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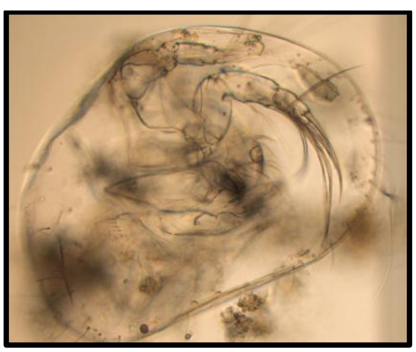
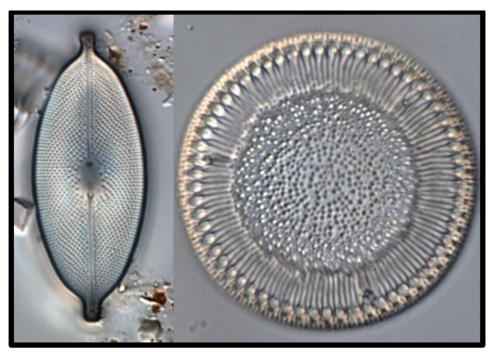
Natural Resource Stewardship and Science

Biological Studies and Mapping of Shoreline Rock Pools in Three Lake Superior National Parks

Natural Resource Technical Report NPS/MWRO/NRTR—2014/907





ON THE COVER

Clockwise from top: Freighter on Lake Superior and rock pools on Passage Island, Isle Royale National Park (photograph by Alexander Egan, 2010); Diatoms *Decussata placenta* and *Handmannia* (*Puncticulata*) *bodanica* from pools at Pictured Rocks National Lakeshore (photograph by Mark Edlund, 2012); Ostracod (photograph by Toben Lafrancois, 2012); Chironomidae tubes in pools near Datolite Mine, Isle Royale (photograph by Alexander Egan, 2011).

Biological Studies and Mapping of Shoreline Rock Pools in Three Lake Superior National Parks

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Executive Summary

The primary objective of this coastal rock pool study was to characterize the biological, physical, and chemical communities and conditions in this relatively unknown habitat in three Lake Superior national park units: Isle Royale National Park (ISRO), Apostle Islands National Lakeshore (APIS), and Pictured Rocks National Lakeshore (PIRO). The focus was on water quality and macroinvertebrates (particularly Chironomidae midges), zooplankton, and diatom communities. We documented community composition, phenology, and habitat preferences of these focal groups, and mapped the distribution of all rock pool habitats along the southern shoreline of Isle Royale, the park with by far the most pool habitat, from Malone Bay (in the center of the park) to Passage Island (east end of the archipelago).

Sampling varied slightly in each of the parks due to unique terrain. Isle Royale contained what we considered to be ideal rock pool habitat, with basaltic bedrock and sloping shorelines creating excellent conditions for pool formation in depressions and cracks. Sandstone bedrock fomations at Apostle Islands and Pictured Rocks have much more limited conditions for rock pool formation, and Pictured Rocks has unique habitat components. Biological sampling occurred in 2009 and 2010 at Isle Royale, and in 2010 at the other two parks. Mapping occurred in 2011 and 2012 at Isle Royale.

Habitats were split into two strata during sampling. The first stratum was composed of a splash zone, where wave wash from Lake Superior is important in disturbance and pool recharge, and a lichen zone that is generally above wave influence and is recharged via rain and overland flow. The second stratum contained both permanent pools—deeper than ten centimeters (four inches) and should maintain a water basin even in mild drought conditions—and ephemeral pools—less than ten centimeters deep and should experience a regular drying-and-refilling pattern depending on precipitation or wave recharge.

Macroinvertebrates

Chironomidae (Insecta: Diptera), commonly called non-biting midges, was the focal group for macroinvertebrate sampling because taxa in this family are generally ubiquitous and abundant in aquatic habitats, and have a well established history of being used as biological indicators. Surface-floating pupal exuviae—the shed exoskeleton following emergence from pupal stage to adult stage—were the focus of chironomid sampling. Exuviae can be collected and identified relatively easily, taxa utilizing inaccessible niches are readily detectable, and there is a reduced impact on the community by minimizing removal of living chironomids.

Fifty-nine genera and subgenera of Chironomidae were detected in the three parks, with 30 genera occurring in at least two parks. Twenty-nine taxa were limited to a single park. Nineteen genera were present in all three parks, with several known to survive in desiccation-prone habitats. At Isle Royale, where most of the sampling effort occurred, 46 genera/subgenera were detected, although analyses estimated 54 genera were actually present. This number is probably a low estimate, but it is higher than expected given that rock pool habitats were expected to be low in nutrients and niches, and have regular disturbance from waves or drying. Much of the diversity was derived from genera where only a handful of individuals were detected, while two genera dominated samples: *Orthocladius*

(*Eudactylocladius*) and *Psectrocladius* (*Psectrocladius*). Apostle Islands had the lowest species richness, with no genera unique to the park, and was faunistically similar to Pictured Rocks. At Pictured Rocks, most diversity occurred in the lichen zone, and the community appeared to be strongly influenced by stream inputs to the pools.

Significant differences occurred in communities between the two zones, and clearly one habitat or the other was favored by most genera. Only 12 of the 46 genera were shared between the lichen and splash zones at Isle Royale. In contrast, significant similarities existed between communities inhabiting permanent and ephemeral pool types, indicating little preference between types despite the apparent abiotic differences between these pools. Suggestions are given for future monitoring sites, timeframes, and target chironomid taxa.

In addition to chironomids, the broader aquatic insect community at Isle Royale was qualitatively sampled using sweep nets, dip nets, a pan and sieve, and an aspirator. There appeared to be a distinct rock pool community composed of springtails (Collembola), darner dragonfly larvae (Aeshnidae), waterboatmen (Corixidae), waterstriders (Gerridae), diving beetles (Dytiscidae), caddisflies (Apataniidae, Hydropsychidae, and Limnephilidae), mosquitoes (Culicidae), crane flies (Tipulidae), and scuttle flies (Phoridae).

Zooplankton

Zooplankton in rock pools at the three national parks were quantitatively sampled with a 30µm mesh net modified for use in rock pools. A total of 115 samples were counted and 177 zooplankton taxa were identified. Rock pool zooplankton includes a diverse assemblage of crustacean (copepod and cladoceran) zooplankton, rotifers, and testate protists. Many species were common for Lake Superior, but most others, particularly the cladocerans and rotifers, are considered rare in Lake Superior proper. Diversity was very high, often an order of magnitude higher than nearby inland lakes. Zooplankton are present in the pools in far greater densities than nearby inland lakes or Lake Superior, indicating an abundant food source for invertebrates and amphibians.

Zooplankton abundance and diversity were highly variable between pools, with standard deviations greater than the means for most community measures. However, regional and local patterns did emerge. Rock pools at Pictured Rocks supported fewer numbers and taxa of zooplankton than the other parks and had fewer unique species. Locally, Blueberry Island and Raspberry Island at Isle Royale and both sites at Apostle Islands (Devils Island and Bear Island) had significantly higher zooplankton diversity and abundance than other sites. Principal components analysis shows that pool zone (lichen vs splash) and type (permanent vs ephemeral) explained about 21% of zooplankton species distribution. Lichen and splash zone pools supported species unique to those zones (42 and 40 respectively), and permanent pools contained the most unique species found only in that type of pool (66). Ephemeral pools and nearshore Lake Superior waters contained few species unique to those zones (15 and 12, respectively).

A surprising result was that total zooplankton abundance and diversity was not significantly different between zones even though there were different characteristic species assemblages for pools in lichen vs ephemeral zones. Broken down into major taxonomic groups, only cladocerans are significantly more abundant in a pool zone (lichen pools). All pool types, including the deceptively empty splash pools, contained high diversity and abundance of zooplankton even as they supported different taxa.

