels of rivers and bays, moving to deeper channels during winter. However, no preference is shown for substrate, structure, or vegetation. Maturity occurs at age 2 in males and age 3 in females, and they spawn throughout the spring in rivers at water temperatures of 11-16˚C. Female fertility consists of 50,000-150,000 eggs, which are released over a period of 10-21 days. Fertilization of eggs occurs randomly, and hatching occurs over a period of 1-6 days. During summer, sub-adults tend to occupy river estuaries and streams, while adults tend to occupy deeper nearshore waters of bays and lakes, a behavior similar to the invasive percid, Ruffe (*Gymnocephalus cernuus*). Normal longevity is 6-7 years, and the maximum observed longevity is 17 years. Diet consists of zooplankton, macroinvertebrates (insect larvae), small fish, fish eggs, and small crustaceans including the invasive spiny waterflea (*Bythotrephes longimanus*).

Invasion Impacts

White perch may be directly impacting the forage fish base as well as some predators in places where they occur in the Great Lakes. Egg predation on native sport fish such as walleye (*Stizostedion vitreum*), with the resulting impact on recruitment, appears to be the most notable impact of white perch. Not only have white perch been documented to prey heavily on fish eggs, but they continue to feed on fish eggs for a long period of time. A collapse of the walleye fishery in the Bay of Quinte, Lake Ontario, is thought to be linked to white perch predation on walleye eggs. White perch have also been observed to prey heavily on spottail shiner (*Notropis hudsonius*) and emerald shiner (*Notropis atherinoides*), two important prey species in the Great Lakes. This may impact growth and limit abundance of native predators in localities where white perch are abundant. The introduction of white perch in a Nebraska reservoir may have led to the extirpation of black bullhead (*Ameiurus melas*) in that waterway. Diet overlap and heavy feeding on zooplankton by white perch was also linked to a growth decline in yellow perch (*Perca flavescens*) in western Lake Erie. Another concern is the hybridization occurring between white perch and the native white bass (*Morone chrysops*), which could dilute the gene pool of both species. Thus, the impacts of white perch appear to be far reaching, affecting native fish recruitment, available food for native adult and juvenile predator/prey species, and the genetic integrity of native white bass.

Control

Since white perch are already established in the Great Lakes and the Great Lakes states, the only element of control available is to prevent their further spread into inland lakes and streams, and maintain healthy native fish communities to prevent further increase of established white perch populations. Although white perch is an important commercial and sport fish in Chesapeake Bay and Maryland, it is currently considered an undesirable fish in the Great Lakes Basin. However, should a recreational and commercial white perch fishery develop in the Great Lakes, this would be a valuable control tool, especially in Green Bay of Lake Michigan. In Green Bay, white perch have been increasing in abundance, while the preferred yellow perch have significantly decreased in abundance. Educating anglers about the impacts of white perch and encouraging removal of captured specimens should also be included in any approach toward control.

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Life History and Invasion Impacts of the Fishhook Waterflea

Image by: Igor Grigorovich, C. pengoi. Male (left); Female (right)

Kingdom	Animalia -- Animal, animals, animaux
Phylum	Arthropoda -- arthropodes, arthropods, Artrópode
Subphylum	Crustacea Brünnich, 1772 -- crustaceans, crustáceo, crustacés
Class	Branchiopoda Latreille, 1817 -- branchiopodes, branchiopods
Subclass	Phyllopoda Preuss, 1951
Order	Diplostraca Gerstaecker, 1866
Suborder	Cladocera Latreille, 1829 -- cladocères, puces d'eau, water fleas
Infraorder	Onychopoda Sars, 1865
Family	Cercopagididae Mordukhai-Boltovskoi, 1968
Genus	Cercopagis G. O. Sars, 1897
Species	Cercopagis pengoi (Ostroumov, 1891) -- fishhook waterflea

Description

The fishhook waterflea is an exotic, predatory zooplankton similar to the spiny waterflea (*Bythotrephes longimanus*) and a member of the same family, Cercopagididae. It is 6-13 mm (1/4-1/2 inch) in length including the tail, and received the name “fishhook” due to the fact that its long tail ends in the form of a “hook”. Like the spiny waterflea, the spiny tail of the fishhook waterflea comprises 80% of its overall length, and has 3 pairs of small barb-like projections on the end of the tail next to the body. The female also has a pouch for carrying eggs, which is pointed on the end away from the body. Similar to the spiny waterflea, the fishhook waterflea is only visible to the human eye when numerous specimens collect and become enmassed on fishing lines, rods, and nets.

Distribution and Status

The fishhook flea is native to the Caspian Sea in Eurasia. From there, it spread across Europe, and then was likely transported to the Great Lakes in the ballast water of a trans-oceanic ship. The fishhook waterflea was discovered in Lake Ontario in 1998. It began to spread quickly with confirmed sightings in 6 New York state lakes and northern and southern Lake Michigan by 2000. In 2001, the fishhook waterflea was sighted in Muskegon Lake, Michigan, and in Lake

Erie near the entry to the Detroit River. However, since 2001, range expansion of the fishhook waterflea has stagnated, with no sightings in Lakes Huron and Superior.
The fishhook waterflea is only considered established in Lake Ontario. Occurrences in Lakes Erie and Michigan are not as widespread. There are no agencies monitoring the range and abundance of the fishhook waterflea specifically. Most reports relating to status are incidental, from anglers observing “large masses of cotton-like creatures like seeds from cottonwood trees” covering the water and fouling their fishing equipment. The same anglers report no problems with fouling of their fishing equipment just a few days later. Usually, no distinction is made between the spiny and fishhook waterfleas in angler reports. However, the fishhook waterflea has been implicated to be more problematic in fouling fishing equipment. Anglers in some Lake Erie locations have at times reported termination of fishing due to excessive fouling of their equipment. This suggests that high densities of the fishhook waterflea do occur in certain locations, and these high densities appear to be short-lived (limited to a few days).

Life History

The fishhook waterflea can reproduce asexually or sexually. One female can reproduce several times during the warm season, producing up to 13 eggs each time. She can also produce “resting” eggs, which lay dormant in the substrate during the winter and hatch in the spring. The fishhook waterflea is classified as a large form of zooplankton, and it feeds on smaller zooplankton such as *Daphnia* spp. Like all zooplankton, the fishhook waterflea is pelagic (lives in the water column), and in its native range, it has evolved to move deep in the water column where it is darker during the day to escape predation, and rise to near the surface at night.

Invasion Impacts

Species that can reproduce asexually are capable of becoming very abundant within a short period of time. Like the prolific spiny waterflea, the major ecological concern with the prolific fishhook waterflea is food competition with small forage fish and juvenile predator fish that rely on small zooplankton in their diet. Due to its long spiny tail, there is a behavior response by planktivorous fish to selectively avoid preying on the fishhook waterflea. Selective prey avoidance by predators will help to enhance fishhook waterflea abundance. In addition, with increased predation on small native zooplankton, the potential exists for small native zooplankton to begin to adopt a stronger diel response with regard to movement. If small zooplankton move deeper in the water column to escape increased predation, their rate of growth and overall growth is likely to decline. Fewer and smaller zooplankton are likely to affect growth and survival of forage fish and juvenile predator fish which will ultimately have far-reaching ecological implications, as well as a degrading effect on the sport and commercial fishing industry.

The negative impact of the fishhook waterflea on the economy of the Lower Great Lakes sport fishing industry has already been established. Walleye and yellow perch anglers and charter boats in some locations in Lake Erie have ceased fishing operations during high densities of fishhook waterflea. Anglers have had to cut their fish lines due to clumps of several hundred fishhook waterfleas attaching to their fishing lines and fouling the guides on fishing poles. The fishhook waterflea is reportedly more problematic in this respect than the spiny waterflea, due to the fishhook coil on the end of its tail.

Control

The fishhook waterflea is established in Lake Ontario, and will likely persist in North America. In addition, the dormant eggs can survive for long periods of time under adverse conditions. The success of the fishhook waterflea will likely be determined by interactions with predators and prey. It is hoped that fish which consume the spiny tail (generally fish older than one year of age) will learn to feed on the fishhook waterflea more heavily, and exert more biological control over waterflea populations, especially in helping to prevent high density outbreaks. Humans can assist in preventing range expansion of the fishhook waterflea by draining all water from watercraft and equipment to include bilges, live wells, bait buckets, outboard motors, transom wells, and nets, before traveling to other lakes. They should also dispose of contaminated line and nets that will not come clean. In addition, implementing one of the following three suggested operations: rinse boat and equipment with water greater than 40˚C; wash boat and equipment with minimal 250 psi water pressure; or dry boat and equipment for at least 5 days before launching in a new waterway.

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Life History and Invasion Impacts of the Quagga Mussel

Kingdom	Animalia -- Animal, animals, animaux
Phylum	Mollusca -- molluscs, mollusks, mollusques, molusco
Class	Bivalvia Linnaeus, 1758 -- bivalve, bivalves, bivalves, clams, mexilhão, ostra, palourdes
Subclass	Heterodonta Neumayr, 1884
Order	Veneroida H. and A. Adams, 1856
Superfamily	Dreissenoidea Gray, 1840
Family	Dreissenidae Gray, 1840
Genus	Dreissena Beneden, 1835
Species	Dreissena bugensis Andrusov, 1897 -- quagga mussel

Description

The quagga mussel is a small (<40 mm) bivalve mollusk related to the zebra mussel, but in comparison, the shell sides of the quagga mussel where the upper surface meets the bottom surface are more rounded, and the coloration near the shell hinge is lighter than the zebra mussel. In addition, the ventral side is convex; the valves are asymmetrical; and the light and dark bands appear more as concentric rings rather than stripes. Both quagga mussels and zebra mussels attach to hard surfaces via byssal threads, but the byssal threads of the quagga mussel subtend from a smaller groove than those of the zebra mussel.

Distribution and Status

The quagga mussel is native to the Ukraine area of western Russia. In North America, it was first discovered in 1989 near Port Colborne in Lake Erie. The quagga mussel occurs in all the Great Lakes except Lake Superior; the St. Lawrence River to Quebec City; and in some inland locations within the states of New York, Ohio, Michigan, and Pennsylvania. Some zebra mussels collected in Lake Superior were suspected to be quagga mussels, but their identification was never confirmed. The vector of introduction into the Great Lakes was likely the discharge of ballast water from a trans-oceanic ship. Cold water temperatures and low calcium concentrations during the majority of the year are limiting factors to their distribution in the northern Great

Lakes Basin. The primary natural vector of range expansion is water movement (currents, seiches, wind), which transports suspended veligers (larvae) as well as pediveligers, juveniles, and adults attached to floating debris. However, human activities are the most significant vector of range expansion, such as interconnection of waterways with canals and transport of veligers, juveniles, and adults attached to vessel hulls. Information is lacking on the current status of quagga mussel population densities.

Life History

Like zebra mussels, quagga mussels attach to hard surfaces, and prefer mesotrophic microhabitats consistent with pipe structures and other areas of constant water flow. Veligers are very sensitive to environmental conditions such as water turbulence, food availability, and chemical composition of the water. Veligers prefer calcium content greater than 30 mg/L; oxygen greater than 20% saturation; and pH range 7.4-9.4. However, quagga mussels can tolerate a wider range of water temperatures than zebra mussels, preferring 4-20˚C, and successfully reproducing down to 8˚C. In addition, quagga mussels can colonize directly on soft substrate, whereas zebra mussels require hard objects on mud or sand to initiate colonization.

The quagga mussel is dioecious. Development of eggs and sperm occurs during the cold season. Spawning begins in spring at a water temperature of 10-15˚C, peaks in summer at 20-22˚C, and declines in fall as water temperature decreases. Depending on environmental conditions, spawning may last from 4-8 months. Successful reproduction is dependent on water temperature exceeding 8˚C, and calcium ion concentration of the water exceeding 20 mg/L. Veligers form one day after fertilization, and evolve into five distinct planktonic forms over a period of 1 to 4 weeks. The fifth larval form, pediveliger, initiates search for substrate attachment. Prior to this stage, veligers are pelagic (suspended in the water column). Pediveligers become juveniles once attached to substrates. Juvenile quagga mussels mature at a length of 5-10 mm. Female fecundity ranges from 10,000 eggs to over 1,000,000 eggs. Quagga mussels are effective filter feeders, removing suspended particles, primarily phytoplankton and some small zooplankton from the water. Particles that are consumed pass through the digestive system, and the unused portion is excreted as feces. Undesirable particles are encased in mucus and released as pseudofeces. An adult quagga mussel can filter approximately one liter of water per day. Quagga mussels benefit from moving water in that they generally receive more food items and higher oxygen concentrations than in still water locations. Quagga mussels use their byssal threads to attach to hard underwater objects including crayfish, native mussels, and human-made structures, but they can also exist on soft mud by connecting to each other and forming a mat. The streamlined design of its shell aids the quagga mussel in remaining attached to an object in current.

Invasion Impacts

Quagga mussel impacts are the same as the zebra mussel, except they would be less likely to cut human skin due to the rounded edges on their shells. The most serious economic impact of quagga mussels is colonization on the inside surface of water pipes, including those pipes used to transport drinking water. Enmassed colonies obstruct water flow through pipes, and may even block water flow altogether. Not only do masses of attached mussels on the inside surface of pipes obstruct water flow, but the attachment also facilitates an increased rate of surface corrosion on pipes, docks, breakwalls, boat hulls, and outboard engine lower drive units.

Competition for phytoplankton and the encrustation of native mussels are two of the most common ecological impacts from quagga mussels. Phytoplankton is necessary for survival of larval and juvenile fish and zooplankton. Dense mats of mussel colonies reduce available forage area for benthic fish. However, impact on fish populations is not consistent. Quagga mussels can completely encrust the shells of native mussels, disabling their capacity to feed.

Filter feeding results in increased water clarity and bioaccumulation of contaminants. Increased water clarity increases light penetration, which encourages rooted vegetation growth. Proliferation of aquatic macrophytes has cascading ecological effects, both positive and negative. Contaminants are passed to the top of the food chain, increasing exposure to wildlife and humans.

Control

Population removal or reduction may be feasible with chemical treatment in small landlocked waters, but there is no known selective treatment. The most effective method to control quagga mussels is to prevent their spread. Quagga mussels attach to vegetation as well as hard surfaces, so recommended measures to prevent their spread include 1) removal of all visible plant and animal material from boats, trailers, outboard motors, and accessory equipment that has come in contact with water; 2) washing with 104˚F water or pressure washing these same surfaces; 3) allowing them to air dry for a minimum of 5 days before recontact with water; 4) draining on land live wells, bilge water, transom wells, and bait buckets; 5) flushing fresh water through the engine cooling system; and 6) learning identification and reporting suspected sightings to the agency of jurisdiction.

Monitoring can aid in the early detection of quagga mussels and facilitate prompt response. Kits are available from State Sea Grant Programs for monitoring veligers. Adults can be monitored by submerging hard objects and checking them periodically during summer and fall, or by checking boat hulls, dock supports, buoys, and shoreline rip-rap. Concentrate monitoring primarily in locations where boats are launched and moored, and where there is an abundance of hard substrate.

The exotic invasive round goby (*Neogobius melanostomus*) preys on the smaller juvenile mussels. Rusty crayfish (*Orconectes rusticus*) are known to prey on veligers. Streams with substantial densities of rusty crayfish may be able to delay colonization of quagga mussels. However, due to the reproductive capability of quagga mussels, biological control is not an effective management tool, and introducing an exotic invasive species to control another exotic invasive is not an acceptable management practice.

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Life History and Invasion Impacts of the Rusty Crayfish

Kingdom	Animalia -- Animal, animals, animaux
Phylum	Arthropoda -- arthropodes, arthropods, Artrópode
Subphylum	Crustacea Brünnich, 1772 -- crustaceans, crustáceo, crustacés
Class	Malacostraca Latreille, 1802
Subclass	Eumalacostraca Grobben, 1892
Superorder	Eucarida Calman, 1904 -- camarão, caranguejo, ermitão, lagosta, siri
Order	Decapoda Latreille, 1802 -- crabes, crabs, crayfishes, crevettes, écrevisses, homards, lobsters, prawns, shrimp
Suborder	Pleocyemata Burkenroad, 1963
Infraorder	Astacidea Latreille, 1802
Superfamily	Astacoidea Latreille, 1802
Family	Cambaridae Hobbs, 1942 -- crayfishes
Subfamily	Cambarinae Hobbs, 1942
Genus	Orconectes Cope, 1872
Subgenus	Orconectes (Procericambarus) Fitzpatrick, 1987
Species	Orconectes rusticus (Girard, 1852) -- rusty crayfish

Description

Rusty crayfish can be distinguished from native crayfish by the presence of black bands on their claw tips; an oval gap at the base of the claws when claws are closed; larger claws with smooth surface, gray-green to red-brown in color; and a dark rusty colored spot on each side of their carapace. However, the rusty spot on the carapace is not always present. Maximum length is approximately 100 mm (4 inches), excluding claws; claw length is approximately 63 mm (2.5 inches). Other features that distinguish rusty crayfish from native crayfish are the absence of a dark patch on the dorsal side of its abdomen, and the absence of white, wart-like bumps on its claws. Males have a pair of hooks on one pair of their legs for use in grasping females during mating.

Distribution and Status

Rusty crayfish are believed to be native to the Ohio River Valley and the states of Ohio, Kentucky, Tennessee, Indiana, and Illinois. From here, they were likely spread by bait bucket transfer (transported and used by anglers as fish bait, with unused specimens released alive into a waterway). Rusty crayfish are currently widely distributed throughout the northeast and Midwest U.S. and Ontario.

Commercial trappers may also have contributed to range expansion in Wisconsin, by intentionally introducing the crayfish into several Wisconsin lakes to increase harvest. Marketers also sold crayfish to schools for educational purposes, and although shipping containers had written warnings not to release the crayfish, the warnings may have been disregarded.

Rusty crayfish can become overabundant in some lakes with observed densities up to 50 per square meter Similarly, a single 5-minute bottom trawl tow in Traverse Bay, Lake Michigan, yielded in excess of 150 rusty crayfish.

Life History

Rusty crayfish are found in lakes, streams, and ponds; in fast water or non-moving water; and over any substrate. They prefer objects for cover such as rocks and logs, and they require a year-round water supply, as opposed to intermittent streams and ponds.

Rusty crayfish usually breed in early spring, but breeding may also occur in late summer or early fall. Males transfer their sperm to the female, and the female stores it until she is ready to release her eggs (April/May). Females lay 80-575 eggs; eggs and sperm are released together; and fertilization occurs externally at this time. After hatching, the young molt (old shells shed to facilitate growth) several times while remaining attached to the female. Maturity usually occurs at approximately one year, and a length of 34 mm (1.375 inches) excluding claws. After maturity, growth slows, requiring females to molt only once per year. Males grow larger than females, which necessitates two molts per year. The rusty crayfish diet consists of aquatic vegetation, benthic invertebrates, detritus, fish eggs, small fish, and larval zebra mussels (veligers), when available. The life expectancy of a rusty crayfish is 3-4 years.

Invasion Impacts

The most serious impact of rusty crayfish is the removal of aquatic vegetation. Aquatic vegetation encourages production of zooplankton, provides shelter for young fish and habitat for some adult fish, provides nesting material and cover for spawning fish, and helps to prevent shoreline erosion by reducing wave impact.

