Minutes
Hess Lake Improvement Board
July 16, 2018
6:30 p.m.

Board Members Present: Rosen, Twing, Lipner, Bosowski
Board Members Absent: Willet
Also Present: Matt Novotny- Savin Lake Services, Tony Grove-Progressive AE, Dan Nowak, Blake Pierce, John Van Dam, Elizabeth Sochacki, Jerry Swendrowski, Jamie Deward, Del Hirdes, John Ripstra, Kevin Houle, David Huistra, Nancy Calvi, Bart Calvi, Betty Pennington

1. The meeting was called to order at 6:30 p.m. by Rosen

2. Agenda:

   Motion by Lipner, seconded by Bosowski to approve the agenda. All ayes. Motion carried.

3. Approval of Minutes of 5/14/2018 Meeting

   Motion by Lipner, seconded by Bosowski to approve the minutes of the 5/14/2018 meeting. All ayes. Motion Carried.

4. Public Comment: (2 minutes)

   A major discussion happened during public comment revolving around diverting Wheeler Drain, dredging the lake, limiting the size of boats, Brooks Township helping with cost of dredging or some other project, the carp issue, sewers and septic systems, seawalls, and all the other issues that contribute to the health of the lake.

   The board was thanked for having a night meeting so that people could attend.

   There were also comments about wasting money on more studies. All of these issues were addressed in a give and take of information and opinion as has happened in the past with differing of opinions and no clear resolution.

   The aerators were talked about also with differing opinions on whether they are good or bad or of any use at all.

5. Financial Report:

   Motion by Lipner, seconded by Bosowski to accept the Treasurers Report as submitted. All ayes. Motion carried. Twing also stated he was going to check on the electric bills to make sure they are paid in full and correctly.

6. Old Business:

Matt Novotny of Savin lake Services explained their treatment and what was allowed by the DEQ. He stated they can only treat for evasive plants and that was Eurasian Milfoil and Curly Pond Weed. He also explained why we don't treat for algae. Because of the high amount of algae in the water at Wheeler the treatment was fairly ineffective so it was discontinued.

Tony Groves discussed what he was doing to help us with developing a good overall lake management plan. He answered some questions and stated that he was using past information and new information to determine the most cost effective ways to improve the lake. Information will be shared as he proceeds with the development of the plan, however most recommendations will come after a thorough accumulation of data is in hand. He supplied the board with handouts that are included in the minutes.

7. New Business

The board discussed the finding of Zebra Mussels by Del Hirdes. It was explained that Brooks Lake has had them for at least 4years and that the local seawall contractor says they have been in Hess for at least 3 years. Currently not much is done about them in other lakes and the conditions don't appear to be such that they would be a big problem in Hess Lake.

A letter was sent from Brooks Township Supervisor Cory Nelson from their attorney, Cliff Bloom on the ablility of the township helping with a lake improvement specifically dredging. His opinion basically state they probably could. A discussion ensued about the costs and the ability to get approval from the DEQ. The board has always been open to this and will continue to keep it in mind to do as soon as we can get a proposal that can be approved and we can afford.

8. Correspondence/Bills

Motion by Twing, seconded by Lipner to approve payment of electric bill for aeration of $215.53 and the Savin Bill of $11,595.70 All ayes. Motion carried.

Motion by Twing, seconded by Lipner to accept the treatment notice and liability coverage statement from Savin. All ayes. Motion carried.

9. Public Comment (2 minutes)

The general lake issues and study process were again discussed at length. Everyone wants to improve the lake, and the board welcomes any and all contributions of thought, and wishes to find as many ways to help the lake that the property owners can afford and that the DEQ will allow.

10. Miscellaneous

Next meeting is October 1st, 2018 at 10:00 a.m.

11. Adjournment

Motion by Lipner, seconded by Bosowski to adjourn the meeting. All ayes. Motion carried.

Meeting adjourned at 8:15 p.m.

____
Dale E. Twing, Secretary

Hess Lake
June 5th Treatment
Legend
Treatment Area
Hess Lake
Red Run Ln
E 88th St
John St
Vista St
Pine St
Lockwood Ct
Park Blvd
S Lovers Ln
E Maple
Redwood Dr
Hess Lake Dr
Maple St
Benjamin Dr
Annie Ln
Ash St
S 1st St
S 2nd St
E Hess Lake Dr
Catalpa Ave
95th St
E 95th St
Forest Ln
E Oak Ave
Clay St
Brooks
E 96th St
Peninsula Dr
S West St
Willow Ave
E 98th St
Google Earth
© 2018 Google
4000 ft
N


Hess Lake
July 9th Treatment
Legend
Treatment Area
Red Run Ln
E 88th St
John St
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Peninsula Dr
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S West St
Willow Ave
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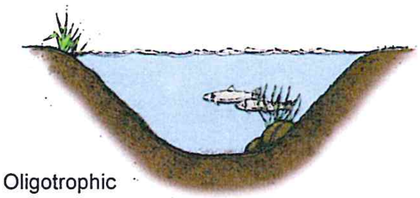
michiganlakeinfo
www.michiganlakeinfo.com

Lake Water Quality

April / 2014

by Tony Groves, Pam Tyning, and Paul Hausler
Progressive AE

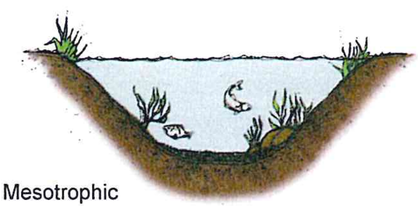
Lakes can be classified based on their ability to support plant and animal life. When classifying lakes, scientists use the broad categories "oligotrophic," "mesotrophic," or "eutrophic."



Oligotrophic

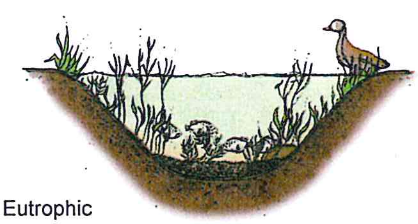
Oligotrophic lakes are generally deep and clear with little aquatic plant growth. These lakes maintain sufficient dissolved oxygen in the cool, deep bottom waters during late summer to support cold-water fish such as trout and whitefish.

Eutrophic lakes have poor clarity and support abundant aquatic plant growth. In deep eutrophic lakes, the cool bottom waters usually contain little or no dissolved oxygen. Therefore, these lakes can only support warm-water fish such as bass and pike.



Mesotrophic

Lakes that fall between the two extremes of oligotrophic and eutrophic are called **mesotrophic** lakes.



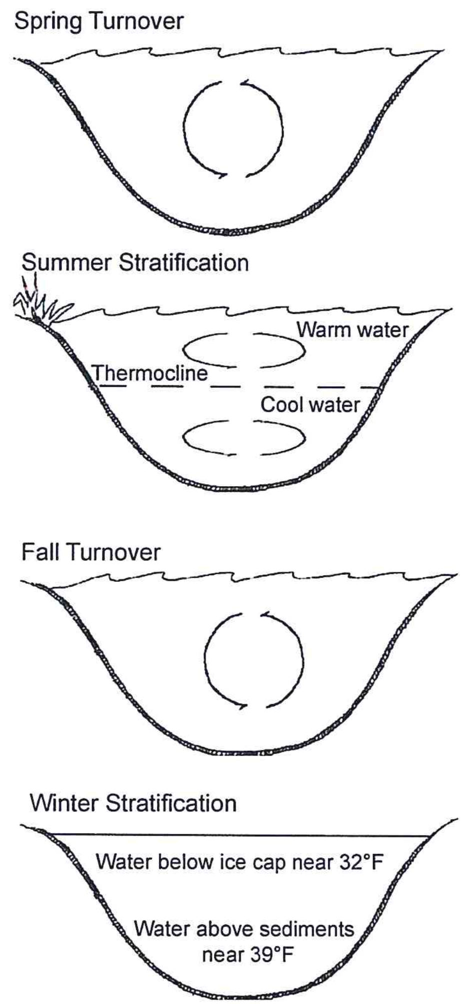
Eutrophic

Under natural conditions, most lakes will ultimately evolve to a eutrophic state as they gradually fill with sediment and organic matter transported to the lake from the surrounding watershed. As the lake becomes shallower, the process accelerates. When aquatic plants become abundant, the lake slowly begins to fill in as sediment and decaying plant matter accumulate on the lake bottom. Eventually, terrestrial plants become established and the lake is transformed to a marshland. The natural lake aging process can be greatly accelerated if excessive amounts of sediment and nutrients (which stimulate aquatic plant growth) enter the lake from the surrounding watershed. Because these added inputs are usually associated with human activity, this accelerated lake aging process is often referred to as *cultural eutrophication*.

There are many ways to measure lake water quality, but there are a few important physical, chemical, and biological parameters that indicate the overall condition of a lake. These measurements include temperature, dissolved oxygen, total phosphorus, chlorophyll-*a*, and Secchi transparency.

Temperature

Temperature is important in determining the type of organisms that may live in a lake. For example, trout prefer temperatures below 68°F. Temperature also determines how water mixes in a lake. As the ice cover breaks up on a lake in the spring, the water temperature becomes uniform from the surface to the bottom. This period is referred to as "spring turnover" because water mixes throughout the entire water column. As the surface waters warm, they are underlain by a colder, more dense layer of water. This process is called "thermal stratification." Once thermal stratification occurs, there is little mixing of the warm surface waters with the cooler bottom waters. The transition layer that separates these layers is referred to as the "thermocline." The thermocline is characterized as the zone where temperature drops rapidly with depth. As fall approaches, the warm surface waters begin to cool and become more dense. Eventually, the surface temperature drops to a point that allows the lake to undergo complete mixing. This period is referred to as "fall turnover." As the season progresses and ice begins to form on the lake, the lake may stratify again. However, during winter stratification, the surface waters (at or near 32°F) are underlain by slightly warmer water (about 39°F). This is sometimes referred to as "inverse stratification" and occurs because water is most dense at a temperature of about 39°F. As the lake ice melts in the spring, these stratification cycles are repeated. Shallow lakes do not stratify. Lakes that are about 15 to 30 feet deep may stratify and destratify with storm events several times during the year.



Dissolved Oxygen

An important factor influencing lake water quality is the quantity of dissolved oxygen in the water column. The major inputs of dissolved oxygen to lakes are the atmosphere and photosynthetic activity by aquatic plants. An oxygen level of about 5 mg/L (milligrams per liter, or parts per million) is required to support warm-water fish. In lakes deep enough to exhibit thermal stratification, oxygen levels are often reduced or depleted below the thermocline once the lake has stratified. This is because deep water is cut off from plant photosynthesis and the atmosphere, and oxygen is consumed by bacteria that use oxygen as they decompose organic matter (plant and animal remains) at the bottom of the lake. Bottom-water oxygen depletion is a common occurrence in eutrophic and some mesotrophic lakes. Thus, eutrophic and most mesotrophic lakes cannot support cold-water fish because the cool, deep water (that the fish require to live) does not contain sufficient oxygen.

Phosphorus

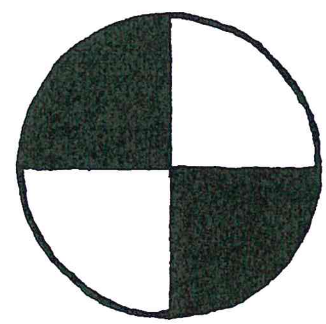
The quantity of phosphorus present in the water column is especially important since phosphorus is the nutrient that most often controls aquatic plant growth and the rate at which a lake ages and becomes more eutrophic. In the presence of oxygen, phosphorus settles to the lake bottom and is unavailable for aquatic plant growth. However, if bottom-water oxygen is depleted, phosphorus is released from the sediments and may be available to promote aquatic plant growth. In some lakes, the release of phosphorus from the bottom sediments is the primary source of phosphorus loading (or input) to the lake. By reducing the amount of phosphorus in a lake, it may be possible to control the amount of aquatic plant growth. In general, lakes with a phosphorus concentration greater than 20 parts per billion are able to support abundant plant growth and are classified as nutrient-enriched or eutrophic.