Some patterns may be obscured at the regional or park unit levels. Detailed analysis of monthly samples from Passage Island (Isle Royale) revealed a finer picture of seasonal changes and differences between pools. Zooplankton abundance increased significantly in July and August. Community composition changed from testate protist dominance in early spring to crustacean and rotifer dominance in summer, tailing off to rotifer dominance in fall. Diversity was significantly higher in lichen zone pools, driven primarily by cladoceran species diversity. Lake Superior supported significantly more testate protists, a somewhat surprising result indicating rock pool systems can harbor far more of the larger zooplankton than nearshore waters.

Water Quality and Diatoms

The basaltic bedrock geology and sloping shoreline topography of Isle Royale produce many reaches of shoreline with abundant rock pool habitat that is well differentiated into lichen and splash pool zones. In contrast, the sandstone geology and less sloping shorelines at Apostle Islands and Pictured Rocks create narrower rock pool zones, with less rock pool habitat and much weaker zonation of rock pools. On three sampling visits (May, July, October) during the 2010 field season, two lichen pools and two splash zone pools at each of four sites on Isle Royale (Datolite Mine, Blueberry Cove, Raspberry Island, and Passage Island) were physically characterized and sampled for water quality and diatom (a type of algae) communities. Rock pool sites were similarly sampled at Apostle Islands (Bear and Devils islands) and Pictured Rocks (AuSable Point, Miners Bay, and Mosquito Harbor), but only twice—once in May and again in August/September 2010. At Mosquito Harbor, in addition to lichen and splash pools, groundwater-fed pools were also sampled (cave pool and medicolous zone). Field and laboratory protocols were specifically developed for both water quality and diatom sampling and analyses.

Although no significant differences exist in size or depth of lichen vs splash pools at Isle Royale, lichen pools are located much closer to treeline and more distant from the lake than splash pools. As a result, water chemistry and hydrology differs between lichen and splash pools at Isle Royale. Lichen pools are characterized by higher levels of nutrients (total phosphorus, total nitrogen, and soluble reactive phosphorus), dissolved organic carbon, and chlorophyll, and lower levels of dissolved inorganic carbon, nitrate+nitrite-nitrogen, and specific conductivity compared to splash pools. Lichen pool hydrology is controlled by direct precipitation and runoff. Water chemistry of splash pools was very similar to Lake Superior indicating that splash pool hydrology is strongly controlled by periodic wave wash or inundation. At Apostle Islands, there are weaker differences in chemistry between pool zones. At Pictured Rocks, water quality measures do not clearly characterize pool types and most pools are further influenced by groundwater inputs.

Thermistors deployed in several pools at Isle Royale in 2010 and 2012 collected continuous temperature data that showed lichen pools commonly experienced diurnal temperature swings of 15°C and reached temperatures over 30°C on many days. In contrast, splash pools did not warm or cool as much as lichen pools on a diurnal basis due to the moderating effect of Lake Superior.

The diatom communities in rock pools are not as diverse as some Great Lakes samples, but are similar in species richness to inland lake sediment samples previously analyzed from these three parks. Rock pool diatom communities in the three parks comprised 83 diatom genera with Pictured Rocks having the greatest genus-level richness, given its multiple pool types. Several generalist diatom taxa are common in all parks and among all pools zones (e.g., *Achnanthidium* and *Gomphonema* species). Lichen pool diatom communities are further characterized by greater abundance of indicators of low pH, low conductivity, and higher productivity waters including chrysophyte cysts, *Nitzschia*, *Encyonema*, *Brachysira*, *Eunotia*, *Pinnularia*, *Tabellaria*, and *Stauroforma* species. Splash pool communities at Isle Royale were characterized by epilithic diatoms commonly found in the nearshore zone of Lake Superior and included *Denticula*, *Synedra*, *Delicata*, *Cymbella*, and *Eucocconeis* species that appear readily able to colonize splash pools. Splash pools also contain plankton (live free-floating) species found in Lake Superior including *Cyclotella*, *Discostella*, and *Ulnaria* species. The diatom communities at Apostle Islands and Pictured Rocks were not as clearly differentiated among pool zones compared to Isle Royale.

Mapping

Mapping along Isle Royale's southern shoreline, between Malone Bay and Passage Island, included all pools along what is likely the most dense and important components of this habitat in the park. Other areas of Isle Royale have either steep gradients (along the north shore) or conglomerate bedrock (along the south shores in the west half of the park), with both likely to limit pool formation. Mapping gave a "snapshot" of habitat availability and amphibian occupancy during site visits, with basic pool dimensions, location on shoreline, and pool permanence included in the database. Invertebrates were not included in mapping because of the challenges and time that would have been required to effectively search for, capture, and identify taxa. A total of 71,931 pools were mapped along a 48-kilometer (30-mile) stretch of shore, with 45,164 (63%) occurring on Passage Island. Chorus frogs were the most common inhabitant, occupying 2,114 pools (3%), mostly on Passage Island and in the Edwards Island vicinity. Blue-spotted salamanders had the broadest geographic range, occupying 945 pools (1%) regularly spread along the entire southern shoreline.

A monitoring protocol is included as an appendix to this report, and a geodatabase was created from the mapping work at Isle Royale. We hope that the geodatabase will be helpful for enhancing coastal resource protection from threats such as shipping-related oil spills, and to help understand any future habitat changes. Understanding amphibian use of pools is extremely important, and range maps are provided in the appendices, but analysis of amphibian populations was outside the scope of this report.

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List of Terms and Acronyms

Ephemeral: A pool with >2 cm (one inch) but ≤10 cm (4 inches) of water. These numbers are based on pilot work in 2009, which suggested that pools deeper than approximately four inches would retain water during a period of no rain lasting up to two weeks, while pools of less than an inch are likely to dry with only a few days of sunny weather.

Forest Transition Zone: Typically a distinct forest edge exists along the rocky shores, but sometimes vegetation such as stunted cedar trees extend into the rocky areas. When a pool juts into the forest or vegetation line it should be included in this zone. If a pool is adjacent to the forest edge and heavily lined with vegetation, or within an area dotted with cedars, include it in this zone. If a pool is completely inside the forest edge, it should not be included as a shoreline pool.

Lichen Zone: The area of shoreline between the splash zone and the forest edge. This zone contains lichens that typically cover all or most of the open rock and are generally colorful (orange, pale green, white, gray). If lichens generally surround a pool (sometimes they are less dense on the downslope side) the pool should be included in this zone. Patches of flora may occur in crevices or on the edge of pools in this zone.

Permanent: A pool with >10 cm (0.1 m, or 4 inches) of water.

Seep or Medicolous Habitat: A water source for a pool that originates from cracks in the bedrock or slow/intermittent running surface water. This does not include streams or strongly flowing surface water. Seeps may not be obvious if there has been no recent rain, but look for stains that indicate seepage through cracks. Medicolous pools may be distinguished from seeps by near constant inputs of groundwater and little variation in diurnal or seasonal temperature, shallow depth, and possible current patterns.

Splash Zone: The area of shoreline between the water and a line where lichens are common. The splash-lichen line is generally very distinct, with colorful foliose or crustose lichens above, and no lichen or only black, crustose lichens below. Sometimes patches of colorful lichens gradually blend into the dark portion of the rocky shore; if so, count a pool in the splash zone if lichens are not present on the downslope side. If a pool is generally not surrounded by colorful lichens it should be considered in this zone. Very little flora is present in this zone.

APIS: Apostle Islands National Lakeshore (Wisconsin)

DCA: Detrended Correspondence Analysis

DIC: Dissolved inorganic carbon

DIN: Dissolved inorganic nitrogen; sum of nitrate, nitrite, and ammonium.