Rusty crayfish out-compete native crayfish for food (benthic invertebrates) and cover, and are better at avoiding predators. Rusty crayfish consume more food than native crayfish due to their larger size and higher metabolic requirements, and leave less food behind for native crayfish, juvenile fish, and forage fish. Rusty crayfish are also more aggressive than native crayfish, and they hold their ground and defend themselves with their claws when threatened by predators. Native crayfish retreat in the face of a potential predator, and abandon their hiding places when confronted by a rusty crayfish. These two behavior characteristics make native crayfish more susceptible to predation. The result is that native crayfish are displaced, or abundance declines when in association with rusty crayfish.

The impact of rusty crayfish on fish recruitment is unproven. Predation on fish eggs and subsequent decline in fish recruitment has been observed and documented in some locations, but not in all locations following a rusty crayfish invasion. One speculation for this selectivity relates to water temperature. Fish that spawn during the warm water season are more susceptible to egg predation by rusty crayfish than fish that spawn during the cold water season. However, this hypothesis has been shown to be inconsistent, and the reason for fish egg predation in selective locations remains unknown. There is no research evidence proving that rusty crayfish impact fish populations.

Control

Chemicals that selectively kill crayfish exist, but are not registered for use. No chemicals are available that kill only rusty crayfish. Trapping may potentially reduce populations sufficiently to allow some recovery of aquatic vegetation. The U.S. Fish and Wildlife Service, the U.S. Forest Service, and MIDNR are planning an experiment to reduce a rusty crayfish population in a Michigan lake by bottom trawling. The goals of this experiment are to reduce the crayfish population sufficiently to initiate recovery of aquatic vegetation and regain biological control of the crayfish by the fish community. No technology currently exists that will selectively extirpate rusty crayfish from a waterway. The best method to control rusty crayfish is prevention of range expansion by fish bait regulation and education of anglers, bait dealers, and other public users of crayfish.

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Life History and Invasion Impacts of the Spiny Waterflea

Photo by Pieter Johnson, Center for Limnology, September, 2003

Kingdom	Animalia -- Animal, animals, animaux
Phylum	Arthropoda -- arthropodes, arthropods, Artrópode
Subphylum	Crustacea Brünnich, 1772 -- crustaceans, crustáceo, crustacés
Class	Branchiopoda Latreille, 1817 -- branchiopodes, branchiopods
Subclass	Phyllopoda Preuss, 1951
Order	Diplostraca Gerstaecker, 1866
Suborder	Cladocera Latreille, 1829 -- cladocères, puces d'eau, water fleas
Infraorder	Onychopoda Sars, 1865
Family	Cercopagididae Mordukhai-Boltovskoi, 1968
Genus	Bythotrephes Leydig, 1860
Species	Bythotrephes longimanus Leydig, 1860 -- spiny waterflea

Description

The spiny waterflea is an exotic, predatory zooplankton that is approximately 13 mm (0.5 inches) long. The tail spine is the distinguishing characteristic of this zooplankton, containing one to four pairs of barbs, and comprising 70-80% of its overall length. The head consists of a single, large black eye, and a pair of sickle-shaped mandibles. One pair of antennae, posterior to the head, assists with movement through the water. It has four pairs of legs that perform two functions. The first pair of legs is longer than the posterior pairs, and is used to capture prey. The shorter posterior legs are used to hold prey during feeding.

Distribution and Status

The spiny waterflea is believed to be native to the British Isles, Scandinavia, northern Europe, and Russia. It was first discovered in Lake Huron in 1984, and the pathway of introduction is thought to be ballast water discharge from trans-oceanic shipping. Resting eggs can lay dormant in ballast tank sediment for a long period of time, and be subsequently dislodged and released into the ballast water during exchange operations. Genetic studies confirm a relationship of the North American specimens to the population near St. Petersburg, Russia.

By back calculating growth and mortality, Sprules et al. (1990) estimate that actual introductions into the Great Lakes likely occurred during the late 1970s and early 1980s. The spiny waterflea has expanded its range to include all the Great Lakes and several inland lakes. Inter-lake spreading has been enhanced by attachment to boats and fishing equipment.

Discovery reports of spiny waterflea across the Great Lakes occurred at the rate of one lake per year. After discovery in Lake Huron, the spiny waterflea was reported in the Lower Great Lakes in 1985, Lake Michigan in 1986, and Lake Superior in 1987. Temperature is a key factor in determining location and abundance of the spiny waterflea. It moves deeper as surface waters approach 25˚C during summer. Since western Lake Erie averages only 7.3 m (24 feet) in depth, spiny waterflea is absent from this area for most of the summer as the entire water column warms toward 25˚C. This temperature limit would also apply to embayments in the Great Lakes and inland lakes. Abundance reaches a climax in late summer and fall. In 1987, the density of spiny waterflea in Lake Michigan was reported to be comparable to densities in its native range, 6.5 per cubic meter. Densities are reported to be low in Lake Ontario, southern Lake Michigan, and offshore in Lake Superior. Densities are reported to be moderate in Lake Huron, and very high in central Lake Erie. Future status is likely to remain stable unless there are significant changes in environmental conditions, especially water temperature.

Life History

The spiny waterflea is pelagic, occupying the upper water column of large and small freshwater and brackish lakes. In Lake Michigan, it occupies depths from 10 to 20 meters (33 to 66 foot). During the day it drops lower in the water column and rises to the 10 meter level at night. It is believed that this daily movement was adapted to avoid predation. The spiny waterflea prefers a temperature range of 10-24˚C, and a salinity range of .04-.4‰.

The spiny waterflea can reproduce asexually or sexually. Water temperature drives the reproductive cycle and determines sex indirectly. The spiny waterflea feeds on small zooplankton, consuming up to 20 specimens per day. When lake water is warm during summer, small zooplankton become abundant, and only female waterfleas are produced asexually. Warmer water continues to produce abundant prey, and the females continue to reproduce asexually, producing one to ten eggs each within a pouch. Within the pouch, the eggs hatch, and the identical female embryos (clones) become fully developed in less than two weeks. During fall, the females detect cooling in the water, food declines, and some males are produced. The males mate with the remaining females who then produce resting eggs. These eggs are carried for a short time in the pouch on the female, and then released to fall, hopefully, into soft substrate where they can survive the winter. During spring when water temperature warms, eggs that were dormant (resting) in the soft substrate of the lake hatch, and the cycle repeats. Life longevity can last up to three weeks. Food consumption with reference to body weight is comparable to other zooplankton.

Invasion Impacts

The spiny waterflea competes with juvenile fish for food, but there is disagreement as to the significance of its impact on native zooplankton abundance. *Daphnia* communities in the Great Lakes have decreased since the appearance of the spiny waterflea. In reference to the general decline of alewife (*Alosa pseudoharengus*) in all the Great Lakes except Lake Superior, and possibly the decline of juvenile yellow perch (*Perca flavescens*) in southern Lake Michigan, the spiny waterflea is likely a contributing factor. Due to the long spines, juvenile fish quickly learn to avoid preying on spiny waterflea. Those juvenile fish that do consume it are likely to die from spines penetrating their stomach walls. This selectivity for prey other than spiny waterflea also contributes to enhancement of spiny waterflea populations. Quality of sport fishing is also impacted from clusters of spiny waterfleas attaching to fishing line and fouling fishing equipment. This impact has become so significant that charters and anglers have suspended fishing during periods of high waterflea abundance.

Control

Due to the optimal environmental conditions of the lakes in the Great Lakes Basin and the prolific reproductive ability of the spiny waterflea, it will persist here. Its success will likely be determined by interactions with predators and prey. Many adult fish prey on spiny waterflea including salmonids, percids, coregonids, shiners, and sculpins. Sufficient predation by adult fish may prevent spiny waterflea populations from exploiting important native zooplankton populations such as *Daphnia*. Dormant eggs can survive for long periods of time under adverse conditions. Humans can assist in preventing range expansion of spiny waterflea by draining all water from watercraft equipment to include bilges, live wells, bait buckets, outboard motors, transom wells, and nets before traveling to other lakes.

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Life History and Invasion Impacts of the Zebra Mussel

Kingdom	Animalia -- Animal, animals, animaux
Phylum	Mollusca -- molluscs, mollusks, mollusques, molusco
Class	Bivalvia Linnaeus, 1758 -- bivalve, bivalves, bivalves, clams, mexilhão, ostra, palourdes
Subclass	Heterodonta Neumayr, 1884
Order	Veneroida H. and A. Adams, 1856
Superfamily	Dreissenoidea Gray, 1840
Family	Dreissenidae Gray, 1840
Genus	Dreissena Beneden, 1835
Species	Dreissena polymorpha (Pallas, 1771) -- zebra mussel

Description

The zebra mussel is a small (<50 mm) bivalve mollusk which has a triangular shell with alternating light and dark bands that resemble the mammalian zebra, although some shells may be entirely light or dark. Its introduced cousin, the quagga mussel (*Dreissena bugensis*), looks similar, but the zebra mussel can best be distinguished by its shell morphology. The shell edge of the zebra mussel, where the top surface meets the bottom surface, is angular (forms a ridge), and that of the quagga mussel is rounded. Zebra mussels vary in shell pattern and color, hence the species name, *polymorpha*.

Distribution and Status

Zebra mussels are native to western Russia, particularly in the area of the Caspian Sea. Human-built canals allowed expansion into Europe. Zebra mussels were discovered in Lake St. Clair of the Great Lakes in 1988, with introduction believed to have occurred in the mid 1980s. The vector of introduction was likely the discharge of ballast water from a trans-oceanic ship. This initial population is believed to have been transported from the Black and Caspian seas, since two nonindigenous, introduced fish, the round and tubenose goby (*Neogobius melanostomus* and *Proterorhinus marmoratus*), originated from these waters, and were introduced into the Great Lakes at about the same time.

The introduction was likely supported by a large number of individuals given the genetic diversity of zebra mussels and the extensive range (75 km x 25 km) of the initial population. Presently, zebra mussel range spans from the Great Lakes down the Mississippi River to the Gulf of Mexico, and from the middle Atlantic states to as far west as Nebraska, Kansas, and Oklahoma, and in many inland lakes and streams. Within the Great Lakes Basin, they occur in more locations in Lower Michigan and southern Lake Michigan with the least number of locations in Minnesota (eastern) and Lake Superior (south shore). Cold water temperatures and low calcium concentrations during the majority of the year are limiting factors to their distribution in the northern Great Lakes Basin. The primary natural vector of range expansion is water movement (currents, seiches, wind) which transports suspended veligers (larvae), as well as juveniles, and adults attached to floating debris. However, human activities are the most significant vector for range expansion. Of these, ballast water transport, interconnection of waterways with canals, and transport of veligers, juveniles, and adults attached to vessel hulls are the most prominent dispersal mechanisms. Maximum densities have reached 700,000 per square meter in the Great Lakes and 100,000 per square meter in Europe. Average population density is 30,000 per square meter. Zebra mussels tend to be smaller (<35 mm), slower growing, and live longer in Europe than in the Great Lakes (shells >40 mm, 2-3 year lifespan). Year to year fluctuation of population size is common, due primarily to environmental conditions and lack of veliger protection by the adults. The environmental conditions include turbidity, food availability, water temperature, water chemical characteristics, chlorophyll *a* concentration, and availability of hard substrate.

Life History

Zebra mussels, especially juveniles, prefer mesotrophic microhabitats consistent with pipe structures, areas of consistent water flow, and subtle pressure changes. It is unknown exactly why mussels prefer pipe structures. Veligers are very sensitive to environmental conditions such as water turbulence and food availability. Veligers also prefer the following chemical composition of the water: calcium content > than 30 mg/L; oxygen > than 20% saturation; and pH range 7.4-9.4.

The zebra mussel is dioecious (either male or female) and also known to be hermaphroditic. Development of eggs and sperm occurs during the cold season. Spawning begins in spring at a water temperature of 10-15˚C, peaks in summer at 20-22˚C, and declines in fall as water temperature decreases. Depending on environmental conditions, spawning may last from 4-8 months. Successful reproduction is dependent on water temperature exceeding 10˚C, and calcium ion concentration of the water exceeding 20 mg/L. Veligers form one day after fertilization, and develop into five distinct planktonic forms over a period of 1 to 4 weeks. The fifth larval form, pediveliger, initiates a search for substrate attachment. Prior to this stage, veligers are pelagic (suspended in the water column). Pediveligers become juveniles once attached to a substrate. Juvenile zebra mussels mature at lengths of 5-10 mm, and maturity does not usually occur until the following warm season. Female fecundity ranges from 10,000 eggs to over 1.5 million eggs. Zebra mussels feed by filtering suspended particles (phytoplankton) from the water, and they are very effective filter-feeders. An adult zebra mussel filters approximately one liter of water per day. Particles that are used to sustain life requirements are excreted out the excurrent siphon as feces, but unused particles are encased in mucus and ejected out the incurrent siphon as pseudofeces. Due to the presence of zebra mussels, water clarity in western Lake Erie increased 85%, while chlorophyll *a* decreased 43% in one year. Because they are filter feeders, water movement around zebra mussels plays a critical role in their success. Faster-moving water provides a higher supply of food, oxygen, and calcium, and this factor assists in defining their distribution, status, and attraction to water-inflow/outflow pipes (water flowing through pipes). Zebra mussels use their byssal threads to attach to hard underwater objects, including crayfish, native mussels, and human-made structures, but they can also exist on soft mud by connecting to each other and forming a mat. Their byssal threads consist of two types, temporary and permanent. The temporary byssal threads are longer and fewer in number than the permanent threads, allowing the mussel to detach them easily from one object in search of a more suitable object. The flat ventral surface and streamline design of its triangular shell aids the zebra mussel in remaining attached to an object in current.

Invasion Impacts

The most serious economic impact of zebra mussels is colonization on the inside surface of water pipes, including those pipes used to transport drinking water. Enmassed colonies obstruct water flow through pipes, and may block water flow altogether. The annual cost to remove zebra mussels from water pipes likely exceeds $400 million per year in the Great Lakes and several billion in North America. Not only do masses of attached mussels on the inside surface of pipes obstruct water flow, but the attachment also facilitates an increased rate of surface corrosion on pipes, docks, breakwalls, boat hulls, and outboard engine lower drive units.

Competition for phytoplankton and encrustation on native mussels are two of the most common ecological impacts from zebra mussels. Phytoplankton is necessary for survival of larval and juvenile fish and zooplankton. A reduction in plankton abundance is one factor which has implicated the zebra mussel in the decline of yellow perch recruitment in southern Lake Michigan. Dense mats of mussel colonies reduce available forage area for benthic fish. However, impact on fish populations is not consistent. Some biologists report a shift in diet or habitat use by fish communities in association with zebra mussels, but observe no negative impacts. Zebra mussels can completely encrust the shells of native mussels, disabling their capacity to feed. As a result, several native mussel populations including rare and endangered species have declined severely or are near extirpation.

Filter feeding also results in increased water clarity and blue-green algal blooms. Increased water clarity increases light penetration, which encourages rooted vegetation growth. An increase in vegetation decreases the water area available for swimming and boating. Dense mats of vegetation also change the nature of fish habitat. During filter feeding, zebra mussels selectively reject blue-green algae and discard them back into the water column, while other algae are consumed and their abundance is reduced. This selective feeding allows blue-green algae to perpetuate and form algal blooms. Zebra mussel shells have sharp edges. Attached mussels in swimming areas and empty shells on beaches can cut human skin, resulting in degradation of recreational experiences and decline of the resource use through avoidance of these infested areas.

Control

Population removal or reduction may be feasible with chemical treatment in small landlocked waters, but there is no known selective treatment. The most effective method for controlling zebra mussels is to prevent their spread. Recommended measures to prevent the spread of zebra mussels include the following: removal of all visible plant and animal material from boats, trailers, outboard motors, and accessory equipment that has come in contact with water; washing with 104˚F water or pressure washing these same surfaces, or allowing them to air dry for a minimum of 5 days before recontact with water; draining of live wells, bilge water, transom wells, and bait buckets on land; flushing fresh water through the engine cooling system; learning identification and reporting new suspect sightings to the agency of jurisdiction.

Monitoring can prevent the spread of zebra mussels by allowing early detection and response. Kits are available from State Sea Grant Programs for monitoring veligers. Adults can be monitored by submerging hard objects and checking them periodically during summer and fall, or by checking boat hulls, dock supports, buoys, and shoreline rip-rap. Monitoring should begin when the water temperature exceeds 10˚C, primarily in locations where boats are launched and docked, and where there is an abundance of hard substrate.

The exotic invasive round goby is known to prey on the smaller juvenile mussels, and the rusty crayfish (*Orconectes rusticus*) is known to prey on veligers. The rate of round goby range expansion may in part be dependent on the availability of zebra mussels. Streams with substantial densities of rusty crayfish may be able to delay colonization of zebra mussels. However, due to the reproductive capability of zebra mussels, biological control is not an effective management tool, and introducing an exotic invasive species to control another exotic invasive is not an acceptable management practice.

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Life History and Invasion Impacts of the Curly-leaved Pondweed


Kingdom	Plantae -- Planta, plantes, plants, Vegetal
Subkingdom	Tracheobionta -- vascular plants
Division	Magnoliophyta -- angiospermes, angiosperms, flowering plants, phanérogames, plantes à fleurs, plantes à fruits
Class	Liliopsida -- monocotylédones, monocotyledons
Subclass	Alismatidae
Order	Najadales
Family	Potamogetonaceae -- pond weed, pondweed
Genus	Potamogeton L. -- pondweed
Species	Potamogeton crispus L. -- curly pondweed, curly-leaved pondweed

Description

Curly-leaved pondweed is an aquatic, submergent (growing below the water surface), rooted, perennial herb. The stem is flattened, and the leaves are long, narrow, crinkled, and alternate with finely toothed edges.

Distribution and Status

Curly-leaved pondweed is native to Europe, Asia, northern Africa, and Australia. It is found throughout the U.S. in both fresh and brackish water, and overall occurrence is reported as common.

Life History

For curly-leaved pondweed, new growth begins in the fall (Sep.-Oct.), from seed pods called turions, vegetative propagules that form and harden at the stem tips. Stems germinate from the turions, and grow until the water is covered by ice; growth then slows. At ice break-up, growth increases rapidly such that the plant tips break the water surface by mid-May, and the plant begins to develop the turions. By late May or early June, a flower develops above the water and all growth stops. The plant then dies, and falls to the bottom as early as mid-June. The turions separate from the dead stems, allowing them to be moved around by currents and wave action. Other turions just lie where they fall, and the cycle starts over again in September. Curly-leaved pondweed can inhabit lakes, rivers, and ponds, and prefers fertile, hard, shallow water. However, it can survive in low light, and therefore it may be found in deep water as well.

Invasion Impacts

Curly-leaved pondweed has several ecological and economic impacts. It can out-compete native aquatic plants, significantly reducing native plant diversity and abundance, and even eliminating them. It has several competitive advantages over native plants, including a quick-developing, extensive root system, growth through fall, winter, and spring dying back during early summer, formation of dense mats of vegetation that grow above and shade out native plants, and the ability to grow over a wide temperature range. The dense stands of underwater stems (up to 2,000 stems per square yard) and the dense mats of surface vegetation obstruct water intakes, and restrict or eliminate recreational activities (boating, swimming, fishing) and fish movement. The early summer die-back provides additional phosphorous for algae blooms. One acre of curly-leaved pondweed contains 2.5 kg (5.5 pounds) of phosphorous. In addition, the dead plants fall to the bottom and consume oxygen. The depletion of deepwater oxygen causes phosphorous to be released from the substrate. The dead plants also wash on to shorelines, giving off a strong odor and making beaches unusable. These problems can lead to increased maintenance costs to keep boat launches, channels, beaches, and water intakes open, dead plant material removed, and a loss of revenue from a decrease in recreational use. However, curly-leaved pondweed is not always a problem where it occurs. In some lakes, it grows in association with native plants and does not cause any problems.