Chlorophyll-*a*

Chlorophyll-*a* is a pigment that imparts the green color to plants and algae. A rough estimate of the quantity of algae present in lake water can be made by measuring the amount of chlorophyll-*a* in the water column. A chlorophyll-*a* concentration greater than 6 parts per billion is considered characteristic of a eutrophic condition.

Secchi Transparency

A Secchi disk is often used to estimate water clarity. The measurement is made by fastening a round, black and white, 8-inch disk to a calibrated line. The disk is lowered over the deepest point of the lake until it is no longer visible, and the depth is noted. The disk is then raised until it reappears. The average between these two depths is the Secchi transparency. Generally, it has been found that aquatic plants can grow at a depth of approximately twice the Secchi transparency measurement. In eutrophic lakes, water clarity is often reduced by algae growth in the water column, and Secchi disk readings of 7.5 feet or less are common.



Secchi disk

Lake Classification Criteria

Ordinarily, as phosphorus inputs to a lake increase, the amount of algae will also increase. Thus, chlorophyll-*a* levels will increase and transparency decreases. A summary of lake classification criteria developed by the Michigan Department of Natural Resources is shown in the table below.

LAKE CLASSIFICATION CRITERIA

Lake Classification	Total Phosphorus (µg/L)[1]	Chlorophyll-*a* (µg/L)[1]	Secchi Transparency (feet)
Oligotrophic	Less than 10	Less than 2.2	Greater than 15.0
Mesotrophic	10 to 20	2.2 to 6.0	7.5 to 15.0
Eutrophic	Greater than 20	Greater than 6.0	Less than 7.5

Recent sampling of 364 lakes across Michigan indicates that, of the lakes sampled, less than about 20% of lakes are oligotrophic, more than 50% are mesotrophic, and about 30% are eutrophic.[2]

pH and TOTAL ALKALINITY

pH is a measure of the amount of acid or base in the water. The pH scale ranges from 0 (acidic) to 14 (alkaline or basic) with neutrality at 7. The pH of most lakes in the Upper Midwest ranges from 6.5 to 9.0.[3] In addition, according to MDEQ[4]:

> While there are natural variations in pH, many pH variations are due to human influences. Fossil fuel combustion products, especially automobile and coal-fired power plant emissions, contain nitrogen oxides and sulfur dioxide, which are converted to nitric acid and sulfuric acid in the atmosphere. When these acids combine with moisture in the atmosphere, they fall to earth as acid rain or acid snow. In some parts of the United States, especially the Northeast, acid rain has resulted in lakes and streams becoming acidic, resulting in conditions which are harmful to aquatic life. The problems associated with acid rain are lessened if limestone is present, since it is alkaline and neutralizes the acidity of the water.
>
> Most aquatic plants and animals are adapted to a specific pH range, and natural populations may be harmed by water that is too acidic or alkaline. Immature stages of aquatic insects and young fish are extremely sensitive to pH values below 5. Even microorganisms which live in the bottom sediment and decompose organic debris cannot live in conditions which are too acidic. In very acidic waters, metals which are normally bound to organic matter and sediment are released into the water. Many of these metals can be toxic to fish and humans. Below a pH of about 4.5, all fish die.

The Michigan Water Quality Standard (Part 4 of Act 451) states that pH shall be maintained within the range of 6.5 to 9.0 in all waters of the state.

Alkalinity, also known as acid-neutralizing capacity or ANC, is the measure of the pH-buffering capacity of water in that it is the quantitative capacity of water to neutralize an acid. pH and alkalinity are closely linked and are greatly impacted by the geology and soil types that underlie a lake and its watershed. According to MDEQ[3]:

> Michigan's dominant limestone geology in the Lower Peninsula and the eastern Upper Peninsula contributes to the vast majority of Michigan lakes being carbonate-bicarbonate dominant [which increases alkalinity and moderates pH] and lakes in the western Upper Peninsula having lower alkalinity and thus lesser buffering capacity.

The alkalinity of most lakes in the Upper Midwest is within the range of 23 to 148 milligrams per liter, or parts per million, as calcium carbonate (MDEQ[3]).

pH AND ALKALINITY OF UPPER MIDWEST LAKES

Measurement	Low	Moderate	High
pH (in standard units)	Less than 6.5	6.5 to 9.0	Greater than 9.0
Total Alkalinity or ANC (in mg/L as $CaCO_3$)[5]	Less than 23	23 to 148	Greater than 148

Variability and Trends

Often there is a desire to evaluate trends in water quality. However, this can be a difficult task. It is important to realize that there are a number of factors that influence water quality. Weather, for example, can have a strong influence on water quality conditions. A lake sampled immediately after several days of strong winds and rain may appear much different than if sampled after several days of calm, sunny weather. There can be significant natural variability in lake water quality daily, seasonally and year-to-year. Because of this natural variability, it can be very difficult to detect subtle changes or trends that may occur in water quality over time. In fact, it may take many years of regular sampling to detect a statistically significant trend in water quality.

References

[1] µg/L = micrograms per liter = parts ber billion.

[2]Fuller L.M. and R.J. Minnerick. 2008. State and regional water-quality characteristics and trophic conditions of Michigan's inland lakes, 2001-2005. U.S. Department of the Interior, U.S. Geological Survey. Scientific Investigations Report 2008-5188.

[3]Michigan Department of Environmental Quality. 2012. Michigan National Lakes Assessment Project 2007. MI/DEQ/WRD-12/006.

[4]Michigan Department of Environmental Quality. 2013. Water Quality Parameters: pH, accessed February 26, 2014, http://www.michigan.gov/documents/deq/wb-npdes-pH_247233_7.pdf.

[5]mg/L as CaCO3 = milligrams per liter as calcium carbonate.

About the Authors:
Tony Groves, Pam Tyning, and Paul Hausler have over 80 years of combined experience working as lake management consultants with Progressive AE in Grand Rapids, Michigan. Tony, Pam, and Paul created MichiganLakeInfo.com, a website for those interested in Michigan's inland lakes. On the site you can find this article and information on topics such as lake water quality, lake and watershed management, aquatic biology, emerging issues, invasive species and more.

State of Michigan's

Status and Strategy for Zebra and Quagga Mussel Management

Scope

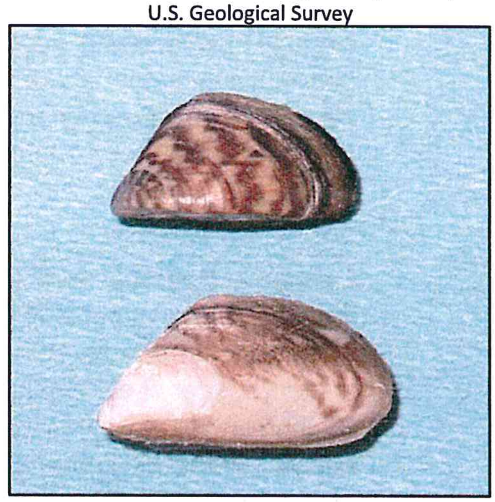
The invasive zebra mussel (*Dreissena polymorpha*) and quagga mussel (*Dreissena rostriformis bugensis*) have severely affected the waters of the State of Michigan. The goals of this document are to:

- Summarize the current level of understanding on the biology and ecology of the zebra and quagga mussel.
- Summarize the current management options for the zebra and quagga mussel in Michigan.
- Identify possible future directions of zebra and quagga mussel management in Michigan.

Biology and Ecology

I. Identification

Dreissena polymorpha (top)
Dreissena rostriformis bugensis (bottom)
U.S. Geological Survey



Zebra and quagga mussels are both non-native freshwater mollusks found in all of the Great Lakes. The zebra mussel's striped shell pattern distinguish it from the quagga mussel. Quagga mussels have a rounded carina, or angle, between the ventral and dorsal surfaces and a convex ventral side (May and Marsden 1992). Contrarily, zebra mussels have a definite carina between the ventral and dorsal surfaces that are flattened on the ventral side (Claudi and Mackie 1994). If you placed both mussels on their ventral side, the quagga would topple over and the zebra would not (Claudi and Mackie 1994). Quaggas are generally rounder in shape and have a small byssal groove on the ventral side near the hinge. Zebra mussels are generally triangular and have a larger groove in the middle of the ventral side (Claudi and Mackie 1994, Marsden et al. 1996). Quagga mussels can develop a variety of shell patterns including black, cream, or white bands, while zebra mussels have dark striped shells or light shells with no stripes (Benson et al. 2014a, Benson et al. 2014b). In Lake Erie, a distinct quagga mussel morph can be found that is completely white (Marsden et al. 1996). Quagga mussels usually have dark concentric rings on their shell and lack color near the hinge. Reaching up to 50mm, Zebra mussels on average can be larger than quagga mussels that reach up to 40mm (Benson et al. 2014a, Benson et al. 2014b).

II. Life History

Zebra and quagga mussels are prolific breeders, reproducing dioeciously with external fertilization. A mature female can produce up to one million eggs per season. After fertilization, pelagic, microscopic larvae known as veligers develop within a few days and soon acquire minute bivalve shells. The veligers drift with water currents for three to four weeks before securing to a substrate via byssal threads; during this drift they feed with hair-like cilia (Richerson 2013). During the transition from planktonic veliger to juvenile, the mussels may experience a mortality rate of 99% due to settlement onto unsuitable substrates (Bially and MacIssac 2000, Richerson 2013, Benson 2014b).

Zebra mussels' oogenesis occurs in autumn. The eggs are released and fertilized in the spring. However, in thermally polluted areas, reproduction can occur continuously. Males become reproductively mature within the first year (or when they reach 8-9mm shell lengths), while females usually reproduce in their second year. Optimal temperatures for spawning range from 14 to 16°C while the optimal temperature for larval development is between 20 and 22°C (Benson et al. 2014b). If the larvae survive and successfully attach to a substrate, they stay attached and morph into the juvenile stage, where they begin to filter feed and grow rapidly (Hart et al. 2000). Veligers do not discriminate between substrates, whereas juveniles prefer hard, rocky substrates and vegetation. Zebra mussels grow at a rate of 1.5 to 2 cm per year and have a typical life span of 3 to 9 years (Benson et al. 2014b).

III. Diet

Quagga and zebra mussel are filter feeders. With both an inhalant and exhalant siphon, the mussels are capable of filtering around one or more liters of water per day. Phytoplankton, zooplankton, algae, and even their own veligers are desired particulate matter (Snyder et al. 1997). Particle-free water is discharged from the exhalant siphon (Richerson 2013). Undesired matter, such as metals, certain algae and bacteria are bound with mucus, known as pseudofeces and expelled through the inhalant siphon. Internal mechanisms, use chemical cues to recognize which materials to expel. Pseudofeces production is a mechanism that helps mussels deal with overabundance of food and helps them reject unpalatable algae and bacteria (Benson et al. 2014b).

Zebra and quagga mussels primarily consume phytoplankton, however other suspended material is filtered from the water column such as bacteria, protozoans, other micro zooplankton and silt (Benson et al. 2014b). While in their larval stage, zebra mussels feed on bacteria while adults prefer larger particles such as algae and zooplankton between 15 and 400 microns (GISD 2009). The zebra mussel does reject cyanobacteria. The feeding rate is determined by the clearance rate (the percentage of algal biomass removed from the water column over time), the biomass of cleared algae, and the amount of feces and pseudofeces production. Zebra mussel size, phytoplankton species, and regional population differences can affect feeding rate (Benson et al. 2014b).

IV. Habitat

Both zebra and quagga mussels inhabit freshwater rivers, lakes, and reservoirs. Zebra mussels attach to any stable substrate present in the water column including artificial surfaces such as pipes, boats, docks, etc., along with crayfish, unionid clams, macrophytes, and even each other in order to form dense colonies. The long-term stability of the substrates affects the density and age distributions found on those substrates. Extensive siltation, certain sessile benthic macroinvertebrates, microalgae, and fluctuating water levels expose mussels to desiccation, which make a substrate less suitable for long-term colonization. These factors also affect spatial patterns of pelagic densities and benthic adult dispersions (Benson et al. 2014b).

Native to the Black, Caspian, and Azov Seas, North American zebra mussel populations have adapted to warmer temperatures. Shell growth can occur at temperatures as low as 3°C with the typical low range at 6 to 8°C. Eggs can be released at 13°C, but the release rate increases at temperatures over 17°C. Zebra mussels can persist in temperatures up to 30°C with an optimal range of 20 to 25°C. The zebra mussel can tolerate anaerobic conditions for a short time, but cannot persist in a hypoxic condition. The oxygen demands of the zebra mussel are similar to that of other freshwater bivalves. Zebra mussels are typically found in hypolimnetic and epilimnetic zones where oxygen levels are 0.1-11.2 mg/L and 4.2-13.3mg/L respectively (Benson et al. 2014b).

North American zebra mussels can only tolerate slight salinity with an upper limit of 4%. Also, North American populations require 10 mg Ca2+/L to start shell growth and 25 mg Ca2+/L to maintain that growth. Optimal larval survival occurs at a pH of 8.4, while optimal adult growth occurs at a pH ranging from 7.4 to 8.0 (Benson et al. 2014b).

Native to the Dneiper River drainage of Ukraine and Ponto-Caspian Sea, quagga mussels tolerate slight levels of salinity with an upper limit of 5%. Water temperatures reaching 28°C cause increased mortality with lethal temperatures between 32 and 35°C. Wave action prevents the quagga mussel from establishing near shore and temperature determines the water depth at which mussels are found. For example, the maximum density of quagga mussels in Lake Michigan is found at 31-50 meters deep (Benson et al. 2014a). Zebra and quagga mussels diverge in their spatial distributions; both species inhabit warm, eutrophic, shallow water, but the quagga mussel range also extends to deep, oligotrophic, cold water (MacIsaac 1994).

V. Effects from Zebra and Quagga mussels

One major impact caused by zebra mussels is biofouling. They colonize water supply pipes of hydroelectrical and nuclear power plants, public water supply plants, and industrial facilities. Zebra mussels constrict water flow through pipes and, therefore, reduce the intake in heat exchangers, condensers, firefighting equipment, and air conditioning and cooling systems. Navigational and recreational boating is also affected. Attached mussels increase boat drag and mussels in engine cooling systems can cause overheating and damage. Fishing gear can be fouled, navigational buoys can be sunk under the weight of attached mussels, and dock pilings deteriorate faster when encrusted with mussels. Continued attachment of zebra mussel can cause corrosion of steel and concrete, affecting the structural integrity (Benson et al. 2014b).

Zebra mussels also disrupt the ecosystems they invade. Zebra mussels may shift lakes from a turbid, phytoplankton-dominated state to a clear and macrophyte-dominated state (Scheffer et al. 1993). In the Great Lakes, large populations of zebra mussels have significantly reduced the biomass of phytoplankton. In Lake Erie, diatom abundance declined by 82 to 91% in the first years of invasion (Holland 1993). Zooplankton abundance also drops dramatically with zebra mussel invasion; this is the result of direct predation on microzooplankton and the reduction of available zooplankton food sources. In addition, zebra mussel invasion reduces chlorophyll-a levels and may promote macrophyte communities. By removing particles from the water column, the mussels increase water transparency that affects plant growth and species dominance; which in turn impacts fish habitats.

Fish spawning can be affected by the dense colonization of hard substrates and foraging could also be compromised by colonization on soft substrates. Increased water transparency may also cause temperatures to rise and thermoclines to become deeper. Inland lakes with zebra mussels have been found to have lower dissolved organic carbon (DOC) concentrations and this may be due to phytoplankton consumption by mussels (Raikow 2002). Macrophyte growth could compensate these lower concentrations, but there may be a lag period during which UV-B light is able to penetrate deeper into the water column. Zebra mussels are also able to assimilate DOC (Roditi et al. 2000). Zebra mussels are more efficient at filtering small particles than unionids and Asiatic clams. It is speculated that the biodeposition of feces and pseudofeces or the increased physical habitat complexity of a mussel colony might cause observed increases in benthic macroinvertebrate populations (Stewart and Haynes 1994).

It is possible for concentrations of pollutants in zebra mussel feces and pseudofeces to transfer to other trophic levels (Bruner et al. 1994). Furthermore, reductions in zooplankton biomass may cause increased competition, decreased survival, and decreased biomass of planktivorous fish. Alternatively, benthic feeding fish may benefit from the mussel invasion because the mussels may cause a shift from pelagically to benthically-based food webs in inland lakes. The depletion of microzooplankton in particular may have a greater impact on larval fish populations than on older fish. Zebra mussels can also extirpate native unionid populations. Zebra mussels are not only in competition with native unionids for food, but they also attach to native unionids resulting in restricted valve operations, smothered siphons, and shell deformities. Zebra mussels impair native unionids' movements and also deposit their metabolic waste onto the native species. Unionids have been extirpated from Lake St. Clair and drastically reduced in Lake Erie.

The quagga mussel also removes significant amounts of phytoplankton and other particles from the water column. Like zebra mussels, quagga mussels decrease the abundance of zooplankton, reduce chlorophyll-a concentrations, increase water transparency, and accumulate pseudofeces, which can foul the environment (Claxton et al. 1998). As the mussel waste decomposes, oxygen is consumed, pH is lowered, and toxic byproducts are produced. Biomagnification of organic pollutants can occur as pseudofeces is passed up the food chain (Snyder et al. 1997).

Current status and distribution in Michigan

The introduction of zebra and quagga mussels into the Great Lakes appears to be the result of discharged transoceanic ship ballast water contaminated with mussels (Richerson 2013). Dreissenid species are prolific breeders that can adapt rapidly and this contributed to both species swift spread throughout the country (Mills et al. 1996, Figure 1). By 1990, zebra mussels were found in all of the Great Lakes (Benson et al. 2014a). The establishment of quagga mussels in the Great Lakes was first observed in 1989 and sightings in all the Great Lakes were confirmed by 2005 (Benson to al. 2014b). Zebra mussels have been reported in Michigan 1,217 times (70 different counties) while quagga mussels have been reported 171 times (15 different counties) to the Midwest Invasive Species Information Network (MISIN, accessed May 22, 2014)(Figure 2). According to the United States Geological Survey (USGS, accessed July 29, 2014), quagga mussels have also been found in Lake St. Clair, Fortune Pond, and Little Black Lake (Figure 2).

Management of Zebra and Quagga mussels

Zebra and quagga mussels have the ability to disperse during all life stages. Passive drift of pelagic larval veligers allows downstream invasion. Yearlings can detach and drift for short distances, and adults routinely attach to boat hulls and floating objects. Transporting recreational boats from the Great Lakes to inland lakes and between inland lakes also allows for the dispersal of mussels (Richerson 2013). The success of overland transport of mussels depends on their ability to tolerate periods of desiccation. Adult zebra and quagga mussels can survive 3-5 days of aerial exposure (Ricciardi et al. 1995). Unlike endemic bivalves, zebra mussels have byssal adult stages, which has also aided in its successful spread throughout the United States (Benson et al. 2014). Many management options have been explored for combating the spread of zebra and quagga mussel populations. Specific plans are usually created by lake managers and are based off of existing response methods, listed below.

I. Monitoring

Note: Monitoring information is based off of California Sea Grant's Early Detection Monitoring Manual for Quagga and Zebra Mussels (Culver et al. 2009). The manual is available for download or in print from California Sea Grant's web page.

Effective monitoring techniques provide opportunities to detect the presence of zebra and quagga mussels in advance of population establishment, when eradication becomes cost-intensive and nearly impossible. Most monitoring is carried out during the summer months since this is when adult mussel populations are highest and easiest to identify. However, when targeting larval stages of mussels, monitoring should be conducted during and just after spawning seasons. Visual identification of mussels and veliger sampling kits are the most common methods for monitoring mussels and rely heavily on volunteer work. Citizens should be encouraged to closely examine docks and other water borne hardware upon removal, as these structures often attract zebra mussels. When monitoring, it is important to identify which life stages are being targeted. Factors such as water temperature, pH, and calcium concentrations influence spawning and should be taken into account especially when doing veliger sampling. Potential invasion corridors determine which life stages should be monitored.

If recreational users are suspected to be transferring mussels, adult and juveniles should be searched for. If water from other sources (live well, industrial exhaust pipes, water discharge) is suspected, veliger sampling should be conducted. Frequency of monitoring will depend on the targeted life stage. When monitoring for veligers, several sampling efforts should be conducted around spawning. A regular schedule should be created based on mussel biology. When sampling for adults and juveniles, monitoring can be more rigorous during the summer and scaled back or halted over winter.

Site selection depends on the amount of public use, proximity to high-risk areas, environmental conditions (temperature, pH, calcium concentration, current, ect.), and potential ecological/economic impacts. High risk areas include water inflows from external sources, high traffic boat access points, and areas with dense potential substrate such as docks, ramps, pipe, and floating or sunken debris. Other than veliger sampling, most monitoring can be carried out with basic equipment such as collection bags and tags, a utility knife, waders or a wetsuit. Deeper areas may require SCUBA equipment. Veliger sampling kits are usually ~ $150 per kit. When monitoring for new populations, veliger sampling or visual identification of mussels are the most common methods.

I. Prevention

Michigan has established Integrated Pest Management (IPM) strategies to prevent the spread and dispersion of aquatic invasive species. These strategies mainly focus on prevention, the initial stage of management and control. Prevention for mussels includes checking for and removing any foreign material, mud or vegetation on boating equipment such as hulls, propellers, trailers, anchors, etc.. In addition, any compartments where water may be stored should be flushed with hot water; the water needs to be 43.3°C to kill veligers and 60°C to kill adult mussels. Compartments that should be flushed may include engine cooling systems, anchor lockers, live wells, bilges, trailer frames, safety light housings, and boat decking. If hot water cannot be accessed, tap water or a 10% bleach solution can be used; however, the boat should be left to dry for five days before entering a water body. If, upon leaving infested waters, mussels persist or algae is present on the trailer or any part of the watercraft, the equipment should be allowed to dry for five days or more before moving to non-infested waters. If any 'gritty' feeling persists on equipment, it is most likely young mussels. The gritty equipment should be scrubbed and rinsed with hot water before use in another lake.

Any adult mussels scraped and removed from the watercraft or trailer should be disposed of properly in a garbage bin. If bait was used in an infested area, it should not be used in another body of water. Bait buckets should be emptied on land to prevent the spread of microscopic veligers into lakes or streams (Hart et al. 2000). Pre-chlorination systems provide extra protection and should be used by the management/monitoring staff to prevent mussels from attaching to equipment.

Management/Control

Note: The majority of this management section strongly applies to industry application and was taken from Spencer and Getsinger (2002) which was based on information pulled from Boelman et al. (1997). For more information and specifics, refer to these mentioned sources.

a. Physical
   i. *Mechanical Removal and Filtration, Repellent Materials and Coatings*

   Mechanical raking/scraping of mussels off surfaces is effective, but less cost-efficient than preventative measures. Automated systems may decrease total cost over time. Manual SCUBA removal has also proven to be an effective method when invasion is detected early enough (Wimbush et al. 2009). Pigging systems by forcing plugs though mussel-infested lines can scrape away the mussels from pipe walls, but drawbacks, including the unavailability of the pipeline during pigging and mussel debris disposal, exist. To overcome pigging problems new and existing facilities could construct secondary systems to maintain uninterrupted service during cleaning. Conventional water screens, in-line debris filters, ultrafiltration, and traveling screens, many of which are now becoming self-cleaning, can be effective in blocking adult mussels and shells, but many still allow passage of veligers.

   For new facilities, choosing antifouling construction materials for structures and pipes, such as copper and galvanized iron, could minimize the mussels' impact. Specialized coatings can also be effective in controlling mussels. Antifouling coatings (cuprous oxide), leach toxins, foul-release coatings (like nontoxic, silicone-based paint) present slippery surfaces, and thermal-spray coatings release metal ions into the water (Spencer and Getsinger 2002). However, these toxic coatings typically only last for 2-5 years and reapplication will be required to maintain protection.

ii. High Pressure Water Jet Cleaning, High-Velocity Flows, Carbon Dioxide Pellet Blasting

Water jets with pressures of 3000 psi are recommended to remove zebra mussels (Claudi and Mackie 1994). Abrasives added to the water stream make this process more effective. The velocities of pipe flow could be increased periodically to help prevent blockage from mussels. Mussels avoid high-velocity flows and juveniles tend to settle in areas with flow rates less than 1.5m/sec (Spencer and Getsinger 2002).

Carbon dioxide pellet blasting is similar to sand blasting, but is preferred because sand only removes the zebra mussel's outer shell. Carbon dioxide pellet blasting removes more organic material and is less likely to damage surfaces (Spencer and Getsinger 2002).

iii. Freezing or Desiccation, Thermal Treatment

Mussels can be eradicated by exposing them to freezing or high temperatures. Clustered mussels are more tolerant to reduced air temperatures than individual mussels – 48 hours at -1.5°C or 2 hours at -10°C will result in 100% mortality of clumps while just 15 hours at -1.5°C or under 2 hours at -10°C will result in 100% mortality of individuals. Mussels can also be controlled during the summer months at extended exposure times. Increases in humidity negatively impacts mortality rates. At high temperatures (25°C) and low humidity levels (5%), 100% mortality can be achieved; however, if humidity increases to high levels (95%), 100% mortality is expected after about 5 days. When heated water is used, a temperature above 32.5°C for more than five hours is lethal (Spencer and Getsinger 2002). For short-term exposure temperatures of >80^0 C for 5 seconds or at least >60^0 C for 10 seconds is required. Current 60^0 C treatments may not be 100% effective if applied for less than 10 seconds (Morse, 2009).

When considering freezing or thermal treatments, effective conditions will have some variation because the temperature tolerances of mussels is directly correlated to acclimation temperatures and immersion times. Smaller mussels also have greater thermal tolerances than larger mussels. Thermal treatments are cost-effective and efficient at zebra mussel control. Heat treatment is generally regarded as more environmentally safe than chemical treatment, but restrictions on the discharge of heated water need to be considered (Spencer and Getsinger 2002).

iv. Reduced Pressure, Pulse Acoustics

When flow consists of raw untreated water, pressures of 14 to 15 psi in air or underwater will suffocate mussels due to reduced dissolved oxygen levels. Sound energy is also being developed as a means to control mussel populations; approaches in sound energy include cavitation, sound treatment, and vibration. Vibration amplitude needed for effectiveness increases with increasing frequencies (Spencer and Getsinger 2002).

v. Electric Fields, Low-Frequency Electromagnetism, Ultraviolet (UV) Light

Electricity has been shown to affect mussel behavior. Direct and alternating currents have been shown to stun and affect the settlement of mussels. Extremely low-frequency electromagnetism exposure can also inhibit mussel establishment given its interference with the mussels' ability to acquire calcium. Low-frequency electromagnetism causes mussels to be unable to grow and develop, reproduce, and preform metabolic functions. UV lamps are another alternative that can be installed in intake bays or pipes to induce mortality of mussels. UV treatment also has additional water quality benefits and would not require discharge permitting. However, water with high suspended loads or turbidity reduces the effects of UV radiation (Spencer and Getsinger 2002).

b. Chemical

i. Oxygen Deprivation

Oxygen scavenging chemicals such as sodium-meta-bisulfite and hydrogen sulfide gas can be added to water to deprive mussels of dissolved oxygen. Mussels can tolerate oxygen deprivation for 6 to 14 days depending on environmental temperatures. However, oxygen deprivation may increase corrosion (Spencer and Getsinger 2002). Benthic mats can also be used as a physical method to separate mussels from their oxygen supply. If placed early enough, these mats can also decrease veliger distribution.

ii. Chemical Molluscicides

Many chemicals kill mussels, but the suitability of the chemical depends on many factors including cost, practicality, byproducts, residual concentrations, and water quality impacts. Moderately successful molluscicides include chloramines, chlorine dioxide, ozone, hydrogen peroxide, potassium permanganate, pH adjustment, and inorganic salts (GISD 2009). Chlorination is the most widely used. It has economic

feasibility, is easy to apply, and is highly effective. However, chlorination forms carcinogenic byproducts. Ozone can also be used as a control method and actually outcompetes chlorine in terms of contact time at comparable residual levels. Ozone treatments result in low pipe residuals and no downstream environmental impacts, but are expensive to purchase, maintain, and difficult to sustain treatment concentrations that result in 100% mortality of established adult populations.
Another oxidizing chemical used for antifouling purposes is bromine (Spencer and Getsinger 2002). The effects and concentrations of bromine are very similar to chlorine. Commonly used oxidizing molluscicides can be found in Table 1 along with nonoxidizing and metallic molluscicides (Spencer and Getsinger 2002). For more information on molluscicides effectiveness and impacts on nontarget species, Waller et al. (1993), Claudie and Mackie (1994), EPRI (1993), and McMahon et al. (1994) can be referenced (Table 2). Table 3 provides toxicology data on nontarget species. A 3% solution of Sparquat 256[1] will kill quagga veligers and mussels after 10 minutes of exposure (Britton and Dingman 2011) and would likely be effective against zebra mussels. Application in open water environments would kill pelagic veligers as well as benthic juveniles greatly increasing management efficiency, but further testing is needed before large-scale application can begin.

To overcome rejection and valve-closing responses seen by the mussels after exposure to toxic chemicals, edible microencapsulation of toxins have been used. Potassium chloride, the active ingredient, is not lethal to most organisms at low levels beside freshwater bivalves. Endocannabinoids, anandamide, and nine other functionally similar compounds have also been tested for their non-toxic interference in mussel byssal attachment (Angarano 2009, GISD 2009).

[1]Sparquat 256 is not included in the toxicology tables, additional information is available at: http://www.fs.usda.gov/Internet/FSE_DOCUMENTS/fsbdev3_014795.pdf

c. Biological

i. Selectively Toxic Microbes

Certain soil and water microbes could be selectively lethal to *Dreissena* when applied at artificially high water densities. One *Pseudomonas fluorescens* bacterial strain CL0145A has been shown to be selectively lethal to *Dreissena*; research is currently being conducted to test for its effectiveness (GISD 2010). Zequanox[2], a toxin using *P. fluorescens,* has recently been approved for open water use by the EPA, and has shown potential for containment. Zequanox is classified as a reduced-risk aquatic biopesticide and can be applied in a matter of hours with basic equipment. Unlike with traditional chemical treatments, mussels do not close in the presence of Zequanox allowing for greater exposure.

Unfortunately, Zequanox is not a silver bullet. Although it is effective against all life stages of both zebra and quagga mussels, it is not 100% (> 90%) effective and the high cost makes large scale application, such as whole lake treatments, unreasonable at this point. However, for industrial applications, Zequanox could provide adequate protection without the need for expensive retrofitting.

Early research examining the detrimental effects of algal blooms on veliger and adult mussel viability is also being conducted in Donna Kashian's lab at Wayne State University.

[2]More information on Zequanox is available on Marrone Bio's website: https://marronebioinnovations.com/molluscicide/zequanox/

ii. Natural Enemies

The high recruitment rate of *Dreissena* populations makes it difficult for natural enemies to control them. Even in their native water bodies, natural predators don't seem to keep the mussel densities low enough to avoid ecological or industrial problems (Spencer and Getsinger 2002). In coastal wetlands, large-molluscivores, including common carp, freshwater drum, and channel catfish, can limit mussel numbers. Other known predators include roach, eel, sturgeon, diving ducks, crayfish, and muskrats (GISD 2009). The sponge *Eunapius fragilis* has been observed colonizing and killing zebra mussels in the southern basin of Lake Michigan (Early and Glonek 1999). Sponge colonization forces the mussels to close, resulting in energy deprivation and eventually death. Although promising mussel control by *Eunapius fragilis* will require more research. The effects and viability of *Eunapius fragilis* in northern waters is unknown and must be evaluated before moving forward.

A combination of treatments will often produce the best results; specific combinations should be tailored to each location, as environment and biological factors are often site specific. Fortunately, many zebra mussel treatments work on quagga mussels and quagga mussel treatments on zebra mussels allowing for simultaneous treatments in most cases. Combing and coordinating efforts with other states within the Great Lakes Basin should be considered as well. A cohesive, multistate effort has potential to achieve better management than any one state alone.

Future Directions for Michigan and the Zebra and Quagga Mussel Management

Once established, it becomes very difficult to eliminate zebra and quagga mussels. Therefore, preventing the spread of zebra and quagga mussels needs to be the goal of management efforts. Since recreational and commercial vessels are the most common modes of transportation, these pathways need to be closely examined. More stringent regulations and more severe legal penalties may encourage recreational users to make cleaning their boats a priority. Posting signs at public assess sites along infested waters would also remind recreational users that they are using an infested water body and to be cautious about taking invasive species with them when they leave. Campaigns, such as Stop Aquatic Hitchhikers!, already work to raise awareness and change behaviors; the simple message - clean, drain, dry, everywhere, every time – can help contain mussels and many other invasive species. Education can also help lake users and associations identify and report zebra and quagga mussels. As for existing populations, managing their spread is the best course of action. Although populations may be reduced, or in the case of new small scale invasions eliminated, it is unlikely that current management techniques will be able to permanently remove zebra and quagga mussels from all infested water bodies.

It is imperative that government agencies reach out to private citizens and lake associations to develop an easily accessible reporting system; government agencies cannot adequately monitor Michigan's waters alone and volunteers are the most cost effective alternative. Industrial solutions are adequate for keeping mussel populations and fouling in check if used correctly, but constant monitoring and treatment results in high costs.

Management and development costs vary significantly based on the level of infestation and size of the affected area. For small scale infestations, plan development costs as little as $10,000, but for infestations similar to those in Michigan, development of a zebra/quagga mussel management plan will likely cost closer to $100,000 and implementation of the plan will likely be in the millions. To compare, costs of development and specific components of plans in other states can be found in the "Quagga-Zebra Mussel Action Plan for Western U.S. Waters (QZAP). These plans may be helpful when developing budgets for zebra/quagga mussel management here in Michigan. A full copy of the QZAP plan is publicly available at online at: (http://anstaskforce.gov /QZAP/QZAP_FINAL_Feb2010.pdf). Although the full details of the plan are beyond the scope of this document, estimated costs for zebra/quagga mussel management in the Western United States via QZAP is $31,140,000 annually with each approved QZAP state receiving $967,742 per year and QZAP states still developing their plans receiving $60,000 per year.

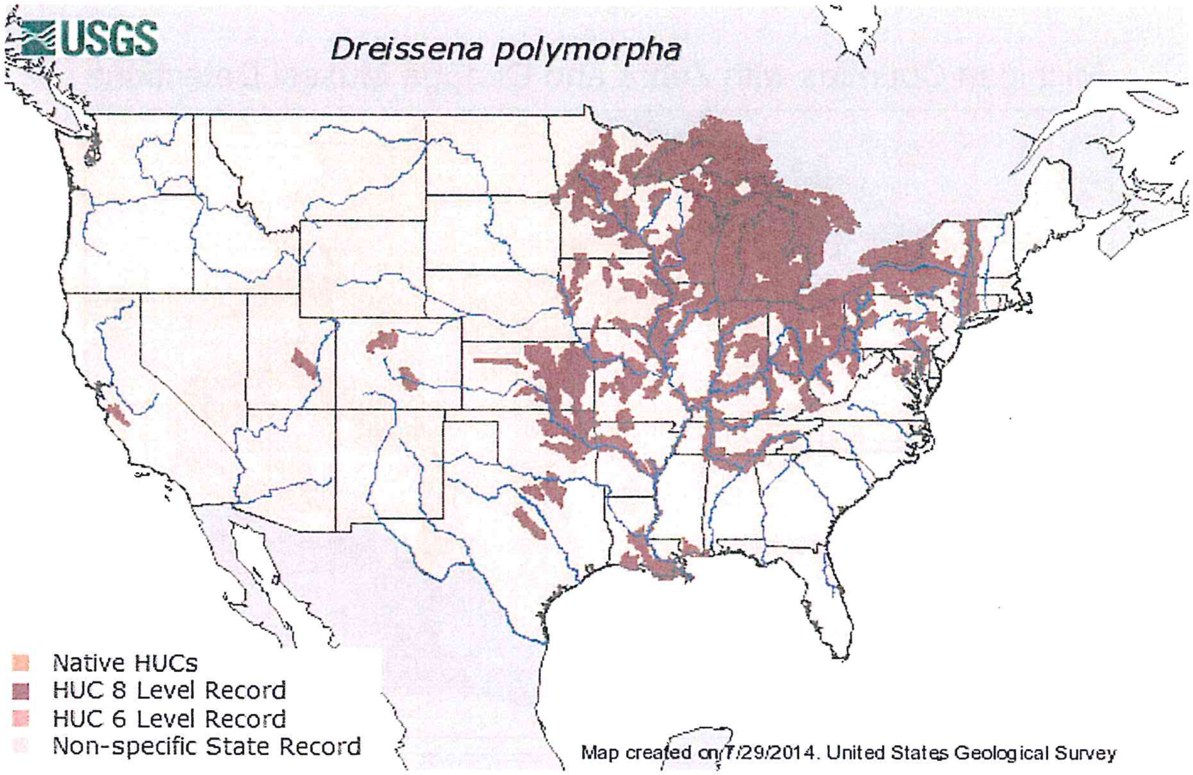


Figure 1. Distribution of zebra mussels in the United States (Benson et al. 2014a). Accessed July 29, 2014.

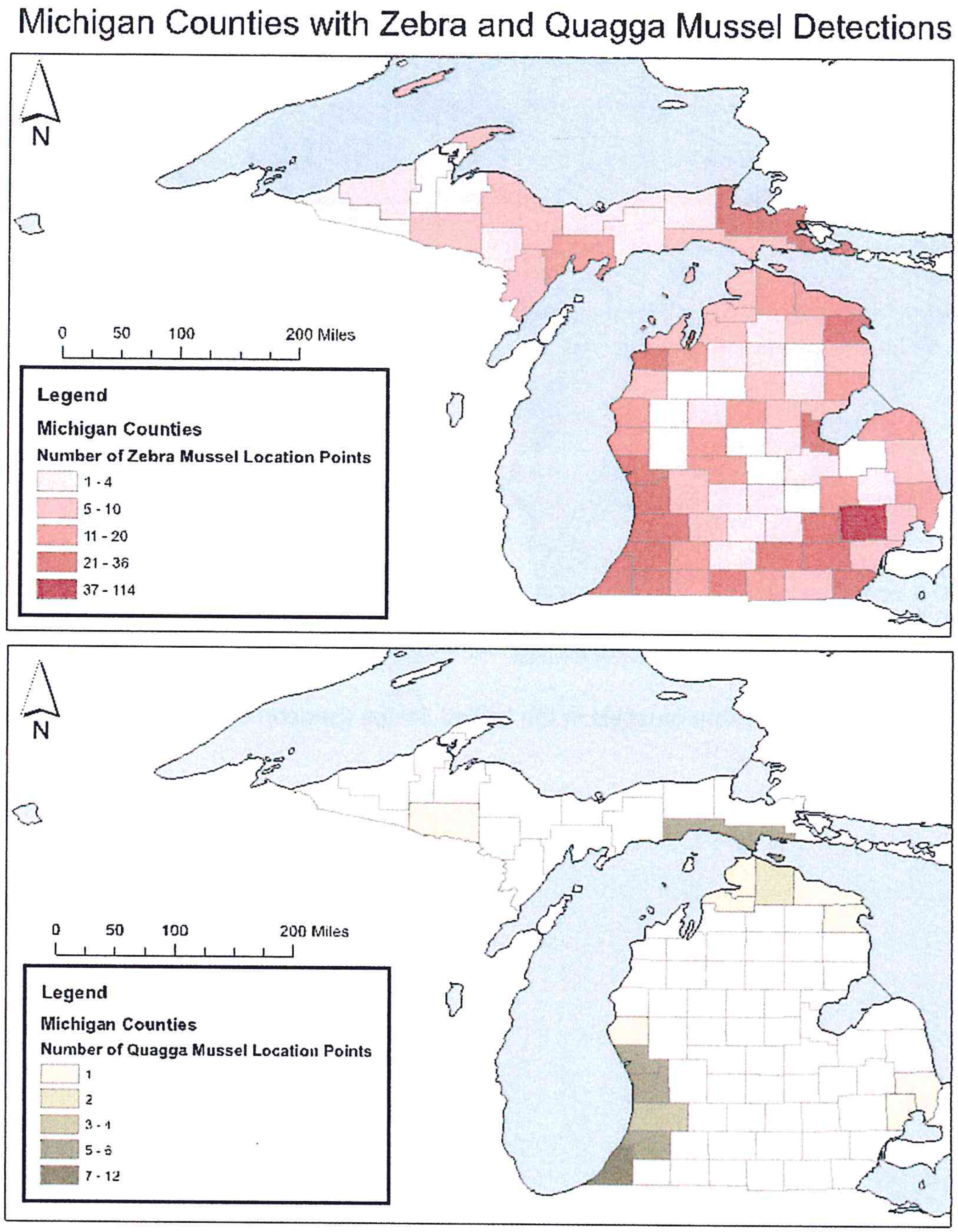


Figure 2. Number of unique coordinate location points within Michigan counties at which zebra and quagga mussels were detected. This data is according to the United States Geological Survey (USGS), Midwest Invasive Species Information Network (MISIN, accessed May 22, 2014) and Biodiversity Information Serving Our Nation (BISON, accessed June 13, 2014) databases.

Table 1. An overview of chemical control methods and their effectiveness (Spencer and Getsinger 2002). Accessed March 31, 2014. Online at
The link provided is no longer valid. This online document was revised 11/6/2017.

TREATMENT METHOD	APPLICATION OF MOLLUSCICIDE	EFFECTIVENESS
	Oxidizing Molluscicides	
Chlorination (adults)	0.5 ppm for 7 days 0.3 ppm for 14 - 21 days	75% kill >95% kill
Chlorination (adults)	2-ppm continuous	90% kill
Chlorine dioxide	0.5 ppm for 24 hours	100% veliger kill
Chloramine	1.2 ppm for 24 hours	100% veliger kill
Ozone	1.5 ppm continuous	Prevents settlement
Cyanuric acid	2,000 ppm for 17 days	50% kill
	Nonoxidizing Molluscicides	
Dichloro-2' nitro-4' salicylanilide	0.05 ppm for 24 hours 0.1 ppm for 24 hours	70% kill 100% kill
N-triphenyl-methylmorpholine	0.5 ppm for 24 hours 0.9 ppm for 24 hours	70% kill 100% kill
Poly [oxyethylene-(dimethyliminio)-ethylene (dimethyliminio) ethylene dichloride]	0.3 ppm for 826 hour 1.2 ppm for 313 hours 4.8 ppm for 197 hours	100% kill 100% kill 100% kill
2 - (thiocyanomethylthio) benzothiazole	0.15 ppm for 758 hours 0.6 ppm for 313 hours 1.2 ppm for 260 hours	100% kill 100% kill 100% kill
Dimethylbenzyl ammonium chloride and Dodecylguanidine hydrochloride	1.95 ppm for 12 hours at 11°C 1.95 ppm for 14 hours at 14°C 1.95 ppm for 6 hours at 20°C 1.95 ppm for 14 hours at 20°C	100% kill after 48 hours 100% kill after 48 hours 100% kill after 24 hours 100% kill after 48 hours
Didecyl dimethyl ammonium chloride	1.0 ppm for 24 hours	100% kill
Akyldimethylbenzyl ammonium chloride and Akyldimethylethylbenzyl Ammonium chloride	10.0 ppm for 48 hours 20.0 ppm for 48 hours	100% kill after 144 hours 100% kill after 72 hours
Endod (plant extract)	15 ppm continuous	100% kill
1, 1', - (methyliminio) bis (3-chloro-2-propanol), polymer with N,N,N',N'-tetramethyl-1, 2-ethanediamine and potassium ion	0.75 ppm for 1295 hours at 20°C 2.25 ppm for 346 hours at 20°C 0.75 ppm for 1295 hours 2.25 ppm for 633 hours	100% kill SL* < 11mm 100% kill SL* < 11mm 100% kill SL* < 14mm 100% kill SL* < 14mm SL* = Shell Length
	Metallic Molluscicides	
Potassium ions - • KH2 PO4 • KOH • KCL	 160 - 640 ppm continuous >10 ppm 50 ppm for 48 hours	 100% kill 100% veliger kill 100% kill
Tri-butyl tinoxide	Surface coatings reapplied every 1 - 2 years	High success
Copper ions	5 ppm for 24 hours	100% kill
Silver ions	5 ppm for 24 hours	72% kill
Mercury ions	5 ppm for 24 hours	57% kill
Zinc ions	5 ppm for 24 hours	5% kill
Lead ions	5 ppm for 24 hours	0% kill
Copper sulfate	100 ppm for 5 hours at 22.5°C 300 ppm for 5 hours at 22.5°C	40% kill 55% kill

Table 2. A guide to further information on various molluscicides (Spencer and Getsinger 2002). Accessed March 31, 2014. Online at

The link provided is no longer valid. This online document was revised 11/6/2017.

Chemical Biocide/Molluscicide	Waller et al. 1993	Claudi and Mackie 1994	EPRI 1993	McMahon et al. 1994
I. Oxidizing Biocides				
Chlorine		X	X	X
Chlorine dioxide		X		X
Chloramine		X		X
Bromine		X		
Ozone		X	X	X
Sodium hypochlorite		X		
Hydrogen peroxide		X	X	
Potassium permanganate	X	X		X
II. Nonoxidizing Biocides/Molluscicides				
Ammonium nitrate		X		
Potassium salts	X	X		X
Clamtrol CT-1	X	X	X	X
Clamtrol CT-4	X			
Calgon H130 M	X	X	X	X
Bulab 6002	X	X	X	X
Bulab 6009	X			X
Baluscide				X
Macrotrol 7326			X	X
Mexel 432*				
Actibrom 1338	X	X	X	
TFM				
III. Metallic Molluscicides				
Copper sulfate	X			X
Potassium chloride	X	X		X
Potassium hydroxide				X
Copper ions				X
Silver ions				X

Table 3. Mussel chemical treatments and their toxicology data on nontarget species (Spencer and Getsinger 2002). Accessed March 31, 2014. Online at

The link provided is no longer valid. This online document was revised 11/6/2017.

ACTI-BROM 1338	Rainbow Trout 96hr Static LC50 >1000 mg/L	Bluegill Sunfish 96hr Static LC50 >1000 mg/L		
Bulab 6002	Rainbow Trout 96hr LC50 0.047 mg/L	Bluegill Sunfish 96hr LC50 0.21 mg/L	*Daphnia magna* 48hr LC50 0.37 mg/L	Fathead Minnow 96hr LC50 0.26 mg/L
Bulab 6009	Rainbow Trout 96hr LC50 0.117 mg/L	Fathead Minnow 96hr LC50 0.037 mg/L	*Daphnia magna* 96hr LC50 0.07 mg/L	
Calgon H-130	Rainbow Trout 96hr LC50 1.1 mg/L	Coho Salmon 96hr LC50 1.0 mg/L	Bluegill Sunfish 96hr LC50 0.32 - 0.59 mg/L	*Daphnia magna* 48hr LC50 0.094 mg/L
Chlorine	Vertebrate 0.040 LC50	Non-target Invertebrate 0.017 LC50 Daphnids		
CLAM -TROL CT - 1	Rainbow Trout Flow-Through 96hr LC50 8.1 mg/L	Fathead Minnow Flow-Through 96hr LC50 2.9 mg/L	*Daphnia magna* Flow-Through 48hr LC50 0.2 mg/L	*Ceriodaphnia* Flow-Through 48hr LC50 0.14 mg/L
CLAM -TROL CT -2	Rainbow Trout Flow-Through 96hr LC50 2 mg/L	Fathead Minnow Flow-Through 96hr LC50 0.72 mg/L	*Daphnia magna* Flow-Through 48hr LC50 0.04 mg/L	Mysid Shrimp Flow-Through 96hr LC50 0.16 mg/L
CLAM -TROL CT - 3	Rainbow Trout Flow-Through 96hr LC50 10 mg/L	Fathead Minnow Flow-Through 96hr LC50 4 mg/L	*Daphnia magna* Flow-Through 48hr LC50 0.2 mg/L	Mysid Shrimp Flow-Through 96hr LC50 0.8 mg/L
EVAC	Largemouth Bass Static 96hr LC50 0.1 - 0.3 mg/L Diamine salt	Bluegill Sunfish Static 48hr LC50 0.8 mg/L	Redear Sunfish Static 96hr LC50 0.1 - 0.2 mg/L Diamine salt	Golden Shiner Flow Through 120hr LC50 0.37 mg/L
Macro Tech Copper nitrate	Striped Bass 96hr LC50 53 - 55 hardness 4,000 - 4,300 mg/L	Largemouth Bass 96hr LC50 100 hardness 6,970 mg/L	White Perch 96hr LC50 53 hardness 6,200 mg/L	
Macro Tech Copper chloride	Cutthroat Trout 96hr LC50 18 - 205 hardness 11.0 mg/L	Rainbow Trout 96hr LC50 42 - 194 hardness 11.0 mg/L	Bluegill Sunfish 96hr LC50 43 hardness 11.0 mg/L	
MEXEL	Rainbow Trout 96hr LC50 11.0 mg/L	Fathead Minnow 96hr LC50 8.06 mg/L	Fathead Minnow 80 minutes LC50 2.8 mg/L	*Daphnia magna* 80 minutes LC50 3.0 mg/L
NALCO Macrotrol	Rainbow Trout Static acute 96hr LC50 1.25 mg/L	Bluegill Sunfish Static acute 96hr LC50 0.42 mg/L	Grass Shrimp Static acute 96hr LC50 2.81 mg/L	
Potassium	Rainbow Trout No observed effect at level 100 mg/L	Fathead Minnow No observed effect at level 100 mg/L	*Daphnia magna* No observed effect at level 100 mg/L	*Anodonta imbecillus* Without sediment LC50 76 mg/L
Sodium Hypochlorite	Rainbow Trout 48hr LC50 0.07 mg/L	Fathead Minnow 96hr LC50 .5.9 mg/L		
Sodium Bromide	Fathead Minnow 96hr Static LC50 16,479 mg/L	*Poecilia reticulata* 96hr Static LC50 225 mg/L	*Daphnia magna* 48hr Static LC50 7,900 mg/L	
Veligon	Rainbow Trout 96hr LC50 0.37 mg/L	Bluegill Sunfish 96hr LC50 0.82 - 1.3 mg/L	*Daphnia magna* Clear water 48hr LC50 0.99 mg/L	*Daphnia magna* In 50ppm clay suspension 48hr LC50 1.2 - 2.5 mg/L

Literature citied

Angarano, M. B., R. F. McMahon, and J. A. Schetz. 2009. Cannabinoids inhibit zebra mussel (*Dreissena polymorpha*) byssal attachment: a potentially green antifouling technology. Biofouling 25:127-138.

Benson, A. J., D. Raikow, J. Larson, and A. Fusaro. 2014 (a). *Dreissena polymorpha*. USGS Nonindigenous Aquatic Species Database, Gainesville, FL. http://nas.er.usgs.gov/queries/FactSheet.aspx?speciesID=5 Revision Date: 6/6/2012

Benson, A. J., M. M. Richerson, E. Maynard, J. Larson, and A. Fusaro. 2014 (b). *Dreissena rostriformis bugensis*. USGS Nonindigenous Aquatic Species Database, Gainesville, FL. http://nas.er.usgs.gov/queries/factsheet.aspx?speciesid=95 Revision Date: 6/28/2012.

Bially, A. and H. J. MacIsaac. 2000. Fouling mussels (*Dreissena* spp.) colonize soft sediments in Lake Erie and facilitate benthic invertebrates. Freshwater Biology 43:85-97.

Boelman, S.F., F.M. Neilson, E.A. Dardeau, and T. Cross. 1997. Zebra mussel (*Dreissena polymorpha*) control handbook for facility operators, first edition. Miscellaneous Paper EL-97-1, U.S. Army Engineer Waterways Experiment Station, Vicksburg, MS.

Britton, D. and S. Dingman.(2011). Use of quaternary ammonium to control the spread of aquatic invasive species by wildland fire equipment. Aquatic Invasions 6: 169–173.

Bruner, K.A., S.W. Fisher, and P.F. Landrum. 1994. The role of the zebra mussel, Dreissena polymorpha, in contaminant cycling: II. Zebra mussel contaminant accumulation from algae and suspended particles, and transfer to the benthic invertebrate, Gammarus fasciatus. Journal of Great Lakes Research 20:735-750.

Claudi, R. and G.L. Mackie. 1994. Practical Manual for Zebra Mussel Monitoring and Control. Lewis Publishers, CRC Press, Boca Raton, FL. 227 pp.

Claxton, W.T., A.B. Wilson, G.L. Mackie, and E.G. Boulding. 1998. A genetic and morphological comparison of shallow- and deep-water populations of the introduced dreissenid bivalve Dreissena bugensis. Canadian Journal of Zoology 76(7):1269-1276.

Culver, C.S., S.L. Drill, M.R. Myers, and V.T. Borel. 2009. Early detection monitoring manual for quagga and zebra mussels. California Sea Grant Extension Program, University of California cooperative Extension, California Sea Grant College Program, University of California, San Diego, CA.

Early, T.A. and T. Glonek. 1999. Zebra mussel destruction by a Lake Michigan sponge: populations in vivo P^{31} Nuclear Magnetic Resonance, and Phospholipid profiling. Environmental Science and Technology. 33(12): 1957-1962

EPRI (Electric Power Research Institute). 1993. Hazard identification of commercially available biocides to control zebra mussels and Asiatic clams. TR-103175, Syracuse Research Corporation, Syracuse, NY.

GISD (Global Invasive Species Database). 2009. *Dreissena polymorpha*. Online at http://www.issg.org/database/species/ecology.asp?si=50.

GISD (Global Invasive Species Database). 2010. *Dreissena bugensis*. Online at http://www.issg.org/database/species/ecology.asp?si=918&fr=1&sts=sss&lang=EN

Hart, S., M. Klepinger, H. Wandell, D. Garling, and L. Wolfson. 2000. Integrated Pest Management for Nuisance Exotics in Michigan Inland Lakes. Michigan State University Extension. http://michigan.gov/documents/deq/deq-water-great-lakes-aquatics-IPM-manual_249296_7.pdf.

Holland, R.E. 1993. Changes in planktonic diatoms and water transparency in Hatchery Bay, Bass Island Area, Western Lake Erie since the establishment of the zebra mussel. Journal of Great Lakes Research 19:617-624.

MacIsaac, H. J. 1994. Comparative growth and survival of *Dreissena polymorpha* and *Dreissena bugensis*, exotic molluscs introduced to the Great Lakes. Journal of Great Lakes Research 20:783-790.

Marsden, J.E., A.P. Spidle, and B. May. 1996. Review of genetic studies of *Dreissena* spp. American Zoology 36:259-270.

May, B. and J.E. Marsden. 1992. Genetic identification and implications of another invasive species of dreissenid mussel in the Great Lakes. Canadian Journal of Fisheries and Aquatic Science 49:1501-1506.

McMahon, R. F., Ussery, T. A., and M. Clarke. 1994. Review of zebra mussel control methods, Technical Note ZMR-2-14. Zebra Mussel Research Program, U.S. Army Engineer Waterways Experiment Station, Vicksburg, MS.

Mills, E. L., G. Rosenberg, A. P. Spidle, M. Ludyanskiy, Y. Pligin, and B. May. 1996. A review of the biology and ecology of the quagga mussel (Dreissena bugensis), a second species of freshwater Dreissenid introduced to North America. Amer. Zool. 36:271-286.

Morse, J.T. 2009. Assessing the effects of application time and temperature on the efficacy of hot-water sprays to mitigate fouling by *Dreissena polymorpha* (Zebra mussels Pallas). Biofouling: The Journal of Bioadhesion and Biofilm Research. 25(7): 605-610.

Raikow, D.F. 2002. How the feeding ecology of native and exotic mussels affects freshwater ecosystems, Doctoral Dissertation, Michigan State University.

Roditi, H.A., N.S. Fisher, and S.A. Sanudo-Wilhelmy. 2000. Uptake of dissolved organic carbon and trace elements by zebra mussels. Nature 407:78-80.

Scheffer, M., S.H. Hosper, M.L. Meijer, B. Moss, and E. Jeppesen. 1993. Alternative equilibria in shallow lakes. Trends in Ecology and Evolution 8:275-279.

Snyder, F.L., M.B. Hilgendorf, and D.W. Garton. 1997. Zebra Mussels in North America: The invasion and its implications. Ohio Sea Grant, Ohio State University, Columbus, OH. http://ohioseagrant.osu.edu/_documents/publications/FS/FS-045%20Zebra%20mussels%20in%20North%20America.pdf.

Spencer, S.L. and K.D. Getsinger. 2002. Zebra mussel chemical control guide, ERDC/EL TR-00-01, U.S. Army Research and Development Center, Vicksburg, Mississippi. Online at *The link provided is no longer valid. This online document was revised 11/6/2017.*

Stewart, T.W. and J.M. Haynes. 1994. Benthic macroinvertebrate communities of southwestern Lake Ontario following invasion of Dreissena. Journal of Great Lakes Research 20:479-493.

Waller, D.L., J.J. Rach, W.G. Cope, and L.L. Marking. 1993. Toxicity of candidate Molluscicides to zebra mussels (*Dreissena polymorpha*) and selected nontarget organisms. Journal of Great Lakes Research. 19:695-702.

Wimbush, J., M.E. Frischer, J.W. Zarzynski, and S.A. Nierwicki-Bauer. 2009. Eradication of colonizing populations of zebra mussels (Dreissena polymorph) by early detection and SCUBA removal: Lake George, NY. Aquatic Conservation: Marine and Freshwater Ecosystems. 19: 703-713.

Invasive Species Alert

Zebra & Quagga Mussel

(*Dreissena polymorpha*) & (*Dreissena rostriformis bugensis*)
Established in Michigan

Identification:

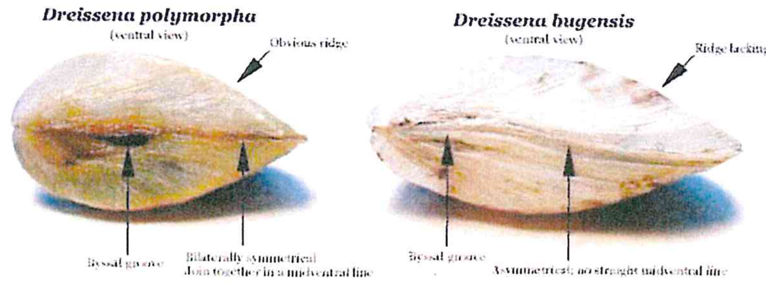


Photo by Myriah Richerson

- Average length of a zebra mussel is about 1 inch, quagga mussels can reach 2 inches
- Zebra shells are triangular in shape and can sit flat on the ventral side
- Quagga shells are more rounded and fan-shaped and won't sit flat on the ventral side
- Colors vary - shell color ranges from almost white to tan or brown in both species, usually have darker concentric rings but are not always pronounced

Habitat: Freshwater lakes, rivers, and reservoirs. Zebra mussels require hard substrates to latch onto, while quagga mussels can attach to hard or soft substrates in water depth up to 130 meters. This leads to a less restricted range of suitable habitat than for the zebra mussel.

Diet: Mussels are filter feeders that consume algae and phytoplankton in the water. Zebra mussels can filter up to 1 liter of water each day.

Native Range: Zebra mussels are native to freshwater rivers and lakes in Eastern Europe and western Asia. Quaggas are native to areas in the Ukraine and the Ponto-Caspian Sea.

Local Concern: Filter feeding removes a substantial amount of food for zooplankton, increases water transparency, and leads to an accumulation of pseudofeces. Pseudofeces accumulation creates foul environments. Zebra mussels have been known to colonize on native mussel species in groups of up to 10,000 individuals, rendering the native mussel immobile and unable to survive. There is also an economic cost associated with mussel attachment to pipes and other underwater structures.

Means of Introduction: Ballast water from transoceanic vessels

Report this species at www.misin.msu.edu or download the MISIN smartphone app and report it from your phone

Hess Lake Bibliography

1. Edmunds Engineering. June 1982. Hess Lake Engineering Feasibility report. 33 pages.
2. Michigan Department of Natural Resources. November 1984. A Benthic Macroinvertebrate Survey of Wheeler Drain and Brooks Creek in Relation to Hess Lake, Newaygo County, Michigan, December 6, 1982. 10 pages.
3. Hess Lake Clean Lakes Grant Application. 1988. 12 pages + Appendix.
4. Hess Lake Improvement Board. 6/27/88. Letter to Michigan Dept. of Natural Resources in response to EPA comments concerning Clean Lakes Program grant application. 19 pages.
5. Progressive Architects Engineers Planners. March 1989. Hess Lake Wheeler Drain phosphorus removal system feasibility evaluation report. Project No. 8809-24. 21 pages.
6. Wade-Trim/Granger. November 1990. Brooks Lake and Hess Lake Water Quality Study. 33 pages.
7. ANATech Analytical Labs. 1991, 1992. Hess Lake, Wheeler Drain, Algers Creek Water Quality Analysis Results. 32 pages.
8. FTC&H. May 1993. Executive Summary for Hess Lake, Grant and Brooks Townships, Newaygo County, Michigan. Project No F92347. 20 pages.
9. FTC&H. December 1993. Hess Lake Diagnostic Evaluation and Feasibility Report for Grant and Brooks Townships, Newaygo County, Michigan. Project No F92347. 23 pages.
10. FTC&H, 1/3/94. Hess Lake U.S. EPA Phase 2 Grant Application.
11. FTC&H. 1/3/95. U.S. EPA Clean Lakes Program Phase II grant application for Hess Lake, Newaygo County FY 1994. 20 pages.
12. Hess Lake Improvement Board. February 1995. Harvesting and Herbicide. 13 pages.
13. Grant application, Michigan's Nonpoint Source Program (319) Water Quality Planning Program (604[b]). 4/3/95. 4 pages.
14. FTC&H. 1996. Engineering drawings of Wheeler Drain project. 11 sheets.
15. Oppie & Sons Excavating, Inc. 9/11/96. Performance Bond. Wheeler Sediment Basin Cleanouts.
16. Brooks Township, Newayo County. December 30, 2005, adopted. Ordinance 05-50. Inspection of residential on-site sewage disposal systems at the time of property transfer ordinance.
17. Spicer Group. May 6, 2009. Initial Summary of Water Quality Data for Hess Lake, Newaygo County. Project #117776SG2009. 9 pages.
18. Spicer Group. August 2010. Hess Lake Bathymetry & Sediment Depth Report. 17 pages.
19. Phil Laven. Hess Lake Phosphorus Reports. 5-2-2013. 1 page.
20. Ben B. Lester, Zone 3 Representative. 6/22/13. Association Annual Meeting Report. 1 page.
21. Jeff Fischer. May 23, 2014. MDEQ permit for installation of 14 aerators. 14 pages.
22. Savin Lake Services. 6/1/2015. Herbicide treatment invoice. 1 page.
23. Tony Cunningham. 6/12/2015. Hess Lake Standard Aquatic Vegetation Summary Sheet. 2 pages.
24. Tony Cunningham. 8/7/2015. Hess Lake Standard Aquatic Vegetation Summary Sheet. 2 pages.
25. Hess Lake Improvement Board. July 11, 2016. Minutes. 2 pages.

Hess Lake Deep Basin Water Quality Data

Date	Station	Depth (feet)	Temper-ature (°F)	Dissolv. Oxygen (mg/L)	Total Phosphorus (µg/L)	Total Susp. Solids (mg/L)	Chloride (mg/L)	pH (S.U.)	Total Alkalinity (mg/L as $CaCO_3$)
12-Apr-18	1	1	45	9.5	19	10.0	13.2	9.0	137
12-Apr-18	1	5	45	9.8	25	8.8	13.2	8.9	137
12-Apr-18	1	10	45	9.8	21	24.0	14.8	8.9	142
12-Apr-18	1	13	45	9.4	20	10.8	13.1	8.9	135
12-Apr-18	2	1	44	9.2	22	8.4	13.1	9.4	139
12-Apr-18	2	5	44	9.6	21	8.4	13.1	9.3	142
12-Apr-18	2	10	44	9.6	5	10.0	13.2	9.4	138
12-Apr-18	2	15	44	9.3	<5	10.8	13.5	9.4	138
12-Apr-18	2	20	44	9.8	16	8.8	13.3	9.4	137
12-Apr-18	3	1	45	9.8	12	4.8	13.2	9.4	138
12-Apr-18	3	5	44	9.5	16	10.4	13.2	9.4	136
12-Apr-18	3	10	44	10.8	<5	14.0	13.1	9.4	136
12-Apr-18	3	15	44	11.5	<5	9.6	13.3	9.3	140

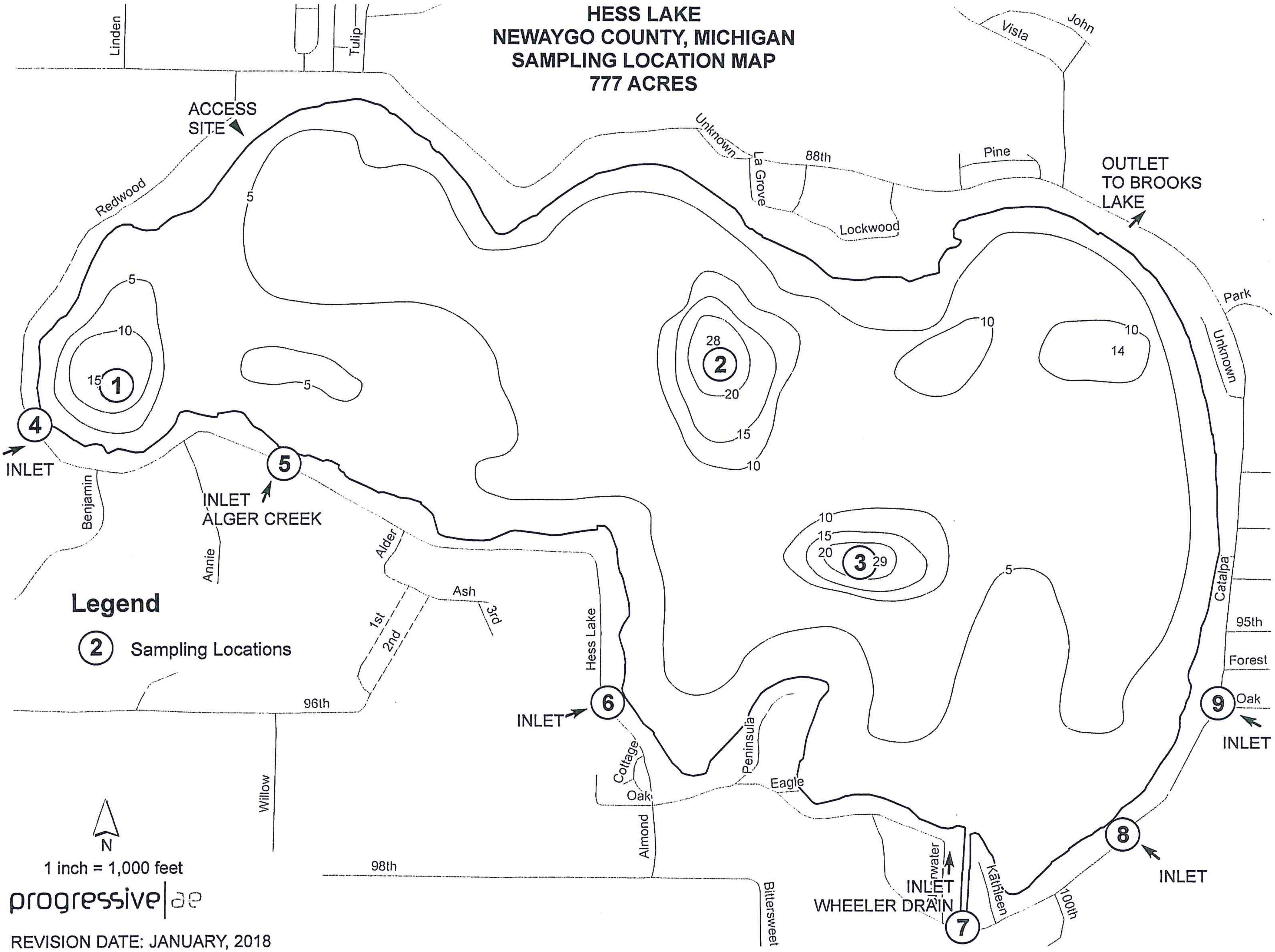
Hess Lake Surface Water Quality Data

Date	Station	Chlorophyll-*a* (µg/L)	Secchi Transparency (feet)
12-Apr-18	1	4	2.5
12-Apr-18	2	11	2.5
12-Apr-18	3	12	2.5

Hess Lake Tributary Water Quality Data

Date	Station	Stream	Total Discharge (cfs)	Phosphorus (µg/L)	Total Suspended Solids (mg/L)
12-Apr-18	4	West trib	0.2	5	<4
12-Apr-18	5	Alger Creek	0.8	45	21
12-Apr-18	6	South trib	0.2	<5	<4
12-Apr-18	7	Wheeler Drain	10.5	329	194
12-Apr-18	8	Southeast trib	0.4	22	19
12-Apr-18	9	East trib	1.4	7	9

HESS LAKE
NEWAYGO COUNTY, MICHIGAN
SAMPLING LOCATION MAP
777 ACRES



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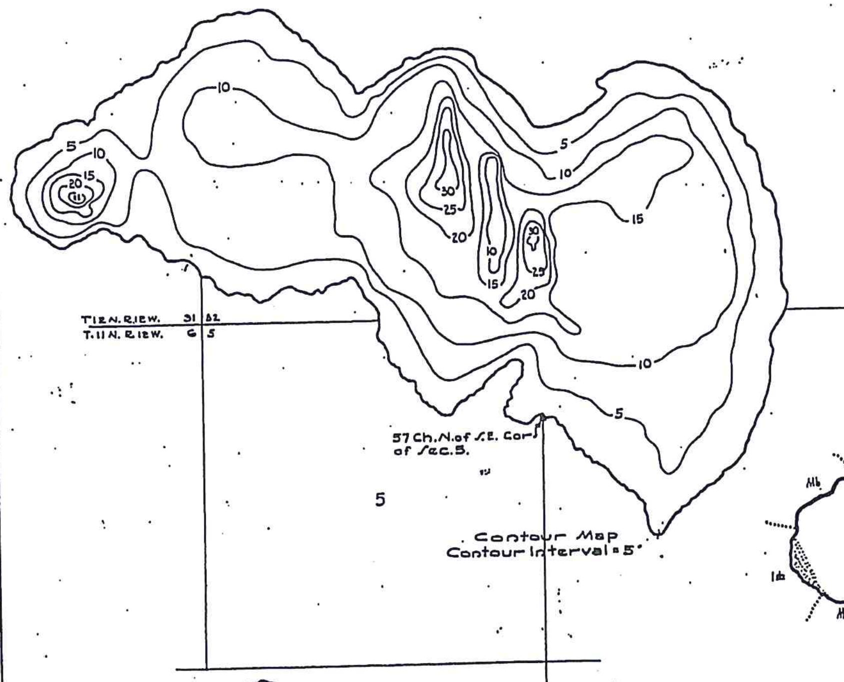
U. S. Department of Agriculture
Forest Service
LAKE AND STREAM SURVEY

HESS LAKE

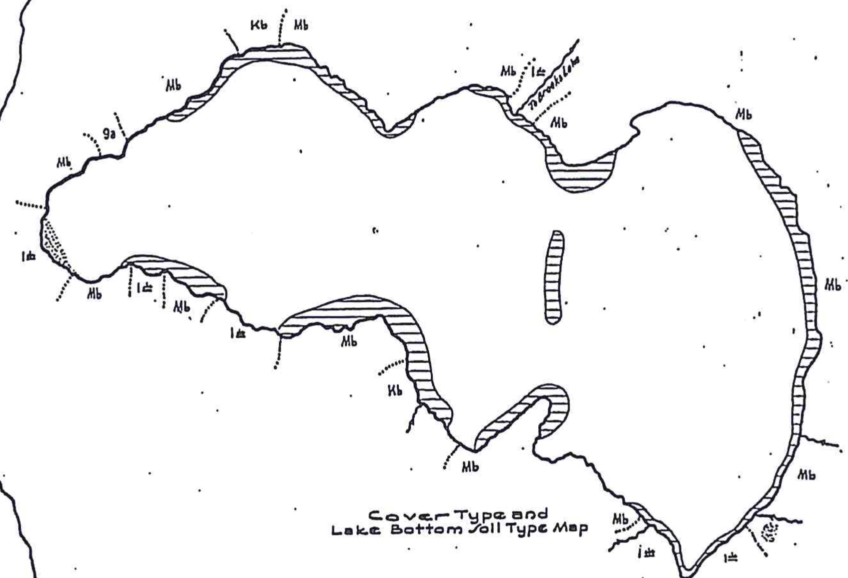
Manistee National Forest
Michigan

Location Sec's. 31 32 33 35 T.12N. R.12W. T.11N. R.12W.
Scale, 4" = 1 Mile

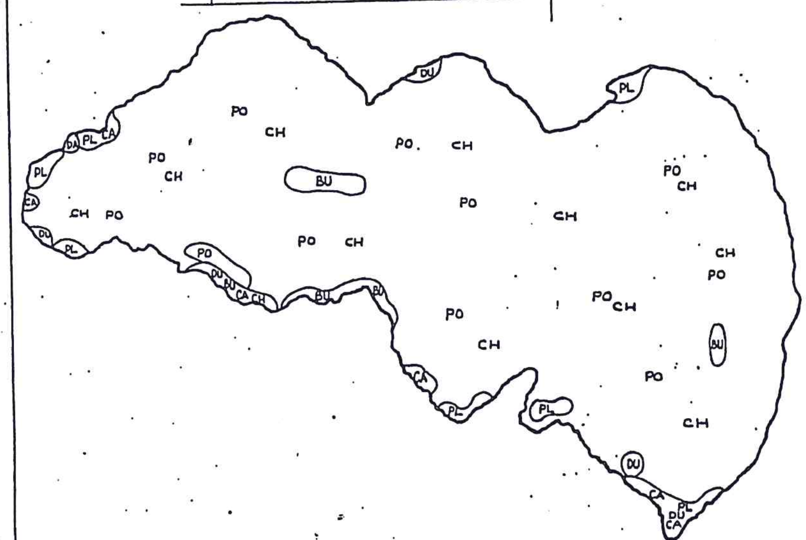
Area 1125 Acres
Length of Shoreline 7 Miles



Contour Map
Contour Interval = 5'



Cover Type and
Lake Bottom Soil Type Map



Aquatic Vegetation Map

J-8-27

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U.S. Department of Agriculture
Forest Service
LAKE AND STREAM SURVEY

MASTER SHEET FOR AQUATIC VEGETATION

	LATIN NAME	COMMON NAME
WC	Vallisnaria spiralis	Wild Celery
RI	Zizania aquatica	Wild Rice
DU	Saggittaria	Duck Potato
CA	Typha	Cat-tail
CT	Ceratophyllum	Coontail
SF	Acorus Calamus	Sweet Flag
SA	Myrica gale	Sweet Gale
MT	Spiraea latifolia	Meadow Sweet
PS	Persicaria amphibia	Water Smart Weed
BR	Sparganium	Bur-reed
BW	Xanthium	Bur-weed
CF	Batrachium trichophyllum	White Water Crowfoot
LD	Lobelia Dortmanna	Water Gladiola
MP	Heteranthera dubia	Water Star Grass
CH	Chara	Musk Grass
QU	Isoetes Braunii	Braun's Quillwort
DT	Pontederia cordata	Pickeral Weed
TM	Fontinalis	True Water Mosses
WA	Radicula	Watercress
JG	Equisetum	Horsetail
WS	Brasenia Schreberi	Water shield
BJ	Agropyron Smithii	Western Wheat Grass
WI	Iridaceae	White Iris
WM	Myriophyllum	Milfoil
ML	Myriophyllum tenellum	Slender Water Milfoil
MI	Myriophyllum spicatum	Spiked Water Milfoil
DW	Lemna	Duckweed
DS	Lemna trisulca	Star Duckweed
DM	Lemna minor	Lesser Duckweed
SE	Carex	Sedges
SG	Carex vesicaria	Inflated Sedge
DA	Dulichium arundinaceum	Dulichium
PL	Nymphaea and Castalia	Pond Lilies
PLI	Castalia tuberosa	White Water Lily
PL2	Nymphaea advena	Large Yellow Pond Lily
PL3	Nymphaea microphylla	Small Yellow Pond Lily
PL4	Nymphaea saggitaefolia.	Arrow Leaved Pond Lily
BU	Scirpus	Bul-rush
SV	Scirpus validus	American Great Bul-rush
ST	Scirpus americanus	Three Square Rush
CR	Scirpus subterminalis	Water Club Rush
TR	Mariscus mariscoides	Twig Rush
SR	Elocharis palustris	Creeping Spike Rush
NR	Elocharis acicularis	Least Spiked Rush
GA	Chlorophyceae	Green Algae
BA	Cyanophyceae	Blue Green Algae
MG	Paneularia canadensis	Rattlesnake Grass
SW	Triadenum virginicum	Marsh St Johnswort
BO	Juncus pelocarpus	Brown Fruited Rush
BL	Utricularia	Bladderwort
WW	Philotria canadensis	Water Weed
RE	Phragmites	Reeds
PW	Eriocaulon	Pipewort
PI	Eriocaulon septangulare	Seven angled Pipewort
HI	Hippuris vulgaris	Bottle Brush

	LATIN NAME		COMMON NAME
PO	Potamogeton		Pondweeds
PH	Potamogeton	heterophyllus	Various Leaved Pondweed
PB	"	dimorphus	Spiral Pondweed
PX	"	foliosus	Leafy Pondweed
PP	"	pectinatus	Sago Pondweed
PR	"	praelongus	White Stemmed Pondweed
PN	"	natans	Common Floating Pondweed
PF	"	pusillus	Small Pondweed
PK	"	amplifolius	Large Leaved Pondweed
PE	"	epihydrus	Nuttalls Pondweed
PG	"	compressus	Eel Grass Pondweed
PA	"	americanus	Long Leaved Pondweed
PT	"	perfoliatus	Red Head Grass
PU	Naias flexilis		Bushy Pondweed
PC	Potamogeton crispus		Curly Muckweed

Legend for Vegetation Mapping on extensive basis where individual species are not indicated.

○ Emergent Vegetation.

● Submerged and Floating Leaved Vegetation.

Boundaries of Vegetation should be indicated and Symbol applicable to the Vegetation Area shown.

WATER VEGETATION
DEGREE OF ABUNDANCE
1 - Abundant
2 - Common
3 - Rare
Example CHara - CH2

Britton and Brown's Illustrated Flora of the Northern States and Canada. Used in compiling this Legend Sheet.