DIN:TP: Ratio of dissolved inorganic nitrogen to total phosphorus.

DO: Dissolved oxygen

DOC: Dissolved organic carbon

ISRO: Isle Royale National Park (Michigan)

NH_4: Ammonium ion

NMDS: Non-metric multidimensional scaling ordination

NO_3-NO_2: Sum of nitrate-nitrite, also expressed as NO_X.

NO_X: Sum of nitrate-nitrite, also expressed as NO_3-NO_2.

PCA: Principal Components Analysis

pH: A measure of the acidity of water.

PIRO: Pictured Rocks National Lakeshore (Michigan)

SID: Simpson's Index of Diversity

SRD: Simpson's Reciprocal Diversity

SFPE: Surface-floating pupal exuviae (the shed pupal exoskeleton following emergence of an adult insect).

SRP: Soluble reactive phosphorus

TN: Total nitrogen

TP: Total phosphorus

TN:TP: Ratio of total nitrogen to total phosphorus

Introduction

Coastal shorelines contain habitats at the transition of two very different systems: aquatic and terrestrial. Some shorelines facing large bodies of open water have wide areas of bedrock inhabited by hardy species that are able to survive in this challenging transition zone. In freshwater systems such as Lake Superior in the Laurentian Great Lakes, important features of these rocky shoreline habitats include small rock pools fed by spring snow melt, rainfall, and wave wash (Figure 1). These discrete pools are the freshwater equivalent of marine tidal pools, which have been well-studied worldwide and have led to an understanding of key principles of ecological theory (Paine 1966). Coastal freshwater rock pools appear to share many attributes with marine pools (Arnér 1997, Murray et al. 2002), inland rock pools (Baron et al. 1998, Jocque et al. 2007), and vernal pools (Keeley and Zedler 1998, Colburn 2004). Also similar to marine coasts, Lake Superior shorelines often have strong heterogeneity of disturbance and pool presence both along the shore-to-forest gradient and parallel to shorelines (Murray et al. 2002), allowing for numerous niches. Coastal habitats are directly exposed to climatic extremes, with ice scouring in winter and high winds, full sun exposure, and frequent wave splash in all seasons.

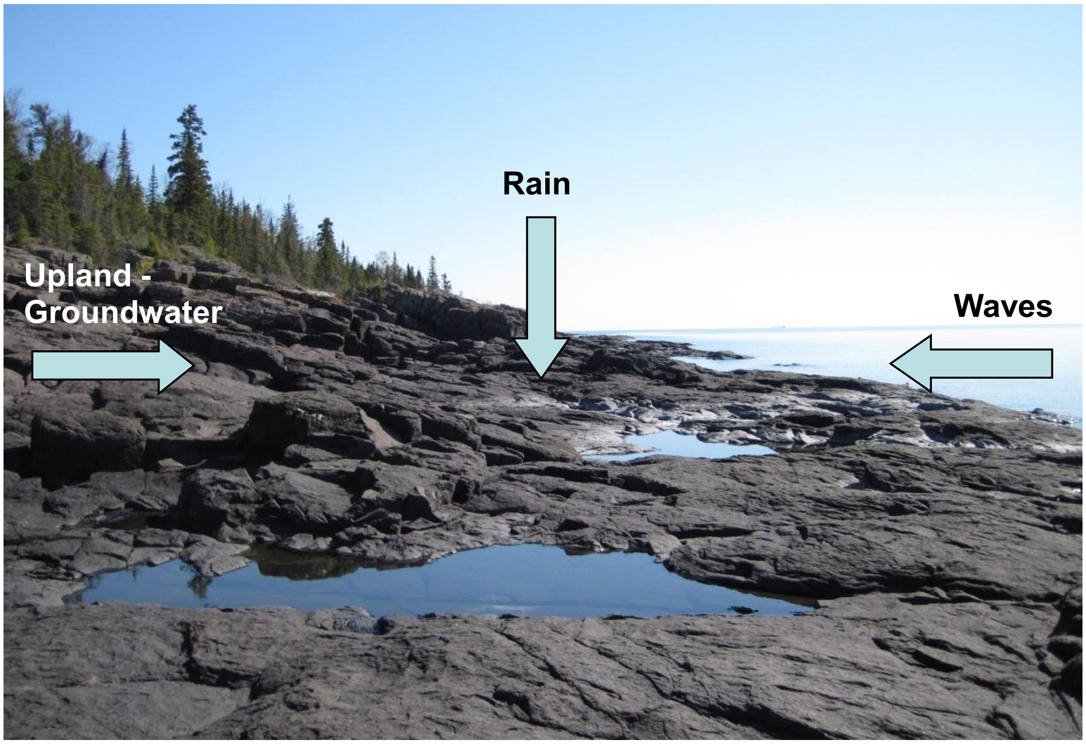

Figure 1. Recharge sources for coastal rock pools at Lake Superior national park units.

Important characteristics of these pools include height above the shoreline (delineating pools that primarily receive rainfall recharge from those that also receive lake splash recharge), water chemistry (e.g., lower pools, with regular wave splash, likely have higher dissolved oxygen), and depth or hydroperiod (shallow pools may become dry during a drought, while deeper pools will remain partially full) (Jocque et al. 2007, Vanschoenwinkel et al. 2009). Aquatic invertebrates are key components of rock pool communities, forming the majority of animal taxa present (Arnér 1997), and are often well-suited to survival despite the unpredictable and harsh nature of this habitat (Baron et al. 1998). Primary producer communities, especially the diversity of algal communities, have been much less studied in rock pool habitats (Arnér et al. 1998). Intuitively, the cold climate, limited physical structure, and low nutrient conditions in Lake Superior and its rock pools should limit diversity and abundance of aquatic organisms.

Organisms in Pools of Coastal Habitats

Shorelines of large bodies of water, such as Lake Superior, often have a colder average local climate than inland areas due to the moderating influence of the lake's large surface area, thermal mass, and depths that can reach well below sea level. Cold-tolerant taxa that are expected to exist in locally cold shoreline habitats may have the capacity to act as indicator species for local climate variations or habitat stressors (Kerr et al. 2000, Luoto 2010, Parsons et al. 2010). These taxa may be part of a disjunct population whose range is separated from that of the main population. They would likely be the first to disappear if warming trends continue, and if their primary range includes arctic or alpine areas, there would be little opportunity for recolonization (Vanschoenwinkel et al. 2008).

Regionally rare plants such as knotted pearlwort (*Sagina nodosa*), butterwort (*Pinguicula vulgaris*), and three-toothed saxifrage (*Saxifraga tricuspidata*) are examples of species typically found in arctic or alpine areas but present along the shoreline of Lake Superior due to the cold climatic regime existing since the last glaciation event (Judziewicz 2004). As with cold-tolerant plants that occupy coastal habitats, it is likely that there are also rare, cold-tolerant, disjunct invertebrates.

At Isle Royale National Park (ISRO), Michigan, a long-term study of boreal chorus frog (*Pseudacris triseriata*) ecology is one of the few studies of rock pool systems in the region (Smith 1983). It appears that chorus frogs in the park only occupy coastal rock pools for breeding, while presumably using inland forests only in the vicinity of breeding pools. From 2002–2006, no chorus frogs were detected inland during frog surveys (Egan 2006). Pilot work for this study suggested use of rock pool habitat by other amphibian species as well.

Invertebrates are typically understudied components of ecological systems, and coastal habitats in Lake Superior parks are no exception. A study of the sedge darner dragonfly (*Aeshna juncea*), which is a predator in rock pools during its larval stage, is the only extensive aquatic macroinvertebrate investigation at ISRO (Van Buskirk 1993). The studies by both Smith (1983) and Van Buskirk (1993) were limited to a small number of islands, restricted to mid-summer months, and did not comprehensively address aquatic invertebrate communities. Therefore, the general community structure, diversity, and seasonal activity of aquatic macroinvertebrates on ISRO and in other Lake Superior coastal rock pools are not known.

During a limited study on two small islands at ISRO, Art (1993) identified nine genera of non-biting midges (Insecta: Diptera: Chironomidae) whose presence seemed to be stratified based on persistence of pools (i.e., desiccation during drought) and location on the shoreline gradient (proximity to either forest edge or waterline). Chironomids are likely much more diverse than other macroinvertebrate taxa and may constitute much of the invertebrate biomass in pools. Fox (1995) found protozoa responding to a change in shoreline gradient, from lake to forest, while rotifers did not show discernible change. Wave wash disturbance was suggested as the mechanism for the change in rotifers. No studies to-date have examined the primary producer communities or water chemistry of Lake Superior rock pools. A limited number of pools on ISRO's Edwards and North Government islands have been physically characterized for size, depth, detrital content, and pool permanence during studies of tadpole survivorship (Smith 1983), use of pools by salamander larvae (Van Buskirk and Smith 1991), and invertebrate diversity (Art 1993, Fox 1995). Similar studies have not occurred in the Apostle Islands or at Pictured Rocks.

Threats to Coastal Rock Pools

Pollution

Lake Superior coastal areas are susceptible to pollution resulting from shipping accidents, with cargo vessels carrying very high fuel capacities (Gertler et al. 2010). Following devastating accidents and spills internationally, coastal land managers have become aware of variable ecosystem responses and the importance of baseline data regarding coastal resources (Peterson et al. 2003, Diez et al. 2009).

Multiple shipping lanes cross Lake Superior, including one that passes through ISRO waters between the main island and Passage Island (see Figure 2). This shipping lane cuts through an area of the park that has been identified as containing highly sensitive coastal habitats and species (Schaefer et al. 2004). Shallow reefs surround ISRO, and many shipwrecks lie within park waters. The risk of running aground has decreased since the advent of Global Positioning Systems and radar technologies, but 800–1,200 ships pass through park waters annually, and one has run aground at ISRO in recent years (Rayburn et al. 2004). Both Apostle Islands National Lakeshore (APIS), Wisconsin, and Pictured Rocks National Lakeshore (PIRO), Michigan, have similar immediate threats from shipping traffic, with traffic between Sault Ste. Marie and ports in Minnesota and Wisconsin.

In conjunction with the U.S. Coast Guard (USCG) and the Environmental Protection Agency (EPA), ISRO has a strategy in place to respond to shipping accidents, which may involve substantial volumes of fuel or oil (Rancilio et al. 2004, Schaefer et al. 2004). The strategy stresses that critical habitats be identified and strategically protected based on known criteria. However, while the degree of impact to particular shoreline habitats and macrophyte populations has been ranked as potentially critical or catastrophic (Schaefer et al. 2004), the response plan has virtually no data on aquatic invertebrate communities. Pre-impact data are important for comparative studies since invertebrates of highly unpredictable systems such as rock pools show variable tolerance to pollutants (Arnér 1997). Due to limited on-site resources in parks and potentially lengthy response times for USCG resources, spill responses will often require a triage perspective, which relies upon solid evidence during decision making. Results from this study will be provided to both the USCG and National Park Service (NPS) for integration into their response plans.

Climate Change

The director of the National Park Service, Jon Jarvis, believes that “climate change is fundamentally the greatest threat to the national parks that we have ever experienced” (Saunders et al. 2011). Rock pools have a potentially important role as “sentinels” for determining regional effects of global climate change, linking variations in precipitation and evaporation with ecological repercussions (Hulsmans et al. 2008). Expected effects of climate change in the Lake Superior region include warmer temperatures and less precipitation (Kling et al. 2003), decreasing the amount of habitat available for aquatic organisms as pools become dry due to increased evaporation and lack of recharge. An expected drop in lake water levels will shift the patterns of wave-recharged pools, stranding many pools in a zone requiring rainfall, while potentially adding some habitat along bedrock with shallow gradients.

Because of its ability to store solar energy, Lake Superior surface water temperatures have been increasing at a faster rate than regional air temperatures, resulting in less winter ice cover, a feedback of even more solar energy stored in the lake, and the likelihood of significant future ecological impacts (Austin and Coleman 2007, Schindler 1997). If nearshore habitats become warmer, without the cool climate created by cold surface water, extirpation of cold stenothermic species could occur, while allowing greater domination by species that prefer moderate temperatures. Increased wave height due to warmer lake temperatures that create greater turbulence within the atmosphere (Austin and Coleman 2007) may increase both recharge to and disturbance of lower pools.

Changing temperature patterns may also cause changes in visitation patterns to parks (Saunders et al. 2011), such that rocky shore habitats in locations with regular foot traffic could be directly affected by human disturbance (Murray et al. 2002). Such locations are rare at ISRO, but more common at APIS, PIRO, and many state and local parks along the Minnesota coast where rock pool habitat can be abundant (A. Egan, pers. obs.). Additional impacts include a heightened risk of successful dispersal and establishment of invasive species (Saunders et al. 2011).

Current Study

Documentation of ecological communities is important for tracking changes caused by both small- and large-scale disturbances. Loss of species, including potential extirpation of rare and sensitive taxa, reduces an ecological community in important ways. Losses may create imbalance in a food web (Townsend et al. 2010) and increase susceptibility to invasive species (e.g., Riley et al 2008). Recently, federal land managers in the Lake Superior region identified coastal studies, including those of rock pool habitats in particular, as a critical gap in knowledge of aquatic resources (Lafrancois and Glase 2005, Crane et al. 2006).

Comprehensive biological and ecological assessments presented in this report will inform managers of the current state of coastal habitats, giving targets for remediation if those habitats become polluted or stressed (Tokeshi 1995, Lytle and Peckarsky 2001). The tools presented in the monitoring protocol (Appendix F) will help managers monitor the condition of coastal aquatic habitats. Methods are transferable to coastal protection from additional pollution sources such as municipal wastewater or industrial effluents. The intensive sampling and detailed protocols presented in this report are a firm starting point for long-term monitoring and research so parks and other coastal land managers have a better understanding of these communities and their likely responses to climate and pollution impacts.

Objectives

This project provides a concerted assessment of rock pool natural resources, including identifying, mapping, and characterizing pools. It is our hope that this assessment will be used to help define policies and best management practices for protecting aquatic communities, and that it will inform messages to the public about the important role these communities play.

The following objectives involving mapping and biological studies were developed to meet the information needs of several Lake Superior national parks in responding to threats of management concern, such as shipping-related spills and climate change:

- Characterize biological, physical, and chemical conditions in rock pools, with a focus on water quality and macroinvertebrate, zooplankton, and diatom communities.
- Document community composition and phenology of rock pool communities through the ice-free period.
- Identify organisms in various taxonomic groups that are rock pool obligates, arctic disjuncts, or otherwise rare or unique within the park unit or the Lake Superior region.
- Develop a protocol for long-term monitoring of rock pool resources by coastal land management agencies in the Great Lakes region.
- Map the distribution of rock pools at Isle Royale with Geographic Information Systems (GIS).
- Determine the distribution of rock pool amphibians based on GIS mapping.

Methods

Study Areas

Three units of the U.S. National Park System were included in this study: Isle Royale National Park, Pictured Rocks National Lakeshore, and Apostle Islands National Lakeshore (Figure 2). All three are located in or along the shoreline of Lake Superior—the largest and deepest of the Laurentian Great Lakes. The volume, depth, and surface area of Lake Superior have a considerable influence on local climate, with a cooling effect in the summer and a warming effect in the winter. Surface water temperatures have an annual mean of 4 °C (39 °F), yet the entire lake rarely freezes over in the winter.

Isle Royale National Park

Isle Royale National Park (Michigan), in the northwest portion of Lake Superior, is an archipelago of one large island (544 km^2, or 210 mi^2) surrounded by many hundreds of small islands. The nearest point to the mainland is approximately 19 km (12 mi) distant. Bedrock formations include the Precambrian Copper Harbor Conglomerate on the west end of the archipelago, which is composed of combined gravel and sand deposits, and the Precambrian Portage Lake Volcanic on the east end of the archipelago, that is made of numerous basaltic and andesitic lava flows (Thornberry-Ehrlich 2008). All study sites are located on Portage Lake Volcanic bedrock. Forests dominate the islands, with mixed boreal tree species such as white birch (*Betula papyrifera*) and white spruce (*Picea glauca*) in the east half of the park, and a stronger hardwood component of sugar maple (*Acer saccharum*) and yellow birch (*Betula alleghaniensis*) in the west half of the park (McInnes et al. 1992). Disjunct or threatened plants that occupy rocky coastal habitats include pearlwort (*Sagina nodosa*), hoary whitlow-grass (*Draba incana*), *Tofieldia pusilla*, and three-toothed saxifrage (*Saxifraga tricuspidata*) (Judziewicz 1997). Based on expected abundant habitat densities and logistical support available, Isle Royale was the focal park for the current study.

Pictured Rocks National Lakeshore

Pictured Rocks National Lakeshore (Michigan) is on the southeastern shore of Lake Superior and includes over 64 km (40 mi) of shoreline. Precambrian, Cambrian, and Ordovician sandstone have been shaped by glaciation and subsequent erosion to form cliffs (Blewett 2012). Sand from erosion and glacial till has formed bluffs, beaches and dunes along the lakeshore. Much of the sandstone coastal areas at PIRO have near-surface groundwater seeps from forest soils at their edges that create unique formations. Forests generally consist of hardwoods such as beech (*Fagus americanus*), maples (*Acer rubrum* and *A. saccharum*), and yellow birch (*Betula allegheniensis*), but conifers such as hemlock (*Tsuga canadensis*), white pine (*Pinus strobus*), and jack pine (*P. banksiana*) are occasionally dominant (Loope 1991). Arctic disjunct plants that require the cool climates provided by Lake Superior include black crowberry (*Empetrum nigrum*), holly fern (*Polystrichum lonchitis*), bird's-eye primrose (*Primula mistassinica*), and thimbleberry (*Rubus parviflorus*) (National Park Service 2014).

Apostle Islands National Lakeshore

Apostle Islands National Lakeshore (Wisconsin), near the southwest corner of Lake Superior, is composed of 21 islands and a 19 km (12 mi) section of mainland along the Bayfield Peninsula.

Bedrock consists of Precambrian sandstone of the Bayfield Group that gives rise to cliffs and numerous sandscapes including beaches and sandspits (Kraft et al. 2007). The islands are predominantly forested and consist of hemlock (*Tsuga canadensis*), white pine, white birch, red maple (*Acer rubrum*) and other northern hardwood species at the transition between boreal and temperate forests (Judziewicz and Koch 1993). Arctic disjunct and state-threatened plants such as bird's-eye primrose, butterwort (*Pinguicula vulgaris*), spike trisetum (*Trisetum spicatum*), hair-like sedge (*Carex capillaris*), and beautiful sedge (*C. concinna*) occur on north-facing rocky shorelines (Judziewicz and Koch 1995).

Site Selection

Isle Royale National Park – 2009

ISRO was the only park unit where sampling occurred in 2009. The eastern half of the park, from the Datolite Mine area to Passage Island (Figure 2), was chosen because the volcanic basalt bedrock favors pool formation and permanence; the southern shoreline was chosen due to the longer, more gradual slopes formed by bedrock tilt (Thornberry-Ehrlich 2008). The western half of the park is generally conglomerate bedrock, where pool formation is less common, and the north shoreline often has a steep cliff aspect which would make sampling difficult and unsafe.

The island chain from West Caribou Island to Blake's Point was chosen as a more narrow focal area because of known coastal plant populations of concern (rare and disjunct taxa), which suggested the potential for similar taxa among invertebrate populations. Additionally, pool habitats could be safely and regularly accessed. Fifteen sites were selected systematically, with regular spacing between sites and no duplicates except for the main island, which had two sites on peninsulas (Scoville and Blake's points) (Figure 2). Sample pools at each study site were not chosen randomly. Generally, a section of shoreline was hiked and a location that had good pools for sampling was used for collections. As a result, 2009 data are representative of the chain of islands and peninsulas between West Caribou Island and Blake's Point, but may not be representative of other locations at ISRO.

Safe boat landing or access points were determined during the initial visit. Sites with numerous pools in both the lichen and splash zones were chosen for sampling, and generally the first suitable site near the landing was chosen as the sample pool. At some locations, such as Davidson Island, Shaw Island and Blake's Point, sampled sites were the only areas with pool habitat available. At other locations, such as Edwards Island, Raspberry Island and Scoville Point, much of the shoreline had suitable habitat. Spatial distance from other sites was also considered; for example, the east end of one island and the west end of the adjacent island would not have been considered appropriate choices, even though sites would have been separated by Lake Superior. See Appendix A for maps and descriptions of sites.

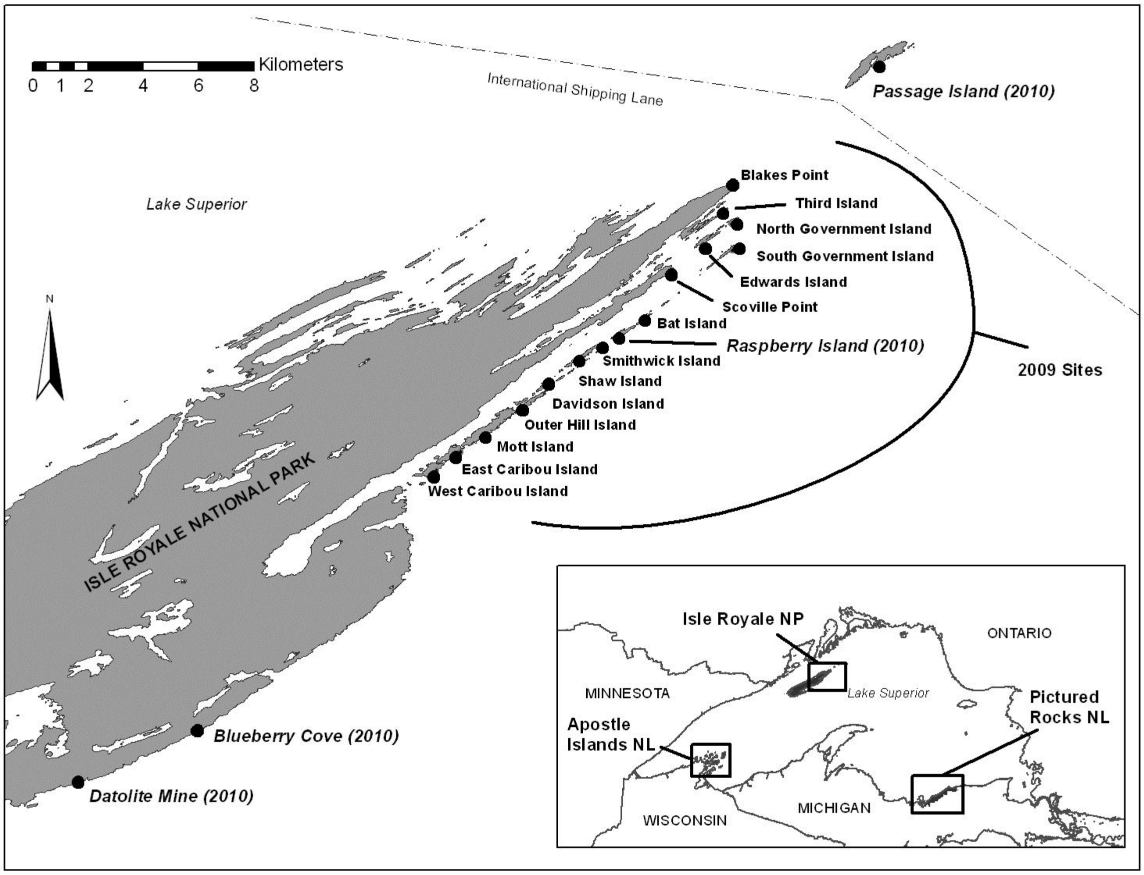

Figure 2. Study sites at Isle Royale National Park, Michigan. Sites from 2009 span the area between West Caribou Island and Blake's Point. Four sites were added in 2010. Only Raspberry Island was included in both years.

Isle Royale National Park – 2010

In 2010, site selection was made using a combination of high-definition aerial imagery (from overflights made in spring 2009) and site visits. Aerial photographs for the area along the south shore between Malone Bay and Passage Island were used in spring 2010 to assess potential site quality for rock pool habitat. All seemingly high-to-moderate-quality sites (n = 16), based on observed pool densities from photographs, were placed in three geographic strata: sites on the main island (Isle Royale) between Malone Bay and Saginaw Point, sites on nearshore islands or peninsulas between West Caribou Island and Blake's Point, and sites on Passage Island. Within these strata, four sites were randomly selected, with at least one site per stratum. Each site was visited to ensure adequate pools were present in both the lichen and splash zones, along with adequate pools based on hydroperiod (permanent and ephemeral). Criteria for rejecting a site included: inadequate pools, unsafe conditions for landing a boat, and presence of sensitive or breeding wildlife. Of the initial sites, only one (Flag Island, located south of Edwards Island) was rejected due to a limited number of lichen zone pools and the presence of a gull colony. All sites were also required to be at least 3.2 km (2 mi) from other sites.

Prior to visits, each site was divided into shoreline sections that were randomly selected for pool searches. During site visits, a subset of four permanent pools were selected for sampling by walking the shoreline in the predefined area, listing permanent pools, and randomly choosing from among these pools using dice. At each site, two permanent pools in the lichen zone and two permanent pools in the splash zone were chosen in this manner. Geographic coordinates were recorded and small rock cairns were placed next to each pool to aid in finding them on subsequent visits. Ephemeral pools for each site were those in the vicinity of the permanent pools (see List of Terms and Acronyms for definitions of permanent and ephemeral pools). Because of randomization in site selection, 2010 sampling should be representative of the study area as a whole (from Datolite Mine to Passage Island).

Apostle Islands National Lakeshore – 2010

High-definition aerial imagery was not available for APIS during planning stages for field work. Site suggestions by NPS Great Lakes Network staff, using known shoreline geology, existing maps, low-definition aerial imagery and prior staff experience, were the basis for scouting in early May 2010.

Pools observed at APIS occurred in limited numbers, often on inaccessible cliffs. Sites were only selected if a cluster of pools was available, and Bear, Devils and Stockton islands were the only areas found via boat that had large groups of pools together (Figure 3). Upon closer investigation, each had acceptable sites for sampling. . Unfortunately, full sampling did not occur at Stockton Island due to mostly dry lichen-zone pools in May 2010 and poor weather during the fall sampling. Sites on Bear and Devils islands had enough pools (multiple pools available in both splash and lichen zones) that the sampling regime from ISRO could be used. Extremely dry fall and winter conditions preceded the May 2010 sampling at APIS, resulting in many potential rock pool sites being dry, including a promising site at Stockton Island. A wider, land-based search for pool clusters should occur if managers expand monitoring of this habitat.

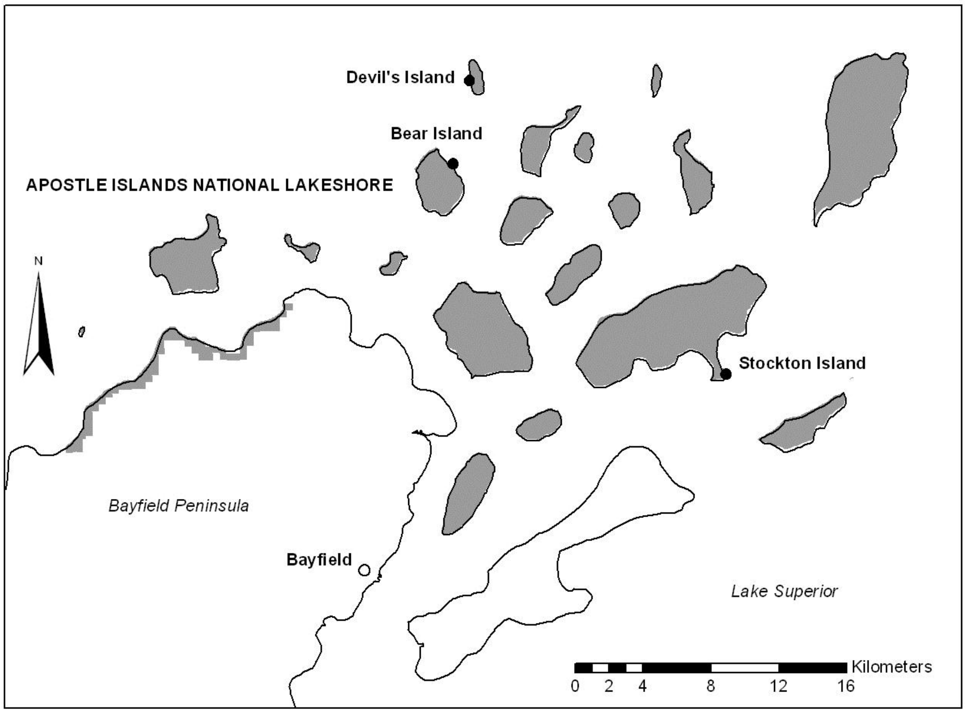

Figure 3. Study sites at Apostle Islands National Lakeshore, Wisconsin, 2010. Though acceptable sample sites were found on Stockton Island, full sampling did not occur there due to many lichen zone pools having dried out after an extremely dry fall and winter prior to the spring 2010 field season.

Pictured Rocks National Lakeshore – 2010

High-definition aerial imagery was not available for PIRO, so staff knowledge was utilized to identify potential sites with adequate rock pool habitat. Of the three sites scouted, Au Sable Point fit with the study design at ISRO, which was based on shoreline stratification of splash and lichen zones with clusters of pools available (Figure 4). The other two sites, Miner's Beach and Mosquito Harbor, had pools but they did not fit the ISRO design well. Because no other sites were known, collection modifications were made to include Miners Beach and Mosquito Harbor in the study. It is likely that these are the only clusters of coastal pool habitat available at PIRO.

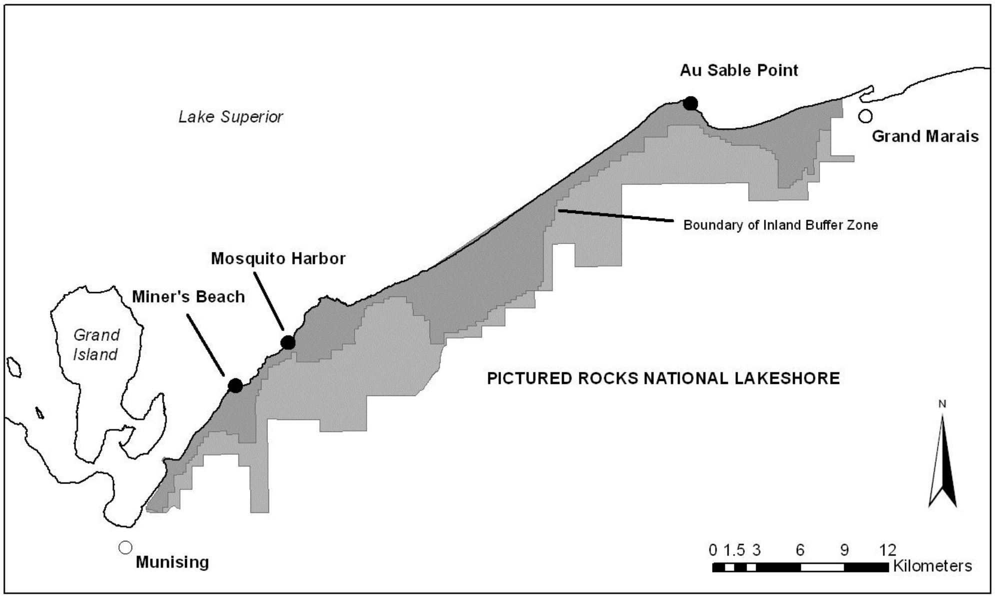

Figure 4. Study sites at Pictured Rocks National Lakeshore, Michigan, 2010.

Sample Number and Frequency

Number of Sample Sites and Samples

In 2009, 30 samples were collected at Isle Royale in mid-April, two from each of 15 sites. From May until October, two sets of pools on Outer Hill Island and East Caribou Island that had apparently filled from snow and ice melt had dried and did not refill, so only 28 samples were collected during each of the following sample rounds. Sampling rounds spanned the entire summer, with regular collections occurring when time permitted. A sampling round began at the southwest end of the study area (West Caribou Island) and progressed northeast, a span of about 18 km (12 mi). So even though the end and beginning of two sample rounds may have been temporally adjacent, they were geographically separated (Table 1). An exception was September–October sampling, which occurred in a scattered pattern due to weather and the need to access more difficult sites when winds were calm. In 2010, six sampling rounds occurred at ISRO from four sites, a reduction from 2009 sampling. Each sampling round at the four sites was tightly clustered within a 3–9 day period. Generally, 2010 sampling was separated by one month, except for April–May due to the park just opening in mid-April, and August–October when a two-month gap occurred (Table 2).

Table 1. Coastal rock pool sample dates, Isle Royale National Park, 2009.

Round 1	**Round 2**	**Round 3**	**Round 4**	**Round 5**
24 Apr–7 May	12 May–3 June	8 June–6 July	9 July–14 Aug	1 Sept–13 Oct

Table 2. Coastal rock pool sample dates for target groups, Isle Royale National Park, 2010.

	Round 1 *22–28 April*	Round 2 *9–17 May*	Round 3 *3–8 June*	Round 4 *3–9 July*	Round 5 *3–6 Aug*	Round 6 *2–4 Oct*
Macroinvertebrates	x	x	x	x	x	x
Zooplankton		x	x	x	x	x
Diatoms		x		x		x
Water quality		x		x		x

Two sampling rounds were conducted at APIS and PIRO in 2010. Sampling occurred from 5–6 May and 14–15 September at APIS, and from 18–20 May and 23–25 August at PIRO. At both APIS and PIRO, all sampling included amphibian assessments and sampling of chironomid exuviae, zooplankton, diatoms, and water quality.

Sampling Frequency

Biweekly sampling is an ideal frequency to document chironomid communities (Coffman 1973, Bouchard 2007). However, monthly sampling between spring and fall for chironomids and other biological groups in coastal rock pools is likely to yield successful taxonomic data. The large majority of chironomid emergence occurs from mid-spring to early autumn, making April-to-September sampling ideal for detecting the majority of richness (e.g., Bouchard and Ferrington 2011, Raunio et al. 2007). Some cold stenotherms may be missed as a result, but coastal pools are likely covered in snow and ice during winter and early spring, making sampling impossible (ISRO) or unsafe (APIS, PIRO). Few of the study pools at ISRO, and none at APIS, were groundwater- (i.e., spring-) fed, so overwinter freezing is likely for all of these pools. If average winter temperatures become warmer, the patterns of ice formation, emergence and occupancy may change, requiring consideration of new sampling times.

Funds and personnel are often stretched thin in Lake Superior regional natural resource management programs, making efficient use of resources important. Sampling events that are close in time often have similar taxa, making temporally-spaced sampling events important for maximizing efficiency (Bouchard and Ferrington 2011). General surveys for diversity will require regular sampling throughout the active time for groups of interest, particularly if samples are expected to be relatively diverse. We deemed monthly samples appropriate, which has been the case in many lotic systems (Bouchard and Ferrington 2011, Raunio and Muotka 2005).

Focal Taxa

Chironomidae

Chironomidae is one of the most ubiquitous and abundant macroinvertebrate families in aquatic systems. Coupled with a high diversity, chironomids are likely to be encountered in substantial numbers in most bodies of water. Because of their sensitivity to chemistry and pollutants, the Chironomidae have been used as water quality indicators based on taxonomic variability, both at a genus and species level (Odume and Muller 2011, Luoto 2011). Some genera are able to take advantage of polluted or poor water quality conditions, while others are intolerant of pollution (Ferrington et al. 2008).

Although ecologists often define communities in terms of species (Murray et al. 2002), many aquatic invertebrates are either challenging to identify to this level or may not be described. For many Chironomidae, generic identification is acceptable for understanding basic ecological requirements and natural history (Wiederholm 1986). Exuviae were identified to genus using Wiederholm (1986) and Ferrington et al. (2008).

Non-Chironomidae Macroinvertebrates

Many other aquatic insect orders were present in the coastal rock pool community, including Ephemeroptera (mayflies), Plecoptera (stoneflies), Trichoptera (caddisflies), Odonata (damselflies and dragonflies), Coleoptera (beetles), Hemiptera (true bugs), and other Diptera (flies). These were identified to genus when possible. Collembola were included in identifications, and Hymenoptera were largely identified only to family. Non-aquatic taxa (or those that could not be confirmed as aquatic) were not included in the results. Non-chironomid aquatic groups were identified using Merritt et al. (2008), Gauld and Bolton (1988) McAlpine (1987), Downie and Arnett (1996), Headstrom (1977), and White (1983).

Zooplankton

Zooplankton include protozoa, rotifers, and small planktonic crustaceans that live in the water column. Zooplankton are primarily size-selective omnivores or algal grazers. Zooplankton provide food for each other as well as larger invertebrates, amphibians, and fish. In small systems, dynamics between amphibian and fish predators often structure the zooplankton community (Schabetsberger et al. 2006). Zooplankton directly mediate between bottom-up processes (e.g., nutrient inputs leading to algal blooms) and top-down processes (e.g., changes in fish or amphibian populations). This trophic position makes zooplankton ideal for monitoring ecological processes including changes in nutrients, climatic changes, invasive species introductions, and others (reviewed in Attayde and Bozelli 1998; Stemberger et al. 2001; Thorp and Covich 2010). More subtle ecological interactions also warrant tracking zooplankton in the rock pool systems. Cladoceran zooplankton, for example, consume *Batrachochytrium dendrobatidis*, the fungus that causes chytridiomyocis in amphibians (Buck et al. 2011).

Diatoms

Diatoms are one of the major groups of microalgae, characterized among the algae by their diplontic life history and having a two-part cell wall composed of biologically produced opaline silica. They are widely used in biological assessments, aquatic monitoring, and paleoecological studies (Smol and Stoermer 2010). Diatoms are well-suited for monitoring of rock pools because they are found in virtually all aquatic habitats (freshwater or marine), even those with ephemeral moisture; their cell wall has species-specific ornamentation that allows ready identification; they have cell division rates of 0.25–1.0 divisions per day, thus responding quickly to changing environmental conditions; and diatoms have a long history of study to assist in understanding biogeographical and ecological patterns (Hustedt 1942, Cholnoky 1968). Recent systematic and morphological work in the diatoms has resulted in major revisions and substantial increase in genus-level diversity that has allowed genus-level identifications to be informative in some ecological assessments (Hill et al. 2001, Potter et al. 2006).

Field Methods

Samples were collected at each site based on two coastal strata. The “splash zone” was an area where wave wash from Lake Superior was expected to have a dominant influence. This zone can generally be identified by a lack of lichens or only having dark (black or grey) lichens. The “lichen zone” was an area above the splash zone, where wave wash was not typical and the bedrock was generally covered in colorful lichens. In 2009, two samples were collected from each site, one from pools in each zone. In 2010, six samples were collected from each site, one from each of four designated permanent pools (two lichen and two splash pools) and one sample from ephemeral pools in each zone.

Field Measurements and Water Quality Sampling

At each pool a set of field measurements and a bulk sample of water were taken to characterize pool location, size, depth, proximity, and water quality. GPS coordinates were taken for each pool at the first seasonal sampling, and the distance (m) from the pool to Lake Superior and pool to treeline was measured. During each sampling the length (m), width (m), and maximum depth (cm) of the pool were recorded. Time and date of sampling, weather, cloud, and wave conditions on Lake Superior were also recorded. A calibrated multiprobe sonde [Hydrolab (Loveland, Colorado) or Yellow Springs Instrument (Yellow Springs, Ohio)] unit was used to measure water temperature (°C), pH, electrical conductivity (µS/cm), and dissolved oxygen (mg/L and % saturation) at each pool on every sampling date. The multiprobe was similarly used to collect the same field measures from Lake Superior at each sampling round and site. For water quality sampling, three 1-L amber polypropylene bottles (acid washed) were filled with pool water by immersion in the center of the pool, capped, and placed on ice for transport to the laboratory.

Macroinvertebrates

Three methods were used to collect macroinvertebrate insects. The surface-floating pupal exuviae (SFPE) technique for Chironomidae was modified slightly for our habitats (Ferrington et al. 1991). Chironomid exuviae remain on the water surface after the pharate adult emerges from a split in the cephalothorax. Exuviae will float for a day to a week, depending on temperature, decomposer activity, and mechanical disturbance such as rain or waves (Kavanaugh 1988). The SFPE technique involves dipping a tray beneath the surface, allowing water and surface material to flow into the tray, and then pouring the water through a sieve where material is captured. A 200 mm (7 inch) diameter sieve with a 250 µm mesh was used so that the smallest exuviae would not pass through the mesh. Trays were cut in half so that a narrow edge could be used to collect from shallow pools. Exuviae were collected from around the entire pool edge. Sieve contents were washed into a collection jar and preserved with 80% ethanol. See SOP 4 in Appendix F for details on this technique in the field. Living specimens were not collected, which limits sampling impacts to communities under consideration.

Collection of SFPE has been shown to be cost efficient, time efficient, and to more effectively collect a broader assemblage of the actual community of interest from the actual waterbody of interest

(Bouchard and Ferrington 2011, Verneaux and Aleya 1999). Exuviae can be identified to genus and often species (or at least morphospecies).

The second method for collecting macroinvertebrate insects involved using a small aquarium dip net to capture insects active within the pool such as Dytiscidae, Corixidae, Gerridae, Trichoptera, and Odonata. Collection of these groups was opportunistic and limited so that collecting was not overly influential to ecological interactions in the pools. If taxa in pools appeared similar to previously collected individuals, notes were taken to indicate likely presence of the same species. For long-term studies, reference collections of and field guides to target species could be made to limit collections while ensuring correct identifications. Collection of odonate exuviae along edges of pools, along with collection of dead aquatic macroinvertebrates, could be another method for identifying species without impacting the community.

Finally, a sweep net and aspirator were used when many flying Chironomidae or other insects were observed. Collections of active adults can be linked to exuviae on the pool, which can assist with identifications. On cool mornings, many chironomids were slow to react and could be collected with an aspirator from the lee side of rocks or structures. The sweep net was used by carefully walking backwards through the habitat, which disturbed insects, and moving the net in a figure-eight pattern. A stick or ruler was sometimes used to knock shrubs, making resting insects take flight.

Zooplankon

Sampling methods for zooplankton communities in similar studies are summarized in Table 3. Most methods used in previous studies are either destructive or not quantitative. Our methods for sampling zooplankton were developed specifically for the rock pools at ISRO, PIRO, and APIS to provide quantitative population estimates while attempting to prevent over-sampling of the smaller pools in ways that would change the community enough to bias future sampling. A 30 µm mesh plankton tow net was used for horizontal tows of nearshore Lake Superior and a few larger rock pools. Tow length was recorded for measuring the volume of water the net passed through in order to estimate zooplankton density. A 25.4 × 16.5 cm rectangular fish net was modified with a 30 µm mesh net for use in the rock pools. The number and length of sweeps of this net depended on pool size. Roughly one 1-m-long sweep was made for every square meter of pool surface area, spreading sweeping efforts evenly over the pool area to cover all microhabitats. Zooplankton were assumed to be well mixed, so the sample represents a stratified randomized design. If the water was shallower than the net height, mean water depth was estimated to calculate amount of water passing through the net. This schema was adjusted to field conditions at discretion of the lead investigators. Ephemeral pools were sampled as a zone based on timed search rather than water volume so results represent an index that is not directly comparable to the permanent study pools.

Table 3. Summary of plankton sampling methodology and results from previous rock pool studies.

Citation	Zooplankton sampling method	Other parameters measured	Results
Dodson 1987	• Non-destructive • Only sampled larger zooplankton (e.g. *Triops*) • Sampled dipteran larvae by counting 3 10 cm squares on bottom (randomly) • Collected pupae and adult invertebrates, representative samples, all other ID based on field observation	• Pool geometry • Max volume (water line) • Predator exclusion and introduction experiments (*Notonecta*, *Dasyhelea*, and *Tanytarsus*) • Desiccation experiments (re-constituted dried sediment)	• 3 distinct animal assemblages correlated to pool volume, assumed to correlate with permanence • Observed predator driven exclusion
Frisch and Green 2007	• Grab sample (250 mL)	• Basic WQ sonde	• Copepods colonize first (both with and without a sediment egg bank)
Ghilarov 1967	• 20 L water pumped out and filtered with #73 gauze (destructive sampling)	• Pool geometry • Salinity (estimated)	• Species associations in light of Hutchinsonian theories
Jocque et al. 2007	• 40 cm x 10 cm diameter tube shoved rapidly into pool, all contents pumped out through 20µm mesh, three times per pool • Kick net / D-net used for qualitative work on surrounding permanent bodies	• Pool geometry, morphometry • Basic WQ sonde	• Permanence and ammonia impacted species richness and composition • Ammonia related to primary production. • Conclude that pool communities mainly structured by permanence
Johnson 2000	• Grab sample	• Pool geometry, spatial distribution	• Constructed numerical model to predict growth limitation / stress of tidal washout and drying • Predicted distribution of stress tolerant and stress-susceptible species (esp. Dinoflagellates)
Levas 2007	• Presence/absence of macroinvertebrates observed • Sampled diel vertical migration of Daphnia using modified sampler	• