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.

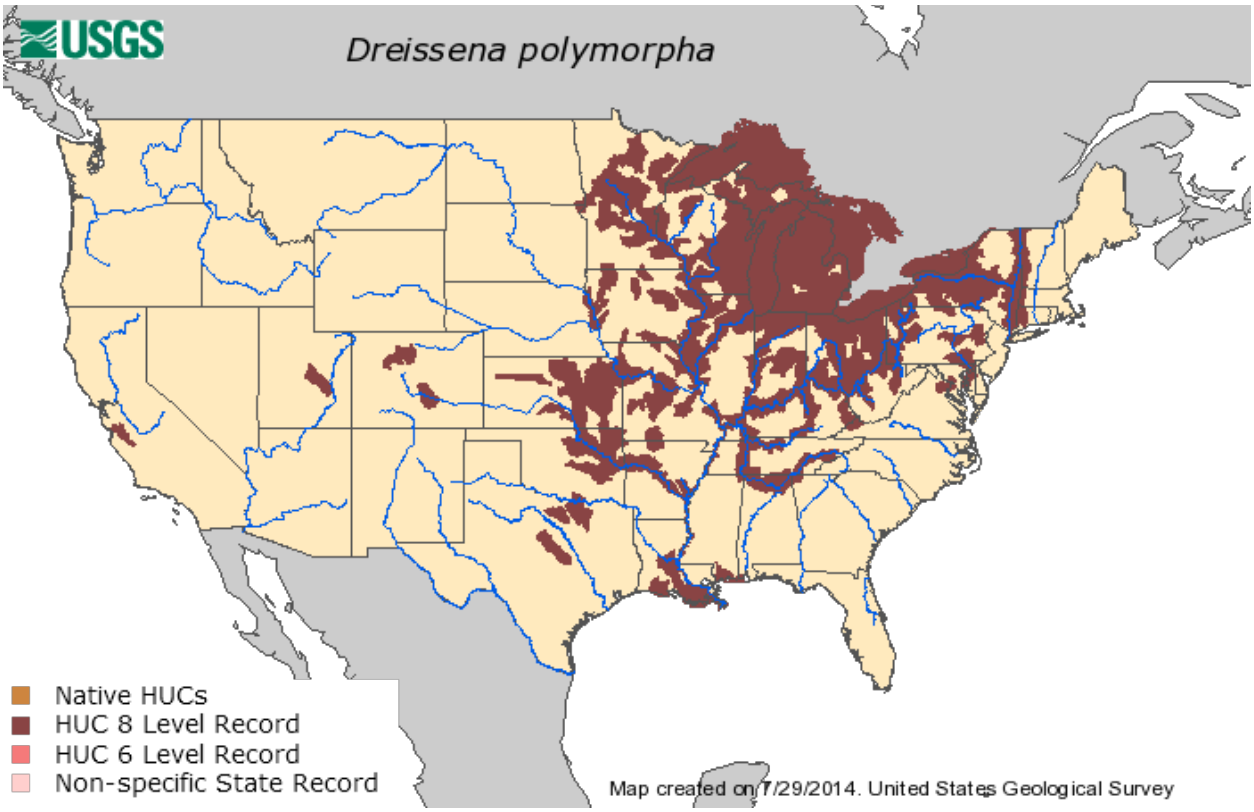

Figure 1. Distribution of zebra mussels in the United States (Benson et al. 2014a). Accessed July 29, 2014.

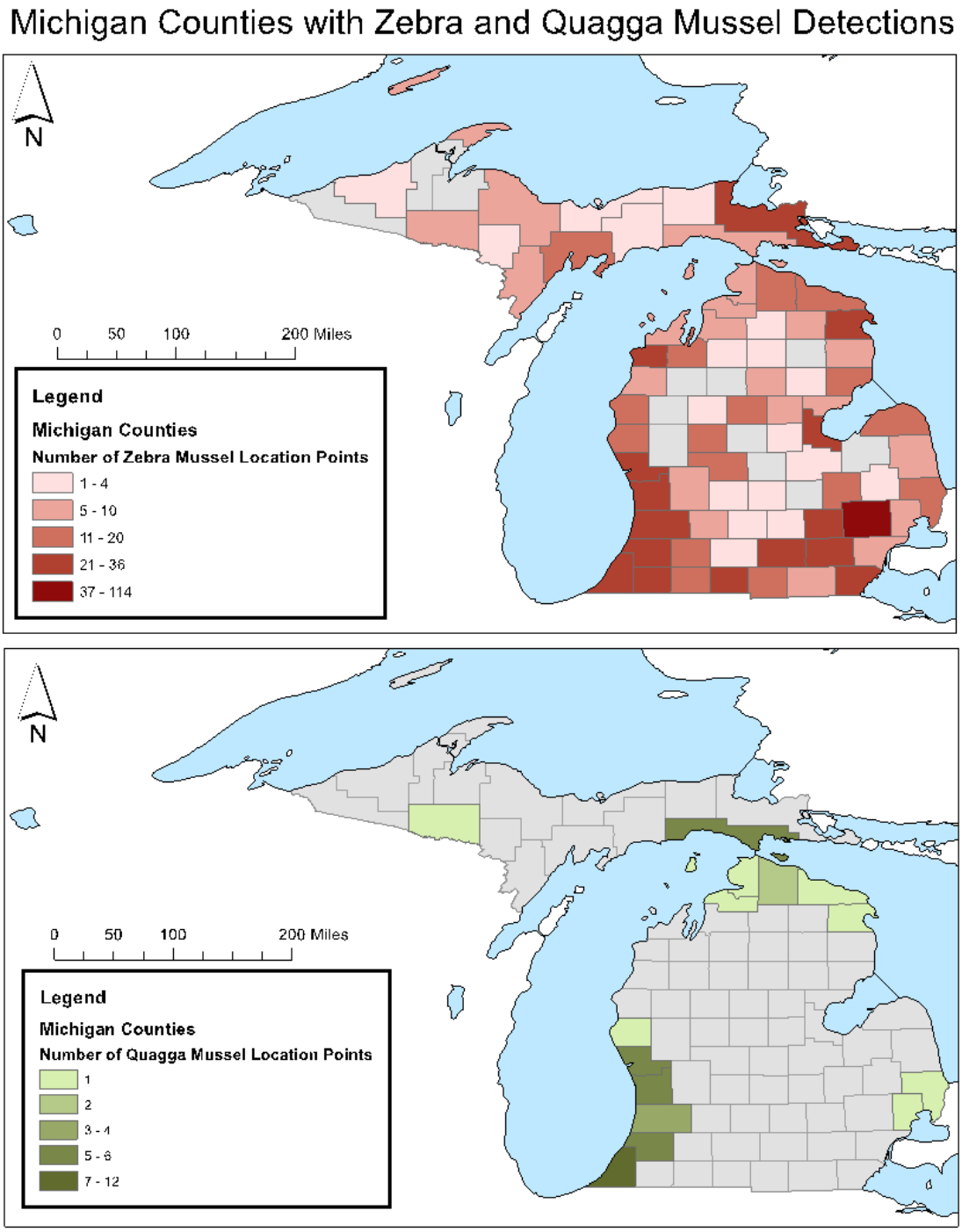

Figure 2. Number of unique coordinate location points within Michigan counties at which zebra and quagga mussels were detected. This data is according to the United States Geological Survey (USGS), Midwest Invasive Species Information Network (MISIN, accessed May 22, 2014) and Biodiversity Information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

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