Control

Control of curly-leaved pondweed is divided into three categories: manual, chemical, and habitat alteration. Whatever method is chosen, it should be initiated during spring to disrupt production of the turions, and have maximum effectiveness.

Manual removal consists of cutting, raking, or harvesting with mechanical harvesters. Manual techniques are effective only for the short-term and for small areas. The effectiveness of manual removal depends on the proximity of the cutting or harvesting to the substrate, which both affect the rate of re-growth. However, if cutting or harvesting is performed during spring before the turions are formed, curly-leaved abundance is more susceptible to decline over the long-term. Manual cutting with boat-towed cutters costs $200 for a cutter and $10-30/acre in operational cost. The cost of a mechanical harvester is $300-600/acre. Four lakes in Minnesota achieved 50% control after three years of spring cutting or harvesting.

Chemical treatment is most effective for treating small areas or spot removal, but it is also short-term. Diquat and endothall are effective herbicides, but only during the year in which they are used. Die-off occurs in early summer with the use of herbicides, so algae blooms are still enhanced. The cost history of chemical treatment in Minnesota is $200-400/acre.

Water draw-down can also be used to control curly-leaved pondweed if the sediment is exposed, but effectiveness is short-term. One to three years at a cost of $15,000+, and there will likely be changes to the fish community. One lake in Minnesota achieved 95% control covering 80 acres by implementing a water draw-down, but the curly-leaved pondweed has been slowly increasing since the draw-down.

No information was found describing any potential bio-control of curly-leaved pondweed. The key to long-term control of curly-leaved pondweed involves manipulating the production of, or eliminating the turions.

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Life History and Invasion Impacts of Eurasian Watermilfoil

Kingdom	Plantae -- Planta, plantes, plants, Vegetal
Subkingdom	Tracheobionta -- vascular plants
Division	Magnoliophyta -- angiospermes, angiosperms, flowering plants, phanérogames, plantes à fleurs, plantes à fruits
Class	Magnoliopsida -- dicots, dicotylédones, dicotyledons
Subclass	Rosidae
Order	Haloragales
Family	Haloragaceae -- water milfoil
Genus	Myriophyllum L. -- water milfoil, watermilfoil
Species	Myriophyllum spicatum L. -- Eurasian water-milfoil, Eurasian watermilfoil, myriophylle en epi, spike watermilfoil, spiked water milfoil

Description

Eurasian watermilfoil is an aquatic, submergent (growing below the water surface), rooted, perennial herb that grows up to 4.5 m (15 feet) in length. It has a long, narrow, lax, cordlike stem usually surrounded by 4 whorls (sometimes 3-5 whorls) of pinnate (feather-like) leaves. Each leaf consists of 12-21 pairs of leaflets that droop when out of water. Flowers are small, not noticeable, unisexual with bisexual flowers developing sometimes, in whorls of 4s, emergent (rising above the surface of the water), and red in color. Fruit is globular, and contains 4 seeds. Stems and leaves near the top of the plant are also usually red in color.

Distribution and Status

Eurasian watermilfoil is native to Europe, Asia, and northern Africa. It arrived in North America sometime between the early 1800s and the early 1940s depending on the authority that is referenced. The plant is capable of propagating from stem fragmentation. Therefore, the initial introduction in North America could have been through the aquarium market, ballast water, or even by migrating waterfowl. Subsequent spreading has occurred by attachment of stem fragments to boat propellers, boat equipment appendages, and boat trailers. In North America, Eurasian watermilfoil occurs in the east, Midwest, and west coast, but is generally absent in the central plains.

Life History

Growth begins in early spring from the roots. Once the stems reach the surface, they branch, forming dense mats of vegetation on and near the surface. Eurasian watermilfoil reproduces both sexually and asexually. Asexual reproduction occurs in three ways; through buds that form on the root crown during spring, the development of unisexual flowers, and by stem fragmentation during summer. On the bisexual spikes, female and male flowers occur together. Both wind and insects are the primary agents of pollination. Flowering occurs from July to August, and a second flowering may occur if the first flowering occurs early enough. Although four million seeds can be produced in abundant areas, the seeds germinate erratically at temperatures > 15˚C, so the primary method of reproduction is considered to be natural fragmentation of the stem and leaves. Autofragmentation occurs after flowering. Eurasian watermilfoil occurs in lakes, rivers and ponds as deep as 8 m (26 feet) in clear water. It prefers eutrophic (nutrient-rich) conditions, and depths of 0.5 m (1.6 feet) to 3.5 m (11.5 feet). It can grow in various substrates, and can tolerate very alkaline (pH 10) or saline (10‰) conditions. The plant prefers sites void of existing vegetation to propagate a new colonies; it has difficulty becoming established in areas with established native plant communities.

Invasion Impacts

Eurasian watermilfoil has several ecological and economic impacts. It can out-compete native aquatic plants, significantly reducing native plant diversity, abundance, and even eliminating them. It has several competitive advantages over native plants, including an extensive, quick developing root system, growth beginning in early spring, formation of dense mats of vegetation that grow above and shade out native plants, and the ability to grow over a wide temperature range. Eurasian watermilfoil provides less forage value for plant-eating waterbirds than native plants, and harbors fewer invertebrates for planktivorous fish. The dense stands of underwater stems and the dense mats of surface vegetation can also obstruct water intakes, restrict or eliminate recreational activities (boating, swimming, fishing), provide habitat for mosquitoes, reduce dissolved oxygen and fish abundance, and foul beaches when they die. These problems can lead to increased maintenance costs to keep boat launches, channels, and water intakes open; dead plant material removed; a decline in property values; and a loss of revenue from decreased recreational use.

Control

Eurasian watermilfoil cannot be totally eliminated from a lake once established. Control concentrates on reducing the plants’ impacts, and preventing further spread. The process of reducing the plants’ impacts is usually divided into three categories: manual, chemical, and biological.

Manual removal consists of hand pulling, raking, harvesting with mechanical harvesters, and water drawdown. Manual techniques are effective only for the short term and usually for small areas such as boat launches and access channels. They are used to create temporary access, and their effectiveness depends on whether the roots are removed or the proximity of the cutting to the substrate, which affect the rate of regrowth. The cost of a mechanical harvester ranges from $300 to $600 per acre. A major drawback with a mechanical harvester is that thousands of stem fragments are produced to spread and propagate new introductions. Water drawdown in the fall exposes the plant to freezing, which has had some limited success.

Chemical treatment is most effective for treating small areas or spot removal, and it is also short term. The most effective herbicides for reducing or retarding Eurasian watermilfoil are 2-4-D fluridone and trichlopyr. Selectivity with these herbicides is possible but difficult. The cost of chemical treatment is $200 to $2,000 per acre.

The use of biological control agents is currently undergoing research, and the species receiving most attention is the native milfoil weevil (*Euhrychiopsis lecontei*). This weevil is native to North America, and preys specifically on watermilfoil. The weevil lives underwater, and lays its eggs on milfoil plants. The hatched larvae retard milfoil growth by mining the stem interior. Results with this weevil have been mixed; it has been effective in some lakes and not in others. The reasons for this selective effectiveness are unknown and remain under investigation. Despite the mixed results, research continues, because the potential advantages of bio-control are long term control, at a reduced cost, and safety to non-target native plants, animals, and humans.

Removing visible plant material from boats, motor propellers, and boat trailers is essential in preventing the further spread of Eurasian watermilfoil. In addition, bilge wells, live wells, gunnel channels, and motor cooling systems should be drained and flushed before departing the boat launch area.

Maintain established populations of native plants; established native plant communities help to prevent the establishment of Eurasian watermilfoil.

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Appendix 3. Aquatic sampling within and near Great Lakes Inventory and Monitoring Network park units.

Species abbreviations: CLP – curly leaf pondweed; EUM – Eurasian water milfoil; FWF – fishhook water flea; LAS – lake sturgeon; LAT – lake trout; QUM – quagga mussel; ROG – round goby; RUC – rusty crayfish; RUF – ruffe; SEL – sea lamprey; SWF – spiny water flea; THS – three spine stickleback; WHP – white perch; and ZEM – zebra mussel.

Park Unit	Agency and Contact Information	Survey Timing and Purpose	AIS Capable of Incidental Capture	Monitoring Frequency	Monitoring Locations	Data Housed	QAP (Good, Fair, Poor)	Sampling Method
APIS	USGS Gary Cholwek 715-682-6163 x12 gcholwek@usgs.gov	Spring Forage Fish Assessment	RUF, ROG, THS, WHP	Annual	Among Apostle Islands	Internal USGS Database	Good	Bottom Trawl
APIS	WIDNR Steve Schram 715-779-4035 x12 schras@dnr.state.wi.us	Summer Fish Community Assessment	RUF, WHP, SWF, FWF	Annually, Even Numbered Years in Apostle Islands	Among Apostle Islands, west of APIS to Superior entry, east of APIS to L. Girls Pt., south of APIS to Chequamegon Bay	Internal WIDNR Database	Good	Experimental Gill Net Plankton Net
		Fall Lake Herring Assessment	RUF, WHP	Annual	North Sand Island Lighthouse	Internal WIDNR Database		1.5"-3" Gill Net
PIRO	MIDEQ Bill Taft 517-335-4205	Macroinvert. Assessment	SWF, FWF, ZEM, QUM	Annual		Internal MIDEQ Database	Good	Ponar
PIRO	MIDNR Phil Schneeburger 906-249-1611-311 Kevin Rathbun 906-249-1611-315 Shawn Sitar 906-249-1611-310	Spring, Summer, Fall Lake Trout Assessment	RUF, WHP	Annual	Au Sable Pt., Grand Portal Pt., Perry's Landing – east of Grand Marais	Internal MIDNR Database	Good	2"-3.5" Gill Net

Park Unit	Agency and Contact Information	Survey Timing and Purpose	AIS Capable of Incidental Capture	Monitoring Frequency	Monitoring Locations	Data Housed	QAP (Good, Fair, Poor)	Sampling Method
PIRO	N. Michigan University Jill Leonard 906-227-1619	Spring, Summer, Fall Brook Trout and other Salmonids Assessment	THS, WHP, ROG, RUF, RUC	Monthly (Apr-Dec)	Mosquito R., Seven Mile Cr., Hurricane R.	Internal University Database	Good	Backpack Electrofishing
PIRO	Van Landschoot Fishery 906-387-3851	Commercial fishing	RUF, WHP, SEL attached to fish	Daily – April to October	Lake Superior	None	N/A	Trap Net 2.5" stretch mesh
GRPO	Grand Portage Tribe Seth Moore (Fish) Margaret Watkins (Aquatic inverts) 218-75-2415	Spring, Summer Lake and Brook Trout and Lake Sturgeon Assessment, Plankton	THS, WHP, RUF, ROG, SWF, FWF	Annual	Lake Superior Shoreline	Internal Database	Good	Experimental Gill Net, Boat Electrofishing, Plankton Sampler
GRPO	MNDNR Steve Geving 218-525-0853	Juvenile Lake Trout Assessment	RUF WHP	Annual	Portage Island	Internal MNDNR Database	Good	1.5-2.5" Gill Net in 1/4"increments
INDU	INDNR Brian Breibert 219-874-6824	Yellow Perch and Round Goby Assessment, Snakehead Surveillance	ROG, RUF, WHP, THS, RUC	2x/month Jun/Jul/Aug (sometimes Sept.)	Lake Michigan (vicinity of Park)	Internal INDNR Database	Good	Bottom Trawl, Boat Electrofishing
ISRO	USFWS Henry Quinlan 715-682-6185	Spring Brook Trout Assessment	THS, RUF, WHP	Annual	Select bays and shorelines	Internal USFWS Database	Good	Boat Electrofishing
SLBE	Glen Lake Assoc. Mike Litch 231-334-3612	Vegetation Survey	EUM CLP	Annual	Big and Little Glen lakes	Internal Database	Good	Observation

Park Unit	Agency and Contact Information	Survey Timing and Purpose	AIS Capable of Incidental Capture	Monitoring Frequency	Monitoring Locations	Data Housed	QAP (Good, Fair, Poor)	Sampling Method
SLBE	Leelanau Conservancy Matt Heiman 231-256-9665	Algae, Water Quality	Plankton	5 Years	Big and Little Glen lakes and other lakes in vicinity	Internal Database	Good	Water samples vertical tows
SACN	WIDNR Terry Margenau 715-635-4162	General Fish Assessment	RUF, ROG, THS, WHP, RUC	Annual	St. Croix R. - 6 stations (Solon Springs to St. Croix Falls)	Internal WIDNR Database	Good	Boom Electrofishing
SACN	MNDNR Roger Hugil 320-384-7721	General Fish Assessment	RUF, ROG, THS, WHP, RUC	every 5 years	St. Croix R. (Taylor Falls-Danby)	Internal MNDNR Database	Good	Boom Electrofishing
MISS	MNDNR Dave Zappetillo 651-772-7963	General Fish Assessment	RUF, ROG, THS, WHP, RUC	every 5 years	Mississippi R. and backwaters (Hastings Dam to Ford Dam)	Internal MNDNR Database	Good	Boom Electrofishing, Gill Net, ¾ - 1.5” mesh, Fish traps
VOYA	MNDNR Kevin Peterson 218-286-5220	Standard Lake Survey	RUF, WHP	Annual	Rainy L., Kabetogama Lake, Namakan L., Sand Point L.	Internal MNDNR Database	Good	Gill Net 3/4-2" mesh
		Coregonid Survey	ROG, RUF, WHP	Annual	Rainy Lake, Namakan Reservoir	Internal MNDNR Database	Good	Gill Net 0.5- 2.5" mesh
		Natural Reproduction Assessment	RUF, ROG, THS, WHP, RUC	Annual	Rainy Lake Kabetogama Lake	Internal MNDNR Database	Good	Seine Electrofishing
		Standard Lake Survey	RUF, WHP, EUM, CLP	2006 only	Net Lake, L. Vermilion Lake	Internal MNDNR Database	Good	Gill Net 0.75-2"
		Fish Population Assessment	RUF, ROG, THS, WHP, RUC	2006 only	Ek Lake, Crane Lake	Internal MNDNR Database	Good	Gill Net, Seine, Electrofishing

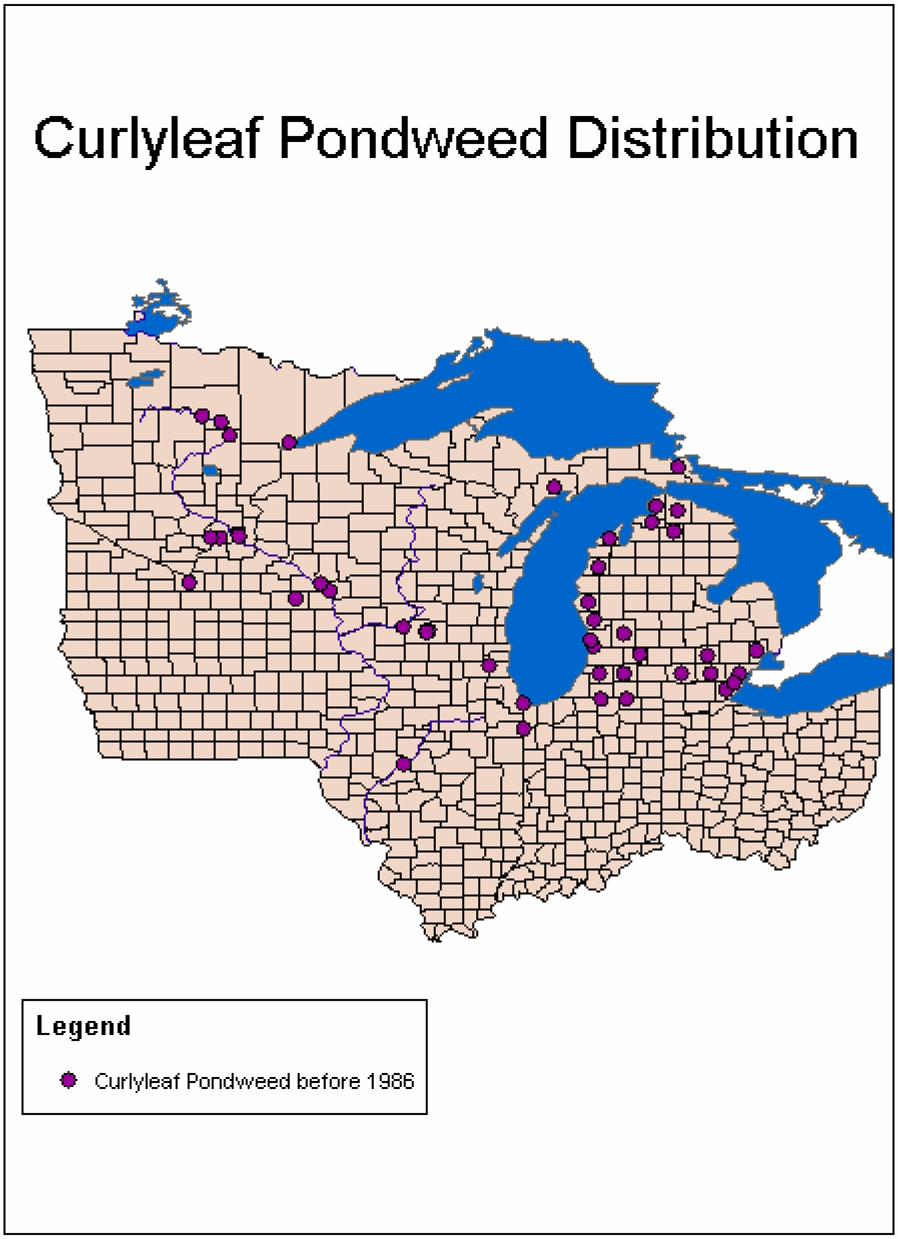
Appendix 4. Data description and time-series distribution maps.

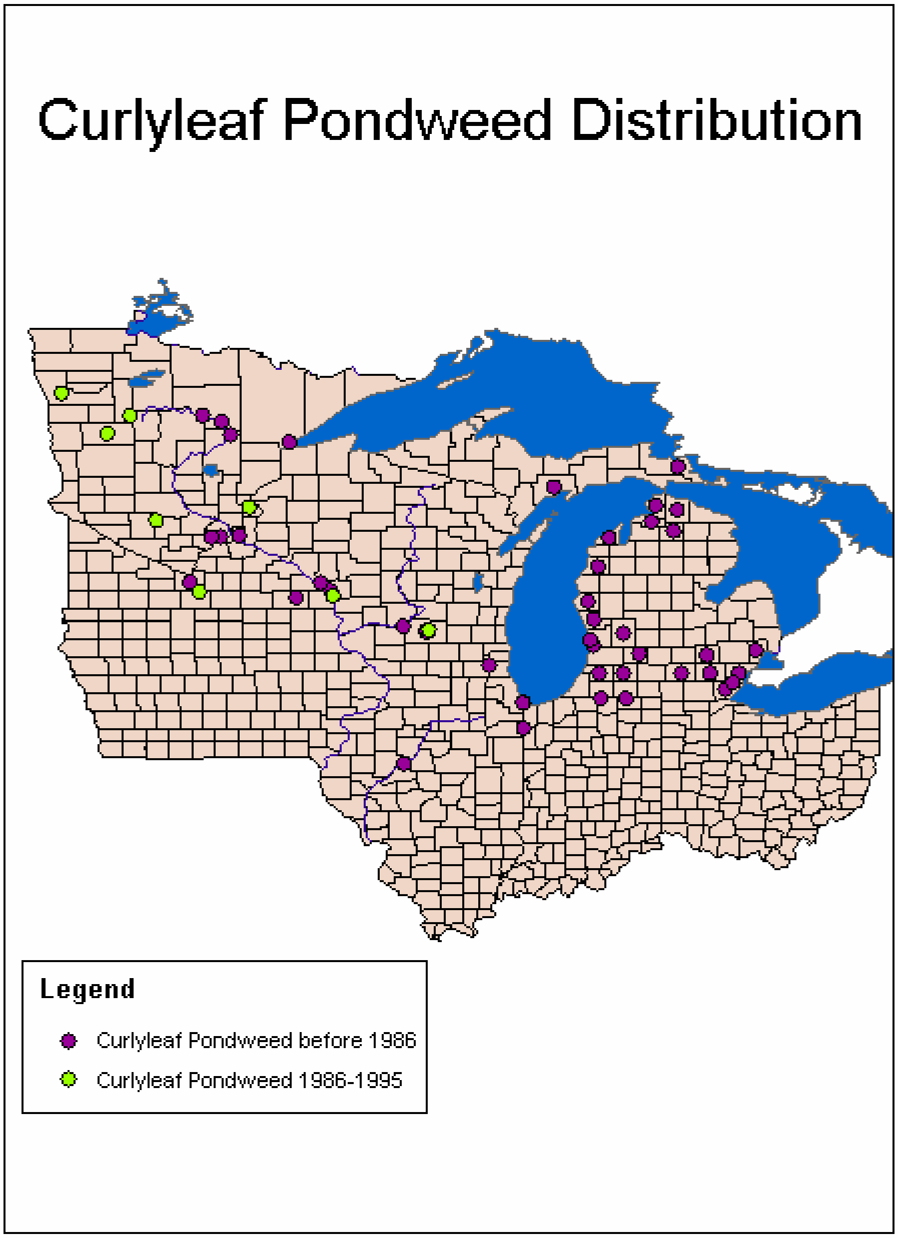
Basemaps
The underlying maps for this project were supplied by the National Park Service Great Lakes Network Office.

Data
The data collected for this effort came from two sources, the U.S. Geological Service (USGS) and the Great Lakes Indian Fish and Wildlife Service (GLIFWC). The U.S. Geological Survey has served as a clearing house for AIS and was able to deliver the shape files for Ruffe, zebra and quagga mussels, and rusty crayfish. GLIFWC uses GIS to monitor AIS and was able to provide files for the other 8 species evaluated. The data for the GLIFWC files were collected internally and incorporated data reported to USGS.

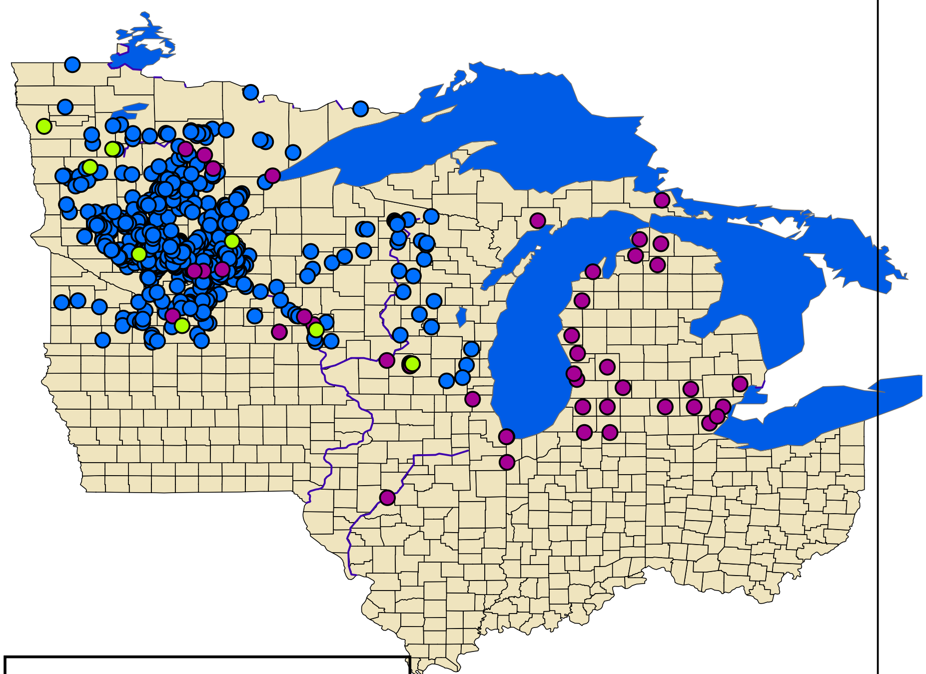
Spatial and temporal coverage was varied for a number of reasons. For example unless a species was of particular concern to someone, information and coverage tended to be fairly localized. Another problem is that data are often collected but not reported or available others. GLIFWC sponsored a conference to address this concern. The ideal situation is a centrally located clearinghouse for AIS information that can make the data available to anybody that is interested.

Time Series Information
While our intent was to provide maps that document the current distribution and rate of range expansion for each of the 12 species addressed in this report, adequate time series data were only available for curly leaf pondweed and ruffe. The maps show a species distribution before 1986, from 1986-1995, and from 1996 to the present.


Curlyleaf Pondweed Distribution
Legend
Curlyleaf Pondweed before 1986

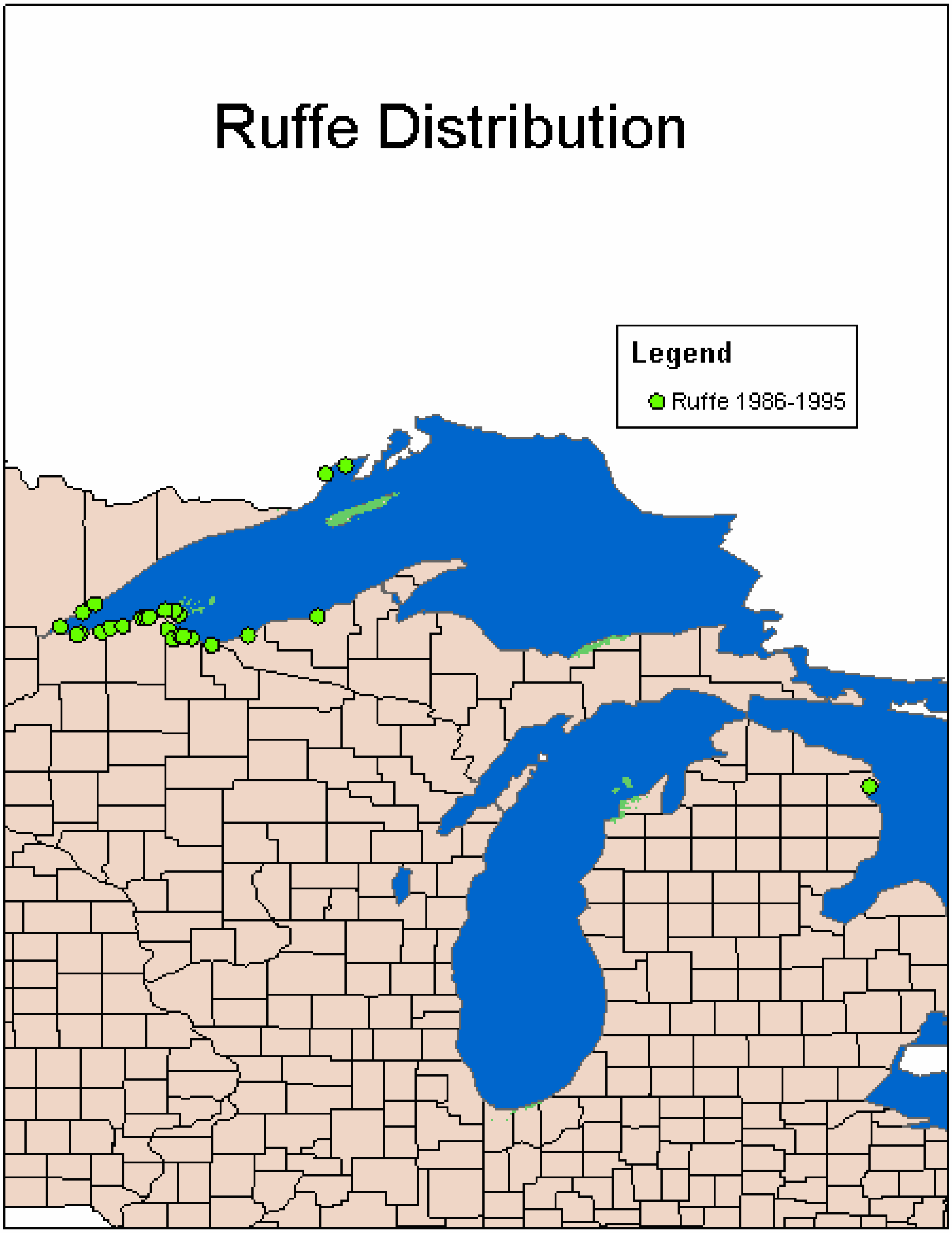

Curlyleaf Pondweed Distribution
Legend
Curlyleaf Pondweed before 1986
Curlyleaf Pondweed 1986-1995

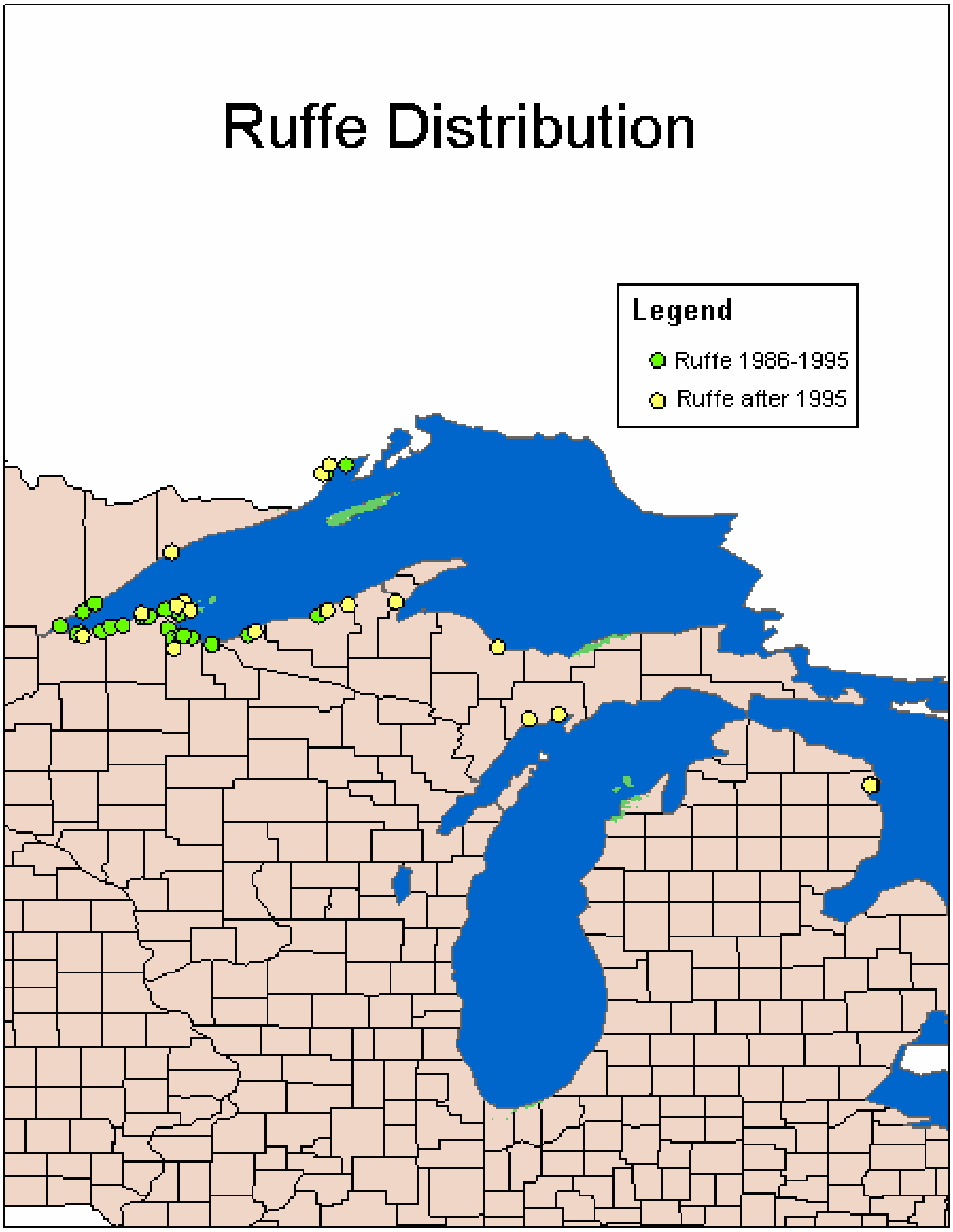
Curlyleaf Pondweed Distribution



Legend

- Curlyleaf Pondweed before 1986
- Curlyleaf Pondweed 1986-1995
- Curlyleaf Pondweed after 1995


Ruffe Distribution
Legend
Ruffe 1986-1995


Ruffe Distribution
Legend
Ruffe 1986-1995
Ruffe after 1995

Appendix 5a. Evaluation criteria and risk ranking for each aquatic invasive species address (maximum score = 50).

Questions:

1	Potential to invade park	1 - 10	1 = Low, 10 = Great; if present, value = 0
2	Significance of impact	1 - 10	Difficult---→Easy
3	Ability to monitor for early detection (ED)	1 - 10	Difficult---→Easy
4	Ability to monitor for early invasion (EI)	1 - 10	Difficult---→Easy
5	Cost of ED monitoring program	1 - 10	Prohibitive---→Manageable
6	Sufficient monitoring underway for ED/EI in park		Y/N
7	Sufficient monitoring underway for ED/EI elsewhere		Y/N

Curly Leaf Pondweed

	1	2	3	4	5	T	6	7
APIS	3	1	8	8	7	27	N	N
GRPO	3	1	8	8	7	27	N	N
ISRO	3	1	8	8	7	27	N	N
PIRO	0	1	8	8	7	24	N	N
SLBE	0	10	8	8	7	33	N	Y
INDU	0	10	8	8	7	33	N	N
SACN	0	10	8	8	7	33	N	Y
MISS	0	10	8	8	7	33	N	Y
VOYA	0	10	8	8	7	33	N	N

Fishhook Waterflea

	1	2	3	4	5	T	6	7
APIS	2	3	2	2	7	16	N	Y
GRPO	2	3	2	2	7	16	N	Y
ISRO	2	3	2	2	7	16	N	N
PIRO	2	3	2	2	7	16	N	N
SLBE	10	3	2	2	7	24	N	N
INDU	0	3	2	2	7	14	N	N
SACN	1	3	2	2	7	15	N	N
MISS	1	3	2	2	7	15	N	N
VOYA	1	3	2	2	7	15	N	N

Eurasian Water milfoil

	1	2	3	4	5	T	6	7
APIS	5	1	8	8	7	29	N	N
GRPO	3	1	8	8	7	27	N	N
ISRO	4	1	8	8	7	28	N	N
PIRO	0	7	8	8	7	30	N	N
SLBE	0	7	8	8	7	30	N	Y
INDU	0	7	8	8	7	30	N	N
SACN	0	7	8	8	7	30	N	Y
MISS	0	7	8	8	7	30	N	Y
VOYA	10	7	8	8	7	40	N	N

Quagga Mussel

	1	2	3	4	5	T	6	7
APIS	2	3	9	8	10	32	N	N
GRPO	2	3	9	8	10	32	N	N
ISRO	2	3	9	8	10	32	N	N
PIRO	2	3	9	8	10	32	N	N
SLBE	0	10	9	8	10	37	N	Y
INDU	0	10	9	8	10	37	N	N
SACN	7	10	9	8	10	44	N	Y
MISS	7	10	9	8	10	44	N	Y
VOYA	6	7	9	8	10	40	N	N

Round Goby

	1	2	3	4	5	T	6	7
APIS	5	5	8	4	9	31	N	N
GRPO	5	5	8	4	9	31	N	N
ISRO	3	5	8	4	9	29	N	N
PIRO	10	5	8	4	9	36	N	N
SLBE	0	5	8	4	9	26	N	Y
INDU	10	5	8	4	9	36	N	Y
SACN	3	5	8	4	9	29	Y	Y
MISS	3	5	8	4	9	29	N	N
VOYA	2	7	8	4	9	30	N	Y

Ruffe

	1	2	3	4	5	T	6	7
APIS	0	7	8	3	9	27	Y	Y
GRPO	8	7	8	3	9	35	Y	Y
ISRO	8	7	8	3	9	35	N	N
PIRO	10	7	8	3	9	37	N	N
SLBE	8	7	8	3	9	35	N	N
INDU	5	7	8	3	9	32	N	Y
SACN	5	7	8	3	9	32	Y	Y
MISS	5	7	8	3	9	32	N	N
VOYA	3	7	8	3	9	30	N	Y

Appendix 5a (continued). Evaluation criteria and risk ranking for each aquatic invasive species address (maximum score = 50).

Rusty Crayfish

	1	2	3	4	5	T	6	7
APIS	5	6	9	9	8	37	N	N
GRPO	10	6	9	9	8	42	N	N
ISRO	1	6	9	9	8	33	N	N
PIRO	5	6	9	9	8	37	N	N
SLBE	10	8	9	9	8	44	N	Y
INDU	10	8	9	9	8	44	N	Y
SACN	10	8	9	9	8	44	N	N
MISS	10	8	9	9	8	44	N	N
VOYA	10	8	9	9	8	44	N	Y

Sea Lamprey

	1	2	3	4	5	T	6	7
APIS	0	10	9	9	8	36	N	Y
GRPO	0	10	9	9	8	36	N	Y
ISRO	0	10	9	9	8	36	N	Y
PIRO	0	10	9	9	8	36	N	Y
SLBE	0	10	9	9	8	36	N	Y
INDU	0	10	9	9	8	36	N	Y
SACN	2	1	9	9	8	29	Y	Y
MISS	2	1	9	9	8	29	N	N
VOYA	2	1	9	9	8	29	N	N

Spiny Waterflea

	1	2	3	4	5	T	6	7
APIS	0	3	2	2	7	14	N	Y
GRPO	10	3	2	2	7	24	N	Y
ISRO	0	3	2	2	7	14	N	N
PIRO	0	3	2	2	7	14	N	N
SLBE	10	3	2	2	7	24	N	N
INDU	0	3	2	2	7	14	N	N
SACN	3	3	2	2	7	17	N	N
MISS	3	3	2	2	7	17	N	N
VOYA	0	3	2	2	7	14	N	N

Threespine Stickleback

	1	2	3	4	5	T	6	7
APIS	0	6	8	1	8	23	Y	Y
GRPO	10	6	8	1	8	33	N	N
ISRO	0	6	8	1	8	23	N	N
PIRO	0	6	8	1	8	23	N	N
SLBE	8	6	8	1	8	31	N	N
INDU	8	6	8	1	8	31	N	Y
SACN	8	6	8	1	8	31	Y	Y
MISS	8	6	8	1	8	31	N	N
VOYA	8	6	8	1	8	31	N	Y

White Perch

	1	2	3	4	5	T	6	7
APIS	8	7	8	3	9	35	Y	Y
GRPO	5	7	8	3	9	32	Y	Y
ISRO	5	7	8	3	9	32	N	N
PIRO	5	7	8	3	9	32	N	N
SLBE	10	7	8	3	9	37	N	N
INDU	10	7	8	3	9	37	N	Y
SACN	10	7	8	3	9	37	Y	Y
MISS	10	7	8	3	9	37	N	N
VOYA	6	7	8	3	9	33	N	Y

Zebra Mussel

	1	2	3	4	5	T	6	7
APIS	2	3	10	9	10	34	N	N
GRPO	2	3	10	9	10	34	N	N
ISRO	2	3	10	9	10	34	N	N
PIRO	2	3	10	9	10	34	N	N
SLBE	0	10	10	9	10	39	N	Y
INDU	0	10	10	9	10	39	N	N
SACN	0	10	10	9	10	39	N	Y
MISS	0	10	10	9	10	39	N	Y
VOYA	7	7	10	9	10	43	N	N

Appendix 5b. Aquatic invasive species risk ranking and recommended species to monitor in each park.

The tables and text below show the scores for the highest 3-6 aquatic invasive species evaluated in each park unit and provide explanation of subjective changes we made to certain rankings. The scores relate to the importance given to monitoring a given species. The higher the score, the greater the monitoring need. In most tables, one or more high ranking species has been crossed out. Species were crossed out despite a high score if we believed that sufficient monitoring was underway to detect a species in or near a park unit or if the species is already known to be present in the park.

In cases where there was an obvious separation in species scores that suggested a substantial difference in the importance of monitoring we subjectively made the determination to limit the number of species recommended for monitoring to at least 3.

Questions 6 and 7 identify species not known to be present in a park where current monitoring in or near the park is believed to be sufficient to detect the species.

Sea lampreys are present throughout the Great Lakes and a well organized control and management program is carried out by the U.S. Fish and Wildlife Service and Department of Fisheries and Oceans, Canada under the direction of the Great Lakes Fishery Commission. Therefore, while sea lamprey are high risk aquatic invasive species we believe that the existing Sea Lamprey Management Program is adequate and do not recommend additional effort be put toward Lake Superior waters, and sufficient monitoring is being conducted by FWS to assess them in and near the park.

<u>Apostle Islands National Lakeshore</u>

Evaluation Question

Species	1	2	3	4	5	Total	6	7
Rusty Crayfish	5	6	9	9	8	37	N	N
~~Sea Lamprey~~	0	0	9	9	8	36	N	Y
~~White Perch~~	8	7	8	3	9	35	N	Y
Zebra Mussel	2	3	10	9	10	34	N	N
Quagga Mussel	2	3	9	8	10	32	N	N
Round Goby	5	5	8	4	9	31	N	N

- White perch are present in vicinity of park, and sufficient monitoring is being done by USGS/ WIDNR to detect them.

- Reproducing populations of zebra mussel and round goby are present in the Duluth/Superior harbor, 70 miles to the west. There is a large amount of recreational boat traffic between the harbor and the Park. Quagga mussels are also present in the harbor.

- Based on threat of invasion, ecological impacts, and ease/low cost of monitoring, we recommend that monitoring be conducted for zebra and quagga mussels, round goby, and rusty crayfish.

Grand Portage National Monument

	Evaluation Questions							
Species	1	2	3	4	5	T	6	7
Rusty Crayfish	10	6	9	9	8	42	Y	Y
~~Sea Lamprey~~	0	10	9	9	8	36	N	Y
Ruffe	8	7	8	3	9	35	N	N
Zebra Mussel	2	3	10	9	10	34	N	N
Quagga Mussel	2	3	9	8	10	32	N	N
White Perch	5	7	8	3	9	32	Y	Y

- Ruffe are difficult to sample in the Pigeon River with a bottom trawl due to abundance of bottom obstructions. Minnesota DNR is conducting some sampling in the vicinity that is capable of capturing ruffe, but it is not sufficient to provide a reasonable chance at capture. A large population of ruffe is established in Thunder Bay Harbour, Ontario, 30 miles to the north along the Lake Superior coastline.

- Based on threat of invasion, ecological impacts, and ease/low cost of monitoring, we recommend that monitoring be conducted for ruffe, white perch, zebra and quagga mussels, and rusty crayfish. Trap nets used to collect ruffe can also capture rusty crayfish, especially when traps are baited. If desired, the GLKN can supplement ruffe and white perch monitoring with boom electrofishing and/or gill netting. The Grand Portage Tribe is considering sampling to detect AIS.

Isle Royale National Park

	Evaluation Questions							
Species	1	2	3	4	5	T	6	7
~~Sea Lamprey~~	6	10	9	9	8	36	N	N
Ruffe	8	7	8	3	9	35	N	N
Zebra Mussel	2	3	10	9	10	34	N	N
Quagga Mussel	2	3	9	8	10	32	N	N
Rusty Crayfish	1	6	9	9	8	33	N	N
White Perch	5	7	8	3	9	32	N	N

- Ruffe and rusty crayfish can be monitored easily and inexpensively with baited trap nets. A large population of ruffe is established in Thunder Bay Harbour, Ontario, 30 miles northeast of the island.

- Zebra and quagga mussels can be monitored easily and inexpensively with cinder blocks.

Voyageurs National Park

Evaluation Questions

Species	1	2	3	4	5	T	6	7
Rusty Crayfish	10	8	9	9	8	44	N	Y
Zebra Mussel	7	7	10	9	10	43	N	N
Quagga Mussel	6	7	9	8	10	40	N	N
Eurasian Water Milfoil	10	7	8	8	7	40	N	N

- We feel that rusty crayfish and zebra and quagga mussels may have more significant ecological impact here than Eurasian water milfoil, and curly leaf pondweed. Zebra mussels are spreading northward in Minnesota as far as Mille Lacs Lake (central Minnesota). In addition, zebra and quagga mussels can be monitored easily and inexpensively. If monitoring resources are limited, we suggest the priority be zebra and quagga mussels and rusty crayfish.

- Rusty crayfish are very abundant in some of the vicinity lakes; rusty crayfish can have a significant ecological impact on native vegetation and crayfish.

Pictured Rocks National Lakeshore

Evaluation Questions

Species	1	2	3	4	5	T	6	7
~~Sea Lamprey~~	10	10	9	9	8	46	Y	Y
Rusty Crayfish	5	6	9	9	8	37	N	N
~~Ruffe~~	8	7	8	3	9	37	Y	Y
Round Goby	10	5	8	4	9	36	N	N
Zebra Mussel	2	3	10	9	10	34	N	N

- Sufficient monitoring for ruffe has been conducted by FWS in vicinity of park. We have also observed that most Lake Superior habitat within the park is not suitable for ruffe.

- Zebra mussels have been detected in a lake near Newberry, Michigan, approximately 50 miles east of the park. We recommend monitoring for rusty crayfish, round goby, and zebra mussel.

Sleeping Bear Dunes

Evaluation Questions

Species	1	2	3	4	5	T	6	7
Rusty Crayfish	10	8	9	9	8	44	N	N
~~Zebra Mussel~~	0	10	10	9	10	39	N	Y
White Perch	10	7	8	3	9	37	N	N
~~Quagga Mussel~~	10	10	9	8	10	37	N	N
~~Sea Lamprey~~	0	10	9	9	8	36	Y	Y
Ruffe	8	7	8	3	9	35	N	N

- Rusty crayfish are known to occur in Lake Michigan near Traverse City, Michigan.

- We suggest that monitoring resources would be most efficiently invested in rusty crayfish, white perch and ruffe.

St. Croix National Scenic Riverway

Evaluation Questions

Species	1	2	3	4	5	T	6	7
Quagga Mussel	7	10	9	8	10	44	N	Y
Rusty Crayfish	10	8	9	9	8	44	N	N
White Perch	10	7	8	3	9	37	Y	Y

- Quagga mussel impacts are similar to zebra mussel. Monitoring the ecological impact of zebra mussel may be most desirable, and it would provide for early detection of quagga mussel.

- We suggest that monitoring resources would be most efficiently invested in rusty crayfish, white perch, and possibly zebra and quagga mussels.

Indiana Dunes

Evaluation Questions

Species	1	2	3	4	5	T	6	7
Rusty Crayfish	10	8	9	9	8	44	N	Y
~~Zebra Mussel~~	0	10	10	9	10	39	Y	Y
White Perch	10	7	8	3	9	37	N	Y
~~Quagga Mussel~~	7	10	9	8	10	37	Y	Y
Round Goby	10	5	8	4	9	36	N	Y
~~Sea Lamprey~~	0	10	9	9	8	36	N	Y

- Zebra and quagga mussels have already been detected in the park.

Mississippi River

Evaluation Questions

Species	1	2	3	4	5	T	6	7
Quagga Mussel	7	10	9	8	10	44	N	Y
Rusty Crayfish	10	8	9	9	8	44	N	N
~~Zebra Mussel~~	0	10	10	9	10	39	Y	Y
White Perch	10	7	8	3	9	37	N	N

- Zebra mussels have already been detected in the park.

Appendix 6. Wisconsin Department of Natural Resources sampling protocol for *Dreissena* (zebra and quagga mussels).

Dreissena Mussel Monitoring Protocol

Zebra & Quagga Mussels

Wisconsin Department of Natural Resources

and

University of Wisconsin Extension

WISCONSIN DEPT. OF NATURAL RESOURCES

April 2006

Acknowledgments

Preparation of this protocol was a joint effort of WIDNR Regional and Central Office staff and UW Extension staff working on aquatic invasive species. This has been a collaborative effort requiring contributions from many individuals. Their assistance was necessary and is greatly appreciated.

Dreissena (Zebra and Quagga) Mussel Monitoring Protocol

Table of Contents

Dreissena (Zebra and Quagga) Mussel Monitoring Protocol

Annually the DNR staff selects over 100 water bodies to sample for zebra and quagga mussel infestations. This protocol is designed to assist DNR staff in collecting samples for adult and veliger (larval form) *Dreissena* mussels. Additionally, this protocol provides guidelines on how the zebra and quagga mussel information is reported and how the information is released for public knowledge. Using this protocol will standardize the collection techniques, improve the quality of collected samples, limit the number of contaminated samples and ensure an accurate zebra mussels infestation database.

The Department updates the zebra mussels listing annually after sampling results are compiled and then issues a press release listing those waters infested with zebra mussels and other invasive species (including quagga mussels). The Department issues a second press release and list prior to the opening of the fishing season reminding boaters to take precautions to avoid spreading zebra mussels and other invasive species. In addition, a list and a map showing the infested waters are included on the DNR web page:
http://dnr.wi.gov/org/water/wm/GLWSP/exotics/zebra.html

Section One
Veliger Monitoring

Sampling equipment

Boat
Anchor
50-cm diameter, 64-micron mesh plankton net
Rope on net with the meter increments marked
Vinegar
Large container to hold plankton net for vinegar bath
250 ml plastic bottles
1-liter plastic bottles
Alcohol, 95% alcohol (190 proof ethyl alcohol)
Lake Maps
Labels for Bottles (contact Ron Martin for labels)
Sharpie
Zebra mussel data collection sheet (see appendix 1)
Cooler with ice
Change for car wash (you may want to make a map of the car wash stations in your area)
GPS unit - *optional*

Ordering Information

Nets

If you need a sampling net, please contact Ron Martin. He has catalogs and information on how to order nets or sampling nets can be ordered from:

Research Nets Incorporated
14207 100th Ave. NE
Bothell, WA
(425) 821-7345

The standard net specifications are:

0.5-meter mouth (for 2-meter tows) or 0.3-meter mouth (for 5-meter tows)
5 to 1 length to diameter ratio
64-micron mesh size
0.5 meter towing ring with a single point bridle
3.5" PVC 2-piece collecting bucket

Ethyl Alcohol

Ethyl alcohol is available from the University of Wisconsin Madison at Materials Distribution Services (MDS). You can place your order by phone or via their web page. You will have to pick up the alcohol in person so please place your order several days in advance of your planned pick up date. MDS will not ship the alcohol to you by common carrier (UPS, US Postal, etc.) because 190 proof ethyl alcohol is a hazardous material. You must also purchase it by the case (4 gallons) because they will not break up a case.

One case (4 gallons) of 95% alcohol (190 proof ethyl) costs $ 38.66, (2006 costs). For ease in ordering, an account number (MD13912) has been set up with MDS by Ron Martin. The catalog number is 2293. Please specify both account and catalog numbers when placing an order.

The address for MDS is:
Material Distribution Services
2102 Wright Street
Madison, WI 53704
Phone # 608-245-2900
http://www.bussvc.wisc.edu/mds/mds.html

For directions on how to get to MDS to pick up your alcohol, go to:
http://www.bussvc.wisc.edu/mds/location.html

Pre-sample Preparation

Contact Lab

Prior to sending samples, notify the DNR Southeast Region analytical lab, (Steve Galarneau, via e-mail at Stephen.Galarneau@dnr.state.wi.us or 920-892-8756 ext. 3051) with specifics on the number of samples, collector, sample sites and dates. Provide your contact information as well so that the lab can get in contact with you. The analytical lab will e-mail back to you a receipt of the samples and the results when they are completed.

Sampling

Sample Frequency

Three samples should be collected from a particular lake on three dates between June and September (for a total of nine samples per lake). Ideally, samples should be collected at monthly intervals after the water temperatures reach 54 degrees. The first collection dates will vary from early to late June. If you choose not to sample the same lakes on each of the three sample periods, please contact the analytical lab so that they can make the appropriate changes to the database.

Sample Location

On each sampling date, veliger samples should be collected from three different locations in a lake. The sites should be in different bays or basins or at several of the more heavily used lake sites. The three sampling sites should be fairly close to hard substrate (i.e. habitat such as rocks or piers) but deep enough to sample, so perhaps in 15 to 20 feet (4 to 6 meters) of water is a good rule of thumb. Avoid collecting the veliger sample from an open-water deep mid-lake site. Additional samples can be taken in bigger bodies of water where there may be multiple fingers, bays, or multiple boat launches. Mark on the lake map where samples were collected. These same sites should be used for each of the sample periods – if not, then submit a revised map with subsequent samples.

Sample Collection

The volume of water that you sample can generally be determined by the trophic status of the lake. A highly eutrophic lake will quickly fill the net-bucket with plankton and provide a dense enough sample to examine for veligers. However an oligotrophic lake requires sampling a greater volume of water to collect a sufficient sample. Using the standard plankton net (50-cm diameter*, 64 micron mesh) the volume of water to collect for the different trophic conditions are:

- Oligotrophic lakes - collect two 2-meter tows from each site. Consolidate to 1 sample for each site. You will have sampled 4-meters of water per site.
- Mesotrophic lakes - collect one 2-meter tow from each site. You will have sampled 2-meters of water per site.
- Eutrophic lakes - collect one 1-meter tow from each site. Samples from eutrophic lakes are more difficult to analyze, so reducing the sample volume will facilitate the process. You will have sampled 1-meter of water per site.

*note: if using the 30-cm diameter net, you should sample five 2-meter tows for oligotrophic lakes, three 2-meter tows for mesotrophic lakes and three 1-meter tows for eutrophic lakes to obtain relatively the same volume of water as above.

Lower the net into the water at the first of the three pre-selected sites. Pull the net up vertically. Care should be taken to pull the net up slowly enough so that no pressure wave is created on the surface of the water. If you are creating a pressure wave, you are under-sampling the water column. Be sure to rinse the net from the outside of the net so that all of the material washes into the plankton collection cup. Record sampling information on the zebra mussel data collection form (see appendix 1).

- Care must be given that the net does not hit the lake bottom. When this happens, the sample is of muddy water, which is very difficult or impossible to analyze. If you hit the lake bottom, rinse out the sampling equipment and try shorter tows (e.g. two 1-meter tows instead of the protocol of one 2-meter tow), or go to a different area of the lake that will provide enough depth for a good tow.
- For shallow lakes where it is impracticable to do a vertical tow, collect a horizontal sample at mid-depth. In shallow lakes, you may split the sampling depth (i.e., two one-meter tows with the 50-cm net or five one-meter tows with the 30-cm net)
- Condense and decant your plankton sample into your bottle after each tow to obtain an accurate enumeration of the larval density in your lake. For example, if you had two 2-meter tows, you would wash down the net from the outside and condense the sample for each of the 2-meter tows. Both samples should be placed into the 250-ml plastic bottle or 1-liter bottle. (Use the smallest bottle size that is practical, but you may need to use a 1-liter bottle due to the sample size).
  Note: If samples are to be shipped by common carrier, size restrictions may apply to the sample containers. The maximum size allowed under the US DOT regulations for plastic containers is 1 liter – check with the shipper for any additional restrictions prior to sampling so that samples are collected in appropriately sized bottles.
- Condense the size of the sample by filtering out as much water as possible in the field. This helps reduce the amount of alcohol that needs to be added and aids in the analyses as well.
- Preserve the sample using 95% alcohol. The ratio should be 4 parts alcohol to 1 part sample. Note: If the prescribed alcohol to sample ratio (4:1) can not be achieved after repeated condensing and decanting, then the sample should be split between two sample bottles. Label each with the same information (as specified under “Processing the Sample – Field”), and label one as “Split 1 of 2” and the other as “Split 2 of 2”.
- Repeat the process at the other two pre-selected sites. You will have 3 samples for each lake
  - Alternatively, you can composite the samples from the three sites into one 250-ml or larger (1-liter) bottle and receive a single enumeration for the lake. Obviously, with the composite sampling, you lose specific sample location information, but either approach is acceptable.
- Transport the sample bottle(s) on ice in a cooler.

Processing the Sample - Field

Attach a State Lab of Hygiene, or comparable, label to each sample bottle and include the following information. Be sure to write legibly and with indelible ink (e.g. Sharpie) – do not use a ball point pen, as the ink is soluble in alcohol. Additional labels may be obtained from Ron Martin.

<u>Label sample bottles with the following information:</u>

- Collector's name
- Collector's phone number - important, as the analytical lab may need to contact you regarding the sample
- Lake name
- WBIC
- County
- TRS
- *optional* - Sample site Latitude/Longitude locational data using a GPS unit
- Site number
- Net opening diameter (0.5m or 0.3m)
- Sample date
- Number of tows
- Depth of the tows
- Preservatives added

Shipping/Deliver Samples

Please deliver the veliger samples to Steve Galarneau in SER at:
Steve Galarneau
Plymouth Service Center
1155 Pilgrim Road
Plymouth, WI 53073
Phone # 920-892-8756 ext. 3051

Shipping Samples Containing Ethanol

Veliger samples, preserved with ethanol (4 parts ethanol: 1 part sample), are hazardous materials because of their flammability (the flash point of a 4:1 ethanol/water solution is approximately 72º F). Ethanol solutions are classified as flammable liquids by the US Department of Transportation and the shipment of such materials is governed by US DOT's regulations - with a couple of exceptions, as listed below.

Transport in State Vehicles

Hazardous materials, including ethanol solutions, can be transported in State of Wisconsin vehicles, without the need to comply with any US DOT regulations. Thus, it is permissible to ship these samples by state vehicle. The hazardous nature of the samples is not diminished simply because no shipping requirements pertain to this mode of transport, and some common sense precautions should be taken when using this means of shipping:

- Samples should be transported on ice to keep them below their flash point temperature.
- Samples should not be kept where hazardous materials would otherwise not be permitted, e.g., in an office.
- Samples should not be transported in the passenger compartment of a vehicle.
- All employees involved in transporting ethanol-containing samples in a state vehicle should be made aware of the hazard that these samples pose and the precautions that should be taken to minimize those hazards.

US Postal Service

The US Postal Service has its own set of requirements for shipping hazardous materials, and therefore it is not subject to the US DOT hazardous materials regulations. Guidance for shipping ethanol solutions via the USPS is available on the Intranet at: http://intranet.dnr.state.wi.us/int/es/science/ls/fpm/EtOH_usps.pdf and also on the USPS website at: http://www.usps.com/cpim/ftp/pubs/pub52.pdf. The USPS regulations provide a limit of one container (not to exceed 1 pint for a non-metal container) per mail-piece. Given this limitation, the USPS will generally not be a practical alternative for shipping these samples.

<u>Common Carriers</u>

The US DOT regulations (49 CFR, Parts 171 – 180) apply when shipping samples via common carrier. Many of the familiar shipping companies provide hazardous material shipping service. The US DOT regulations begin with training requirements for those who offer hazardous materials for shipping via a common carrier. Therefore, any staff member who prepares ethanol-containing samples for shipment via a common carrier must be trained in accordance with the requirements of the US DOT regulations. Additional requirements apply to packaging, packing, marking, labeling and documentation. The regulations are available at: http://www.myregs.com/dotrspa/. Some shipping companies have additional requirements, or more restrictive requirements than the US DOT regulations. Table 1 summarizes some of the basic information about shipping hazardous materials with some of the common shipping companies.

Table 1 – Shipping Hazardous Materials via Common Carrier

Shipping Company	Ship Hazardous Materials?	Website	Service Restrictions
DHL	No		
Dunham Express	Yes	http://www.dunhamexpress.com/index.html	
Fed Ex	Yes	http://www.fedex.com/us/services/options/ground/hazmat/	Must be qualified before shipping. Packaging must be approved. Maximum volume per package = 16 L. Not accepted at all offices. Pick up service may not be available.
Spee-Dee Delivery	Yes	http://www.speedeedelivery.com/faqs.html	
UPS	Yes	http://www.ups.com/content/us/en/resources/prepare/hazardous/index.html	Service only on contract basis. Must use UPS compliant software. Packaging must meet specifications. Not accepted at all offices.

Decontamination Procedures

When multiple lakes are sampled on the same day, the net, boat and all other sampling equipment must be decontaminated between lakes**.** Decontaminating will eliminate cross contamination and reduce the risk of transporting veligers from lake to lake. You do not have to decontaminate equipment between sample sites on the same lake.

- The net and sample equipment can be decontaminated using regular household vinegar. The acidity of the vinegar will kill the veligers. An easy method of vinegar decontamination is to use a large, round rubber storage container that will fit the outside diameter of the net. Put in enough vinegar to cover the net. Keep the storage container in the truck rather than in the boat. Every time you take your boat out of a lake, place the net in the vinegar. Dipping equipment into 100% vinegar for 5 minutes will kill veligers. Take the net out at the next lake and let it rinse in the water a minute or so before taking your first sample. Rinse the net without dipping the ring below the surface, so that the vinegar is rinsed from the outside of the net. There is no need to change vinegar between lakes, just add more vinegar when the level gets low. Be aware that vinegar attracts wasps, bees and hornets.
- You should wash the boat between lakes following the DNR boat cleaning procedures. Refer to boat cleaning procedures in the watercraft inspection handbook.
- Another approach that has been quite effective in some areas is to benefit from citizens that offer to take our technical staff onto the lake with their boats. This saves time because we don't have to launch and then decontaminate the trailer and boat upon departure.
- If multiple lakes are sampled in one day, it is recommended to sample any lakes that are not on the watch or infestation lists before sampling lakes on those lists, to minimize the potential for transport.

Section 2
Adult Zebra and Quagga Mussel Monitoring

The adult monitoring serves several purposes: (1) to verify a reproducing population if veligers have been identified as being present in a water sample, (2) to determine the population densities of mussels after an infestation has occurred, (3) to track the spread by collecting additional data on lakes where veliger monitoring is not being conducted, and (4) to monitor for the quagga mussel.

Adult *Dreissena* (zebra and quagga) mussel monitoring for inland waters is accomplished using one of two methods:

1. Shoreline surveys and regular inspections of structures in the water to determine the presence/absence of zebra and/or quagga mussels.
2. Substrate sampler monitoring (substrate refers to any substance in the water that zebra or quagga mussels may attach to) to estimate population densities.

<u>Sampling Equipment</u>**:**
Substrate samplers
Rope
Buoy float
Anchor (e.g. concrete block)
Rubbing alcohol
Zebra mussel data sheets, Method A and Method B (see appendices 2 & 3)
- quagga mussel reporting is discussed below and requires lab verification

Hand lens 30X

Method A: Shoreline Surveys

Shoreline surveys and inspections of structures in the water are conducted to identify the presence or absence of adult *Dreissena* mussels. A single observer can monitor thousands of square meters of substrate at a given location in a short period of time – covering a larger surface area than a set of substrate samplers. Monitoring for the presence or absence of adult zebra or quagga mussels will address two important objectives: 1) document the presence of adult *Dreissena* mussels when veligers have been found during the veliger monitoring, and 2) document multiple year classes of zebra or quagga mussels. These data provide important information regarding the viability of the population of zebra or quagga mussels in that waterbody.

<u>Collecting Samples:</u>

- Conduct shoreline surveys about once every two weeks from ice out to ice on.
- Target areas around public boat ramps or areas that are likely to have a lot of boating traffic in the vicinity (for example, fishing hot spots, resorts, campgrounds, etc.).
- Any solid surface is a suitable substrate to observe. Rub your hands along some of the submerged surfaces. Zebra mussels on the surface will feel like sandpaper.

- Zebra mussels are often found in cracks and crevices of rocks and structures. Small zebra mussels can be attached to plants as well (Figure 1).

Figure 1. Zebra mussels attached to native water-milfoil.


NOTE: It is especially important to pay attention to structures removed from the water in the fall of the year or before winter ice forms (for example, docks, piers, boats, buoys, etc.).

<u>Reporting Zebra Mussel Monitoring Results</u>:

Collect any mussels that you believe are zebra mussels, place them in rubbing alcohol and send it to the zebra mussel regional coordinator for confirmation. See zebra mussel contact list in watercraft inspector handbook. If you find what you believe are zebra mussels and note various shapes of mussels of similar size, collect those as well for evaluation of whether or not quagga mussels may also be present.

For tracking the movement of zebra or quagga mussel infestations, a negative report is as important as finding *Dreissena* mussels at a location. All monitoring efforts should be reported on the zebra mussel datasheets and submitted to Ron Martin at 101 South Webster St., Madison, WI 53707. Complete the zebra mussels reporting form Method A, electronically available as form 3200-122 A at http://intranet.dnr.state.wi.us/itworks/forms/eforms.asp or a paper copy in Appendix 2 below.

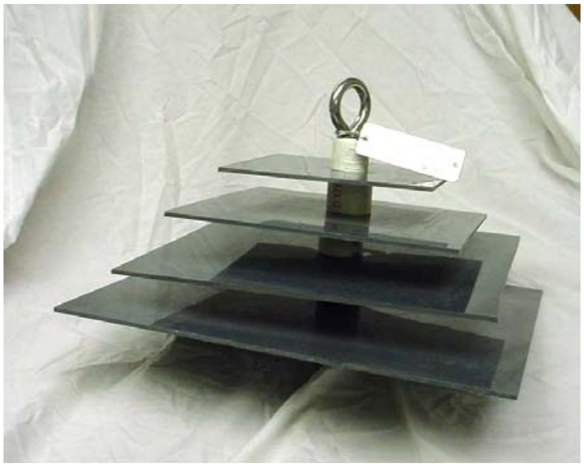
Method B: Substrate Monitoring

Substrate samplers can be used to determine if zebra mussels or quagga mussels are present. When placed in waterbodies without known populations of *Dreissena* mussels, substrate sampler monitoring documents the arrival of zebra or quagga mussels and tracks the spread of these mussels. Substrate samplers can also be useful on lakes with known zebra mussel populations for determining zebra mussel population growth and seasonal abundance, and also for monitoring for the presence of zebra mussels and quagga mussel.

<u>Substrate Sampler Materials</u>

The sampler is a series of four square-plates that are 6, 8, 10 and 12 inches in size, pyramiding from smaller plates at the top down to larger plates at the bottom (Figure 2). The plates are made of 1/8 inch grey plastic PVC stock with ¾-inch CPVC grey pipe for spacers (1-inch sections) between the plates. The sampler is held together with an 8 inch long 3/8 inch diameter stainless steel eyebolt, plus washers and a wing nut. Each sampler has a DNR tag attached that provides a phone number for further information. Samplers are ordered from Cathy Cleland (Rhinelander, 715-365-8997). Directions to build a substrate sampler are found in Appendix 4. Note - the substrate samplers are easily disassembled and cleaned for the next sampling season.

Figure 2. Substrate sampler for zebra mussel monitoring.



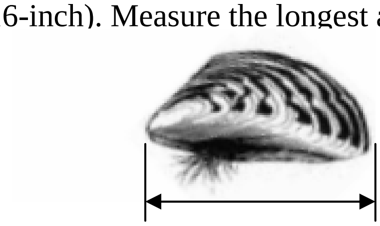
<u>Placement of Substrate Samplers</u>

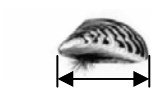
- Place the substrate sampler in an area where there will be little chance of vandalism.
- Hang the substrate sampler from a dock, pier or other structure found in the water. (A float or buoy may be used to suspend the sampler in the water column. If a float is used, a waterway marker application and permit form is necessary before the substrate sampler is placed in the water).
- Put two samplers at each location chosen for monitoring. The top sampler is removed and analyzed every four weeks, then placed back into the lake for the next sampling period. The second (bottom) sampler remains in the water for the entire monitoring season. Securing the two samplers on the same line with clips makes it easy to replace the top one every four weeks.
- A small concrete block anchor works to hold the sampler(s) in place (and provides an additional substrate sampler to examine). Rope can be used to suspend the sampler, but sometimes wildlife will sever the rope. Chains work well to better secure the samplers in those locations.
- Suspend substrate samplers in water, preferably where the water is at least 6 feet deep, but shallower is acceptable. Samplers should be placed at a depth of 6 feet or at mid-depth, whichever is less.

- Place samplers in areas where zebra mussels are most likely to be found. Pay special attention to areas in which zebra mussels may have been transported from infested waterways (for example public boat ramps, water access sites, fishing hotspots, resorts, campgrounds or areas where diving ducks tend to reside).
- Avoid placing substrate samplers in areas where there is strong current.

<u>Analysis of Samples for Quantitative Monitoring of Adults</u>

1. Put the samplers in small white or clear garbage bags when they are removed from the water.
2. In the lab, disassemble the sampler and examine each plate with a hand lens. Scan the entire plate looking for zebra mussels.
3. Recently settled post-veligers can be very small. If you were to rub your hands along the plate, the surface will feel like sandpaper. If you believe that you have detected post-veligers, please mail them to the Watercraft Inspector to have it examined by the Region Biologist or send it to Steve Galarneau following the shipping protocol cited in the veliger monitoring section for verification.
4. Count the number of zebra mussels found on the top and bottom of each plate and record these numbers separately. (use zebra mussel Method B forms, see appendix 3).
5. Report the lengths of the smallest and largest mussels on the plate to the nearest millimeter (1/16-inch). Measure the longest axis of the shell. See diagrams below.





6. For an initial discovery, all zebra mussels collected should be placed in rubbing alcohol for expert verification.
7. For lakes that have zebra mussels and the monitoring is to detect if the quagga mussels are also present:
   - sort the mussels by relative size and shape,
   - compare the shapes
     1. Zebra mussels (*Dreissena polymorpha*) have a flat attachment edge, usually a dark striped shell and are almost as wide as it is tall.
     2. Quagga mussels (*Dreissena bugensis*) have sharp edges on both sides and are fan-shaped. The quagga mussel shell is wider and narrower than the zebra mussel. Coloration may range from nearly all white to coloration similar to the zebra mussel.
   - If you believe that you have two differently shaped mussels of the same relative size, place those mussels in alcohol and send them to the Region biologist or Steve Galarneau for verification.

8. If there are a large number of mussels attached to the plate (Figure 3), a subset of the plate can be evaluated and reported. The sub-sampling method used must be reported on the lab reporting form.
9. Note: Sampler plates can be thoroughly scrubbed, dried, reassembled and reused next year.

Figure 3. Substrate sampler from Metonga Lake with attached zebra mussels.

Reporting

Complete the zebra mussels reporting form Method B, electronically available as form 3200-122 B at http://intranet.dnr.state.wi.us/itworks/forms/eforms.asp or a paper copy in Appendix 3 below. Send the completed form to Ron Martin, 101 South Webster Street, Madison, WI, 53707.

Samples should be clearly labeled with all requested information. Both field staff and volunteers that monitor for adults use the same data sheets. For tracking the movement of zebra mussel infestations, a negative report is as important as finding zebra mussels at a location. All monitoring efforts should be reported on the zebra mussel reporting form and submitted to Ron Martin.

Field staff should also provide Ron Martin with a lake map showing the location of the monitoring sites. The zebra mussel monitoring sites, along with the names and addresses of the monitors, are maintained and updated periodically. Maps showing all the sampling locations (for adults and veligers) are recorded on the GIS network and are available on the DNR web page: http://dnr.wi.gov/org/water/wm/GLWSP/exotics/zebra.html.

Section 3
Zebra Mussel Listing Guidance

The Wisconsin Department of Natural Resources is monitoring some of our inland waters for the presence of zebra mussel veligers using plankton tows and for the presence of post-veligers and adult zebra mussels using substrate samplers. Occasionally sample results from a lake will detect low numbers of zebra mussel veligers in a sample or very few adult mussels of the same size. In both incidences it is clear that a zebra mussel introduction has occurred, but these data do not substantiate that a reproducing population exists in that waterbody. Quagga mussels are present in Lake Michigan coastal areas but have not been found in Wisconsin's inland lakes at this time.

This guidance document provides listing recommendation criteria for when to place a water body onto a Watch List versus an Infested List (a determination has to made whether there is an established reproducing population). It also presents criteria for delisting a lake in the event that zebra mussels are no longer detected. A review team consisting of the Exotic Species Statewide Coordinator, Ron Martin, and region representatives make the final determination for listing or de-listing a waterbody.

<u>Infested List</u>
DNR lists a waterbody as infested for zebra mussels when we have data indicating that there is an established reproducing population. Generally speaking, that would mean that we detect evidence of in-lake reproduction.

<u>Watch List</u>
When veligers, post-veligers or adults are detected in a lake sample, the regional biologist and statewide coordinator (Ron Martin) are contacted. The biologist is requested to conduct a lake survey at their earliest opportunity. In general, the survey would include examining shores, piers and other available substrate near where the plankton tow with the veliger was collected. Additional plankton tows and scuba diving or underwater video surveys may be warranted upon the discretion of the biologist. If the veligers, post-veligers and/or adult zebra mussels found are all from the same year class, then the waterbody is placed on the "Watch" list to be targeted for additional follow-up work by DNR staff. Waterbodies on the Watch list are monitored for zebra mussels and usually include increased public information and education efforts.

Table 2 provides the zebra mussel listing recommendation criteria. A water body is recommended to be placed on the "Watch" or "Infested" list based on meeting one or more of the criteria. Ultimately, the Regional resource managers, in consultation with the aquatic invasive species program coordinator, determine the appropriate listing for a waterbody.

Table 2. Zebra mussel listing recommendation criteria.

Watch List	Infested List
Adults all of the same size, or post-veligers (from substrate analysis) or veligers all from the same year.	Adults zebra mussels of different sizes.
Low numbers of veligers and a substrate evaluation was negative.	Adult zebra mussels found in more than one location of the waterbody. Veligers need not be found.
	Veligers or post-veligers (from substrate analysis) from consecutive years.
	Veligers and adult zebra mussels present.

Footnote: Although the listing criteria reflect monitoring for both veligers (larvae) and adults, it should be noted that only lakes are monitored for zebra mussel veligers, not streams or rivers.

Delisting Criteria

Unfortunately, once zebra mussels become established in a waterbody it is unlikely that they will be eradicated. Nonetheless, in the event that a waterbody is listed as infested for zebra mussels, but subsequent information indicate that they are no longer present, we can use the following criteria to delist a waterbody as infested.

All of the following need to occur for **at least two years** to delist a waterbody:

- Based on additional monitoring, no veligers are observed in any of the samples collected from May through September following the standard monitoring protocol.
- Substrate samplers are deployed at three or more locations in the lake from May through September, concurrent with the veliger collections above, and all are negative for zebra mussels.
- A lake survey is conducted and survey results of suitable habitat show no adult zebra mussels are present. (Obviously, the survey(s) should include the location where zebra mussels had been detected in the past).
- *Optional* - a scuba and/or underwater camera survey is conducted in the area(s) where zebra mussels had been detected in the past.

* When these conditions are met the waterbody may be delisted if regional resource managers recommend delisting and the statewide coordinator concurs.

Delisted waterbodies would be moved to the Watch list and continue to be monitored for zebra mussels. Waterbodies on the Watch List remain on that list unless they are moved to the Infested List or if subsequent long-term sampling results indicate that no veligers or adults are present. Hence, no delisting criteria are recommended for Watch List waterbodies.

Section 4
Notification/Releasing Zebra Mussel Information

Standard Notification Sequence

Following the zebra mussel listing criteria cited above, the regional biologist and the statewide coordinator have concurred that the waterbody should be listed. The regional biologist informs the Public Information Officer (PIO), wardens, fishery and water resources staff, volunteer monitors, and management at the regional level about the sighting. The regional staff, in consultation with the statewide coordinator, determines if a press release is needed. Prior to issuing a press release, the regional biologist notifies the local entities affected by the sighting (lake association or district and industries or water utilities). Attached, as an addendum to this report, is a sample press release that can be used as a template. After the press release is issued, the regional biologist coordinates any follow-up actions that are necessary including posting signs, additional monitoring, or information and education (I&E)/outreach efforts.

Appendix

Appendix 1. Zebra Mussel Data Collection Form
Appendix 2. Method A Data Sheet
Appendix 3. Method B Data Sheet
Appendix 4. Substrate Sampler Construction Directions
Appendix 5. Press Release Templates

Zebra Mussel Data Collection Form

vers. 3/11/2005

COLLECTOR INFORMATION

Name: ________________________________

Phone number: ___________________________

e-mail: __________________________________

Region: _________________________________

LOCATION

Waterbody Name: ___________________________ WBIC: _________________________

County ___________________________________

Township: _______ Range: _______ Section: ______ 1/16 Section: ______ 1/4 Section ______

Optional
Latitude: _________________ Longitude: __________________ Method: ___________
Datum: ____________

Date (MM/DD/YYYY): _____________ Time: ______________

COLLECTION INFORMATION

Site Location: ____ Site #: ______________
Site depth (m): _____________ Secchi (m): ___________

Site Location: ____ Site #: ______________
Site depth (m): _____________ Secchi (m): ___________

Site Location: ____ Site #: ______________
Site depth (m): _____________ Secchi (m): ___________

Net diameter (circle one): 0.5 m or 0.3 m

Number of net tows: ___________ Depth of tows (m): _____________

Number of zebra mussel samples sent to lab: ________________

Preservatives added: ______________

COMMENTS / OBSERVATIONS:

State of Wisconsin
Department of Natural Resources
PO Box 7921, Madison WI 53707-7921
dnr.wi.gov

Zebra Mussel Monitoring Report
Method A – Presence / Absence

Form 3200-122A (3/05) Page 1 of 2

Notice: Information on this voluntary form is collected under ss. 33.02 and 281.11, Wis. Stats. Personally identifiable information, including such data as volunteer name, address, phone number, will be used for management of DNR programs. Wisconsin's Open Records laws, s. 19.32-19.39, Wis. Stats., require the Department to provide this information upon request.

Instructions: Fill out form and mail to the WI DNR Zebra Mussel contact person for your area.

Collector Data

Last Name	First	MI	Are you a Volunteer? ☐ Yes ☐ No

Address

City	State	ZIP Code

Telephone Number	Email Address (optional)

Monitoring Location

Date	Time	County

Lake Address, further explanation town/range/section and latitude and longitude optional

Body of Water	Waterbody ID Number (if known)

Nearest Landmark (boat ramp, bridge, pier, etc.)

Distance and Direction from Landmark

Water Body Type:
☐ River / Stream ☐ Natural Lake / Pond ☐ Marsh / Swamp
☐ Canal / Ditch ☐ Man-made Reservoir ☐ Estuary / Bay

Water Depth where ZM Found

Describe exactly how the zebra mussels were found (what they were attached to or other comments)

Monitoring Results

Total Number of Zebra Mussels Found:	**Note:** If more than 20 zebra mussels are found, measure 20 mussels chosen randomly from the sample. If less than 20 mussels are found, measure all mussels. Report results in the table on page 2 of this form.
Size of Largest Zebra Mussel Found:	
Size of Smallest Zebra Mussel Found:	

Zebra Mussel Monitoring Report
Method A – Presence / Absence
Form 3200-122A (3/05) Page 2 of 2

Length of Zebra Mussels from Sample

If more than 20 zebra mussels are found, measure 20 mussels chosen randomly from the sample. If less than 20 mussels are found, measure all mussels.

Number	Length (mm)
1	
2	
3	
4	
5	
6	
7	
8	
9	
10	
11	
12	
13	
14	
15	
16	
17	
18	
19	
20	

Note: All initial discoveries should be placed in rubbing alcohol until verification by an expert is obtained.

State of Wisconsin
Department of Natural Resources
PO Box 7921, Madison WI 53707-7921
dnr.wi.gov

Zebra Mussel Monitoring Report
Method B – Quantitative

Form 3200-122B (3/05) Page 1 of 2

Notice: Information on this voluntary form is collected under ss. 33.02 and 281.11, Wis. Stats. Personally identifiable information, including such data as volunteer name, address, phone number, will be used for management of DNR programs. Wisconsin's Open Records laws, s. 19.32-19.39, Wis. Stats., require the Department to provide this information upon request.

Instructions: Fill out form and mail to the WI DNR Zebra Mussel contact person for your area.

Collector Data

Last Name	First	MI	Are you a Volunteer? ☐ Yes ☐ No

Address

City	State	ZIP Code

Telephone Number	Email Address (optional)

Monitoring Location

Date	Time	County

Lake Address, further explanation town/range/section and latitude and longitude optional

Body of Water	Waterbody ID Number (if known)

Nearest Landmark (boat ramp, bridge, pier, etc.)

Distance and Direction from Landmark

Water Body Type: ☐ River / Stream ☐ Canal / Ditch ☐ Natural Lake / Pond ☐ Man-made Reservoir ☐ Marsh / Swamp ☐ Estuary / Bay

Water Quality Parameters

Water Temperature	Secchi Depth	Water Depth at Site

Sampler Information

Substrate Condition: ☐ Plants ☐ Sand ☐ Rocky ☐ Soft ☐ Sediment ☐ Other: ____________

Sampler Depth	Date Sampler Deployed	Date Sampler Recovered

Other Observations or Comments

Monitoring Results

Number of Zebra Mussels Found:	**Note:** If more than 20 zebra mussels are found, measure 20 mussels chosen randomly from the sample. If less than 20 mussels are found, measure all mussels. Report results in the table on page 2 of this form.
# on Top Side of Plates:	
# on Bottom Side of Plates:	
# Total on Sampler:	
Size of Largest Zebra Mussel:	
Size of Smallest Zebra Mussel:	

Zebra Mussel Monitoring Report
Method B – Quantitative

Form 3200-122B (3/05) Page 2 of 2

Length of Zebra Mussels from Sample

If more than 20 zebra mussels are found, measure 20 mussels chosen randomly from the sample. If less than 20 mussels are found, measure all mussels.

Number	Length (mm)
1	
2	
3	
4	
5	
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7	
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10	
11	
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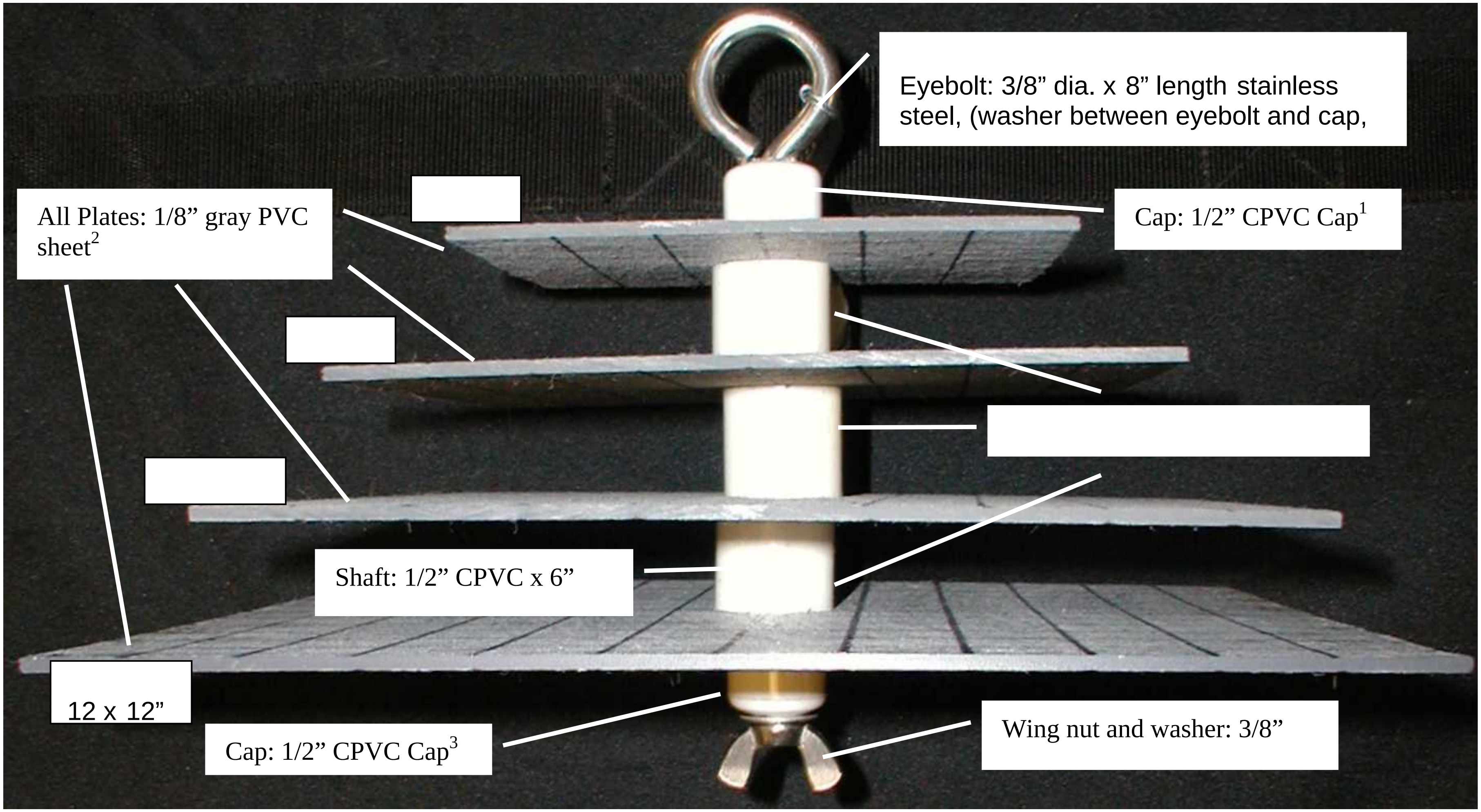
20	

Note: All initial discoveries should be placed in rubbing alcohol until verification by an expert is obtained.

Appendix 4. Substrate Sampler Construction Directions



[1]Solvent weld top cap to shaft and drill a 13/32" hole in cap
[2]Drill 5/8" hole in plates to accept shaft
[3]Drill a 13/32" hole in bottom cap

Appendix 5. Press Release Templates for the Watch List and Infested List.

Press Release Template for Waterbodies to be Placed on the Zebra Mussel Watch List

_____ (numbers) zebra mussel larvae/juveniles/adults were recently found in ____ (waterbody) in ___(county). The larvae were detected as part of routine monitoring on ____waterbody OR the juvenile/adult zebra mussels were found attached to a plate sampler OR were discovered as part of a general lake/river survey (i.e., examining shores, piers and other hard substrates).

"The zebra mussels/larvae that were found do not provide sufficient proof that a reproducing population of zebra mussels is established in ___ (waterbody)" according to _______ from the ____ Regional office. "The ____(waterbody) will be placed on DNR's 'Watch List' because it's too early to tell whether they will survive and thrive in ____ (waterbody).

Placing ____ waterbody on the 'Watch List' means DNR will target this waterbody for additional follow-up monitoring efforts and will work with local units of government to increase public awareness efforts and outreach efforts. It also affords volunteers the opportunity to become more involved by helping monitor ____waterbody for zebra mussels and/or through watercraft inspection efforts.

If additional monitoring efforts by DNR staff indicate that there is an established, reproducing population of zebra mussels present in _____ waterbody, then it will be placed on the list of infested waters. DNR would follow-up by posting the 'Exotic Species Advisory' signs with zebra mussel decals at all the landings to notify the public that ____ waterbody is infested.

"Zebra mussels are sometimes introduced into waters, but they do not survive in all cases," says ____. "That's why it's particularly important that boats are always clean when they leave a waterbody so there are fewer introductions and less chance of zebra mussels getting established."

Zebra mussels first arrived in the Wisconsin waters of Lake Michigan in the Racine harbor in1990 as stowaways aboard foreign freighters entering the Great Lakes. Since then they have been making their way into our inland waters. The zebra mussels form dense clusters that attach to hard surfaces, and can decimate native mussel populations, decrease the oxygen that fish and other aquatic species need, and worsen smelly, unsightly algal blooms. In addition, the zebra mussels can clog boat engines and intake pipes for utilities, and their sharp shells can wash up on shore and make walking on the beach hazardous.

“The good news is that there are prevention steps that everyone should take when boating, fishing and otherwise enjoying the water that can help prevent the spread of invasive species,” says _______.

Before moving your boat/equipment to a new waterbody:

1. Inspect and remove plants, animals, and mud from your boat and equipment;
2. Drain all water from your boat’s live wells, bilge, motor, etc.;
3. Dispose of your unwanted live bait in the trash;
4. Spray/rinse your boat and equipment with high-pressure and/or hot water, especially if moored for more than a day; OR
   Dry your boat and equipment thoroughly for at least 5 days.

FOR MORE INFORMATION CONTACT: Regional ANS Coordinator and/or regional biologist (two contacts are preferred)

Press Release Template for Waterbodies to be Placed on the Zebra Mussel Infested List

_____ (numbers) zebra mussel larvae/juveniles/adults were recently found in ____ (waterbody) in ___(county). The larvae were detected as part of routine monitoring on ____waterbody OR the juvenile/adult zebra mussels were found attached to a plate sampler OR were discovered as part of a general lake/river survey (i.e., examining shores, piers and other hard substrates).

"The zebra mussels/larvae that were found indicate that there is a reproducing population of zebra mussels is established in ___ (waterbody)" according to _______ from the ____ Regional office. "The ____(waterbody) will be placed on DNR's 'Infested List'.

Placing ____ waterbody on the 'Infested List' means DNR will post the 'Exotic Species Advisory' signs with zebra mussel decals at all the landings to notify the public that ____ waterbody is infested. The DNR will also work with local units of government to increase public awareness efforts and outreach efforts to control the further spread of zebra mussels. It also affords volunteers the opportunity to become more involved by helping monitor ____waterbody for zebra mussels and/or through watercraft inspection efforts.

Zebra mussels first arrived in the Wisconsin waters of Lake Michigan in the Racine harbor in 1990 as stowaways aboard foreign freighters entering the Great Lakes. Since then they have been making their way into our inland waters. The zebra mussels form dense clusters that attach to hard surfaces, and can decimate native mussel populations, decrease the oxygen that fish and other aquatic species need, and worsen smelly, unsightly algal blooms. In addition, the zebra mussels can clog boat engines and intake pipes for utilities, and their sharp shells can wash up on shore and make walking on the beach hazardous.

"There are prevention steps that everyone should take when boating, fishing and otherwise enjoying the water that can help prevent the spread of invasive species," says _______.

Before moving your boat/equipment to a new waterbody:

1. Inspect and remove plants, animals, and mud from your boat and equipment;
2. Drain all water from your boat's live wells, bilge, motor, etc.;
3. Dispose of your unwanted live bait in the trash;

4. Spray/rinse your boat and equipment with high-pressure and/or hot water, especially if moored for more than a day; OR
   Dry your boat and equipment thoroughly for at least 5 days.

FOR MORE INFORMATION CONTACT: Regional ANS Coordinator and/or regional biologist (two contacts are preferred)

Appendix 7. Wisconsin Department of Natural Resources sampling protocol for Eurasian water milfoil and curly leaf pondweed.

Eurasian water-milfoil & Curly-leaf Pondweed Monitoring Protocol

Citizen Lake Monitoring Network

Pilot to be used in 2006

Eurasian water-milfoil

Curly-leaf pondweed

Eurasian water-milfoil and Curly-leaf Pondweed Overview

Education is the best defense against Eurasian water-milfoil and curly-leaf pondweed. It is estimated that human carelessness accounts for 95-97% of the spread of Eurasian water-milfoil. Eurasian water-milfoil only needs a 2-3 inch plant fragment to start a new colony on a “clean” lake. If you recreate on a lake with Eurasian water-milfoil and pick up a piece of plant material on your boat, trailer, jet ski, fishing equipment, etc., you could haul that plant material to an un-infested lake. That piece of Eurasian water-milfoil has the potential to grow roots and settle to the bottom of the lake starting a new colony of Eurasian water-milfoil. Curly-leaf pondweed turions are sometimes carried in muck attached to an anchor or dropped in the bottom of your boat. These turions can sprout and grow new curly-leaf pondweed colonies. Be sure to remove all aquatic plants from boating equipment, including your trailer, boat, motor/propeller and anchor before launching and after leaving the water. By removing aquatic plants from boating equipment and encouraging others to do the same, you can help protect Wisconsin lakes from exotic invasives. Anther way to protect your lake from invasives is to protect native plants beds. “Research has shown that abundance of Eurasian water-milfoil is inversely related to cumulative native plant cover. It is important to maintain aquatic plant communities as a buffer against non-native plants.” Madsen, 1998 Predicting Invasion Success of Eurasian water-milfoil, US Army Corps. of Engineers.

When an invasive plant is suspected or found, contact your local DNR Aquatic Plant Management Specialist. Your lake organization will want to consider control efforts for these invasives. Your DNR Lake Coordinator can go over grant options and control methods at this point. It is easiest to control and potentially eradicate an invasive plant if the invasive is found in the pioneer stage. In the pioneer stage, the plants can be hand pulled. For shallow water areas, a rake can be used to remove the roots. In deeper areas, you may want to consider hiring a SCUBA diver to hand pull the plants and roots. Dispose of the plants well away from the lake so that they do not wash back into the lake during the next rain event. If caught early enough, hand removal may eradicate the invasive from the lake. When the plant beds get larger, you may want to consider chemical control followed up by hand pulling. Chemical control is not 100% effective, so a second control method is often used to increase the control. Once Eurasian water-milfoil or curly-leaf pondweed become well established in your lake, it may be impossible to fully eradicate them. Early detection gives you the best chance of eradicating the invasive and will save you money.

Eurasian Water-milfoil

Eurasian Water-milfoil Background

There are 11 native water-milfoil species in North America. Of these 11 native species, 7 are found in Wisconsin. Eurasian water-milfoil (EWM) is a plant introduced to the United States from Europe, Asia and northern Africa. EWM may have been brought in to the United States via the aquarium trade. Since it is an exotic (not native to Wisconsin or the United States) it has very few natural predators. The first authenticated record of EWM in the United States was in 1942 in a Washington D.C. pond. Since then, it can now be found in 48 of the 50 states. EWM was first documented in Wisconsin in the 1960’s. As of December 2005, EWM has been verified in 458 water bodies in Wisconsin. EWM poses a serious threat to the lake’s native aquatic plant communities and the animals that depend on these diverse ecosystems. EWM can form thick underwater stands of tangled stems and vast mats of vegetation at the water's surface. It can crowd out native plants and can become so thick that the larger fish cannot swim through the tangled mats. EWM can adversely affect property values. Under severe conditions, channels are needed to allow access from the shoreline out into deeper water areas. EWM is now one of the most troublesome of submerged aquatic plants in Wisconsin. Volunteers play an integral part in learning to recognize the plant and checking local lakes for the presence of EWM. Early identification of the plant makes control much easier, and can help prevent the spread into other waterbodies. Refer to Appendix 6 for lakes with Eurasian water-milfoil.

Eurasian Water-milfoil Identification

In your packet is a card that shows you a picture of Eurasian water-milfoil on one side of the card and northern water-milfoil on the other side. Northern water-milfoil is a Wisconsin native that is sometimes confused with EWM. The native milfoils are not as aggressive as the exotic milfoil and have natural predators. Some Wisconsin species of water-milfoil are quite rare and on the Threatened and Endangered list.

Eurasian water-milfoil:

- EWM has delicate feather-like leaves.
- The thread-like leaflets, on the lower part of the leaf, are mostly the same length.
- Leaves are fairly limp when pulled out of the water.
- Leaves are arranged in whorls (circles) of 3 to 5 around the stem.
- Usually there are 12-21 leaflet pairs per leaf.
- In the summer, the plants can be 20 feet tall.
- In the summer, the distance between the leaf whorls can be several inches.
- EWM does not produce winter buds.
- Upper part of the plant stem often has a pink or reddish color. Other water-milfoils may also be pink.

EWM whorl showing 4 leaves with leaflets.
Note distance between whorls on plant stem.
WI DNR photo

EWM is limp when out of water
www.weedmapper.org photo

EWM often develops adventitious roots along its stem
WI DNR photo

Northern water-milfoil

- Northern water-milfoil has rigid feather-like leaves.
- Leaves are arranged in whorls (circles) around the stem.
- When looking at an individual leaf, you may notice a Christmas tree shape.
- The lower leaflets are usually longer than the upper leaflets.
- Usually there are 7-10 leaflet pairs per leaf.
- Stems are often whitish or whitish green in color.
- Leaves are stiff when the plant is removed from the water.
- Most native water-milfoils produce winter buds while EWM does not.

Northern water-milfoil whorl showing 4 leaves with leaflets. Note distance between whorls on plant stem. Note winter bud at tip of plant.
WI DNR photo

Northern water-milfoil is stiff when out of water.
www.wes.army.mil/el.aqua photo

Most native species of water-milfoil have winter buds.
Eurasian water-milfoil does not.

Eurasian Water-milfoil Life Cycle
Eurasian water-milfoil is an evergreen plant. The plant remains alive over the winter and starts growing when water temperatures reach 50^{o} F (Bode, J. et al. 1992). In spring and summer, Eurasian water-milfoil can grow up to 2 inches a day and can shade out native plants. If EWM plant growth reaches the surface of the lake, the plant will continue to grow and will canopy over the surface of the lake often making this area almost impassable with a motor boat. Excessive growth affects recreational use of lakes by interfering with swimming, fishing, boating and reduces the aesthetics of the lake. EWM grows in water depths of less than a foot to water depths of a little over 20 feet. Thick beds can form in water depths from 3 to 20 feet deep (Smith, C and J. Barko, 1990), but most commonly reach nuisance levels in water depths of 6-15 feet. Eurasian water-milfoil produces seeds and runners, but the main method of spread is through plant fragmentation (vegetative propagation) by boats and wave action. In the late summer and early fall, auto fragmentation may occur. Auto fragmentation is where the plant will "break itself apart". Some of the plant cells at leaf nodes and side-branch connections become weak and die off. These newly formed sections and branches break off and float to new locations where they fall to the substrate, take root and establish new beds of EWM.

Curly-leaf pondweed

Curly-leaf Pondweed Background
Curly-leaf pondweed (*Potamogeton crispus*) is native to the fresh waters of Eurasia, Africa and Australia. This aquatic plant first found its way to the United States in the mid 1800's. It is thought to have made its way to Wisconsin in 1905 along with fish imported from Europe. Agency staff has just begun tracking lakes that have Curly-leaf pondweed thus there is not a complete list of lakes with Curly-leaf pondweed. We need your help in this tracking.

Curly-leaf pondweed has a unique life cycle. The plant begins growing in the fall, grows very slowly under the ice, has a large growth spurt from ice out to early spring, and then dies back in July. In June and July, CLP can form dense mats of vegetation on the surface. The die back of curly-leaf can cause rafts of dying plants. When this die-back takes place, nutrients (phosphorus) are released and these nutrients fuel algal blooms.

Curly-leaf pondweed is one of 80 pondweed species found throughout the world. In some situations, native vegetation can be displaced by CLP. Curly-leaf pondweed is tolerant of disturbance and can grow in most water conditions.

Curly-leaf Pondweed Identification

Curly-leaf pondweed:

- CLP is recognized by alternate leaves that are minutely toothed (you may need a magnifying glass to see the teeth).
- The leaf edges are also wavy giving it a crispy appearance.
- Most leaves have a prominent red-tinged mid-vein.
- The stem is slightly flattened.
- A short flower stalk rises above the water's surface, though the rest of the plant is submersed.
- CLP does not form floating leaves.
- CLP produces turions, vegetative buds, that sprout in late summer and produce new plants.

Curly-leaf pondweed can be confused with Clasping-leaf pondweed (*Potamogeton richardsonii*). Clasping-leaf pondweed does not have toothed leaf edges.

CLP leaves are often light green and fairly transparent. www.ppws.vt.edu/ photo

Note the "lasagna" wavy leaves of CLP. S. Knight photo

CPL turion. Frank Koshere photo

Clasping-leaf pondweed. www.mlswa.org photo

Curly-leaf Pondweed Life Cycle
Most of our native aquatic plants come out of dormancy in spring and reach their maximum growth in late summer or early fall. But, curly-leaf is different and has a natural inclination for low water temperatures, which helps it to avoid competition with other plant species. Seeds are produced that may be fertile but vegetative reproduction tends to be more important for the dispersal of this plant. Turions are probably the most reliable form of reproduction. A turion is a dormant shoot segment or vegetative bud that can form most anywhere on the plant. It is a hard structure that looks a little bit like a burr or pinecone.

In northern Wisconsin, curly-leaf plants usually complete their life cycle by late June or early July. The turion, which has developed on the plant, falls to the bottom of the lake. The turions begin to sprout in late summer, responding either to the shortening day length or to water temperature. The new growth continues even under the ice of winter. A few days after ice off, CLP begins to grow more rapidly and attains its spring foliage (the leaves on the plant in winter and very early spring are quite narrow and lack the wavy edges). The fast growth allows the stems to reach the water's surface before any other plant. By late spring, a dense canopy of curly-leaf may have formed blocking sun light from reaching other plants. At this time, the curly-leaf pondweed develops turions which drop to the bed of the lake and the plant itself dies back and begins to decay. If you notice that plants on your lake are dying back in late June or early July, you will want to check to see if it is Curly-leaf pondweed.

Eurasian Water-milfoil and Curly-leaf Pondweed Surveys

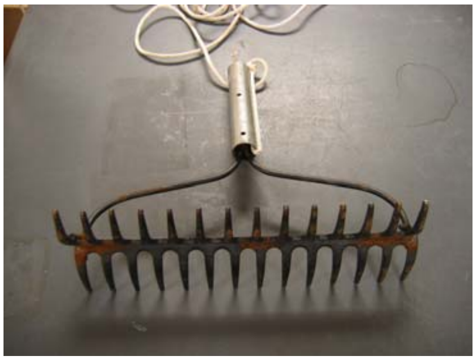
Equipment

- Boat (canoe, kayak, fishing boat, paddle boat, etc.)
- Personal Floatation Device (PFD)
- Long-handled rake with attached rope (see pictures)
- Lake map for marking suspect EWM or CLP beds and keeping track of where you have been.
- Pencil for marking on map
- Data forms (appendix 4)
- Clip board or other hard surface for writing
- Ziploc bags

- Waterproof sharpie pen (to write on Ziploc bags)
- Cooler to keep plants in
- Plant density data sheet (optional)
- GPS unit (optional)
- Polarized sunglasses (optional)
- Aqua-View Scope (optional). Construction directions in Appendix 3.


The “2-headed” garden rake


A rope is tied to the handle of the “2-headed” rake


This “2-headed” rake is used in deep water.

Since it is sometimes difficult to identify plants under water, volunteers rake up plant samples. The “2-headed” garden rake is made by purchasing 2 garden rakes (try looking at garage sales for broken rakes). Disconnect the head from one rake and wire or weld the rake heads together (teeth facing out). Drill a hole in the handle end of the rake. Tie a rope on the handle, and you can sample in deeper water. When the rake is thrown into the water, it settles to the bottom of the lake. When the rake is hauled back into the boat, aquatic plants come with it making for easier identification. With the two heads, no matter which way the rake falls to the lake bed, the teeth will catch the roots of the plants making plant collection a lot easier. If you need to make the rake heavier, you can attach some duck decoy weights. Some volunteers do not like to mess with the rake handle in deeper water, so they cut off the rake handle and attach the rope directly to the rake heads. No matter which rake is used in deeper water, please make sure you tie the loose end of the rope to the boat. This way you will not lose your sampling rake.

Make sure the weather will allow for successful and safe sampling. Clear calm weather is the best for sampling. Sunny skies make it easier to see into the water. Polarized sunglasses or Aqua-View Scope (Appendix 3) will help you to see the plant beds. Check your lake from ice off until mid-September. If you notice that plants suddenly disappear in late June, it may be CLP. If you notice water-milfoils growing when the water temperature is cold, it could be EWM.

Please complete and return one of the enclosed reporting forms each time you sample, whether or not you find EWM or CLP. **Please mail the EWM and/or CLP reporting forms to your local Citizen Lake Monitoring Network contact (pages vii - viii).**

Minnesota research has shown that the most susceptible lakes to invasives are those that are close to lakes with established aquatic invasive species (especially if the nearby lake has had the invasive for years). These will be lakes you want to target in your monitoring.

Setting up a Monitoring Team

Refer back to Section 1, pages 6-7 for suggestions on how to set up a monitoring team and how to divide up the workload.

When to Conduct Surveys

Many groups will monitor for Eurasian water-milfoil several times a season as Eurasian water-milfoil is an evergreen plant and begins growing early (when water temperature is about 50 degrees F) and keeps growing late into the fall. For lakes with known Eurasian water-milfoil you should look for new beds early in the season so that these beds can be treated (scuba diving, hand pulling, chemical, etc.) while the beds are still small. For lakes without know Eurasian water-milfoil infestations, you may want to conduct your monitoring late spring to mid summer when the Eurasian water-milfoil biomass is at its greatest. Some teams will monitor from ice out to ice on. You will want to monitor several times through out the open-water season so that you catch the beds as early as possible. Most groups monitor on a 3-4 week interval. Please note that spring drought conditions cause high growth in EWM during the early growing season so you will want to monitor earlier is these years.

Curly leaf pondweed is often at peak densities in May and June and begins to die back in July, thus you would want to conduct your monitoring in May or June. Since CLP is normally only dense for a few months, most groups monitor every 2-3 weeks. Some groups also check for Curly-leaf pondweed in the late fall as the new plants will be growing at this time and the native pondweeds are dying back. This way they can treat those beds as early in the spring as possible. This will increase the chances for control of CLP.

Where do I Look?

Eurasian Water-milfoil Habitat Background

EWM probably has the capability to survive in all lakes in Wisconsin if it gets established. It can tolerate a wide range of conditions. EWM can grow in water depths of less than 1-foot to water depths of a little over 20-feet. In Wisconsin, EWM gets the most dense in water depths of 6-15 feet, but can reach nuisance levels in as little as 2 feet. EWM can grow in the clearest of lakes to some of the most turbid lakes, but it does best in moderate to highly fertile lakes. EWM can grow in rocky areas to sandy areas to mucky areas, but does best in areas with silt and sediment. It even has the ability to survive in wetland areas, although it will not grow to be dense in these areas. Please remember, EWM will grow throughout the entire lake where water depths are less than 20 feet, so do not just rely on monitoring these “prime habitat” areas.

Curly-leaf Pondweed Habitat Background
Curly-leaf pondweed can survive in a wide range of lake conditions. It grows in water depths of less than 1-foot to water depths of about 15-feet. In Wisconsin, CLP gets the most dense in water depths of 3-10 feet, but can reach nuisance levels in as little as 1-foot to as deep as 15-feet. CLP does best in moderate to highly fertile lakes and does well in turbid water conditions. CLP is often associated with degraded water quality. CLP can live in sandy soils, but prefers soft substrates. Please remember, CLP will grow throughout the entire lake where water depths are less than 15feet, so do not just rely on monitoring these “prime habitat” areas.

Where to Start Monitoring
Even before looking for the beds of EWM and CLP, you will want to look for floating plants. Think about your lake. Which way does the wind blow from and where does the wind blow the plants and floating debris to. Go to the areas where you have seen the piles of plants and debris. Look in these piles to see if you can find any EWM or CLP plant fragments. It is especially important to visit these areas **after storms and high boat traffic times** as this is when the plant fragments will be the heaviest. If you find any EWM or CLP fragments here, you know that the invasive is in your lake. Check **beach areas, inlets, boat launches, high use areas and perimeter of the lake**. In mid-summer, EWM may start to break up into smaller pieces and these pieces often wash up along shorelines. These smaller fragments – pieces of stem torn by waves or boaters – may establish new populations.

Whole Lake Monitoring
Boat or walk around the shoreline of your lake and look for the invasives in the shallow water areas. Look for EWM and CLP in both sand areas and in mucky areas. Both EWM and CLP will grow in a variety of sediment conditions, but will do the best in areas with a mucky bottom. Once you think you have monitored a variety of near shore areas, go out in your boat and begin to collect plants in the deeper water areas. It will be easiest to see the plants if you are wearing polarized sunglasses and/or using an Aqua-View Scope. Use the rake and the rake on a rope to collect plants that are hard to reach or difficult to identify. You can lower the rake to the bottom of the lake and drag the rake along. Pull the rope so that the rake pulls along several feet of the lake bed. This makes for relatively easy monitoring of deep water areas. This method will also help you pull up roots and collect plants that are not readily visible from the lakes surface. Be sure to monitor over sand as well as muck areas.

NOTE: Please do not throw plants that you collect back into the lake. Instead, dispose of them on shore or take them for mulch or compost for your garden. If you toss back plants, you may inadvertently spread plants to different locations on the lake. Since many do not know which plants are native and which are non-native, it is best not to throw any plants back into the lake.

Mapping
Most people comprehend faster when given information in a visual format. A map is a very quick and reliable way to assure that everyone knows the place you are talking about when you describe a certain point on your lake. A map will assist you in locating plant communities, recreational and habitat use areas, and more. A map will also assist your team in deciding who will monitor where. At the end of the season, you can map all of the sites visited. Refer to Section 1, pages 6-7 on websites where you can download maps. Mark the following

information on your lake map: lake name, county, date, volunteer(s), and any additional observations.

GPS
If you have a GPS unit, you may want to mark in the edges of the beds, and then you can load this data into a mapping program and print out maps of the beds.

What to do with Suspect Plants
Even if your lake group controlling EWM or CLP, you still want to monitor for these plants. You want to know if they have spread to any new locations so that you can begin control of these new beds ASAP. The earlier you catch a new infestation, the easier it will be to control.

- Note the "suspect" plant's location on your map, making sure you can find the spot(s) again. Use report form in Appendix 4.
- You will need to bring a fresh sample to your local contact. To collect a specimen of the plant, gently pull the plant from the lake bottom. Be sure to collect as much of the plant as possible, paying special attention to getting the leafy and flowering portion, if present. Try not to break up or rip the plant as the pieces of the plant that float away can form roots and start new plants.
- Use a permanent marker and record the following information on the plastic bag:
    a. Date
    b. Water body
    c. Description of where the sample was found.
- Put the sample in a plastic bag and keep it in a cool place (a cooler in your car or refrigerator at home). Take the specimen to your team's point person, your local Land and Water Conservation District personnel, UW-Extension office or the local DNR contact for identification. You will want to get these plants vouchered ASAP, so that control can take place in a timely manner. **NOTE – if you lake has been verified to have EWM or CLP, samples do not need to go to the DNR for vouchering – you can just take the plants to your point person.**
- If you cannot bring the plant in for vouchering, rinse the plant under running tap water or in a large pan. This will slow the rotting process.
- Blot the plant dry with a paper towel.
- Spread the plant out on a dry paper towel or newspaper. For water-milfoil, try to spread the leaflets apart to help with identification.
- Cover with a dry paper towel and press in a catalog or phone book for about a week.
- Complete the label (Appendix 2) and reporting form (Appendix 4)
- When the plant is dry, place it between sheets of thin cardboard (like a cereal box). Mail the plant, map and the reporting form to your local Citizen Lake Monitoring Network Contact.
- Remember to make a copy of your map and data sheets for your records.

Remember if you find "something," don't give up; there are a variety of control and management options to address invasive species on your lake. Early detection is the key to controlling the situation!

If you find beds of EWM and or CLP, you may want to determine how dense the beds are. This information will be very useful when determining the proper control method for your invasive.

PLANT DENSITY

Use the following numbers to denote the plant density for each invasive aquatic plant bed found: Rake fullness ratings are given from 1-3. Conditions of the ratings are described below:

Rating	**Coverage**	**Description**
1		A few plants on rake head
2		Rake head is about ½ full Can easily see top of rake head
3		Overflowing Cannot see top of rake head

NOTE: Please do not throw plants that you collect back into the water. Instead, dispose of them on shore or take them for mulch or compost for your garden.

Other Data You may want to collect (appendix 2)

Sample Location: Record the sample GPS position.

Depth: Measure depth at each sampling site regardless of whether vegetation is present. A variety of options exist for taking depth measurements, including SONAR guns, depth finders that attach to the boat, or an anchor attached to a line with depth increments.

Dominant Sediment Type: Record sediment type (based on how the rake feels when in contact with the bottom) at each site where plants are sampled as: (a) mucky, (b) sandy, or (c) rocky.

Here are a few plant identification sources you may find helpful:

Through the Looking Glass. 1997. Susan Borman, Robert Korth, Jo Temte. Wisconsin Lakes Partnership. DNR publication # FH-207-97.

Common Aquatic Plants of Wisconsin list prepared by Stan Nichols, Wisconsin Geological and Natural History Survey, Madison, WI. (This is not a true key, but it is easy for all to use)

Aquatic and Wetland Plants of Northeastern North America. Garrett E. Crow and C. Barre Hellquist. The University of Wisconsin Press.

A Manual of Aquatic Plants by Norman C. Fassett. 1957. University of Wisconsin Press.

Aquatic Plants of Illinois by Glen S. Winterringer and Alvin C. Lopinot. 1966. Department of Registration and Education, Illinois State Museum Division and the Department of Conservation, Division of Fisheries.

Michigan Flora by Edward G. Voss. 1985. University of Michigan Press.

Appendix 8. Great Lakes Indian Fish and Wildlife Commission aquatic invasive species survey structure.

The following text was provided by D. Olson in 2006. Great Lakes Indian Fish and Wildlife Commission, P.O. Box 9, Odahnah, Wisconsin 54891.

Each survey lake is visited twice during the field season. Two visits allow an increased likelihood of observing plants by accounting for differences in phenology. Two visits also allowed for zebra mussel veliger samples to be taken twice in the season, increasing the chances of detecting veligers.

Surveys targeted the most likely areas for introductions. Boat landings were a high priority. All public and some private boat landings on each lake are surveyed. Shoreline, shallow water areas, pier supports, rocks, floating fragments and beach debris are inspected at the landings for invasive plants and animals.

Surveys also focused on inlets, outlets, shallow or protected bays, wetland areas, disturbed areas, and developed shorelines or ones in close proximity to roads. As much of the lake shoreline as possible is surveyed within the time available focusing first on target areas. Shorelines are surveyed from the outer edge of or occasionally within the littoral zone from a slow-moving boat checking any suspicious looking patches of vegetation including submerged, emergent, and riparian plants. While checking out suspicious plants, the area is also surveyed for invasive animals or evidence of these animals.

Locations of invasive plants are recorded at approximately the center of the patch, along with basic information on the number of plants, patch size and habitat. If unable to access the center of the population or doing so would cause unnecessary disturbance, these populations are hand plotted using maps on the GPS unit and noted in data collection.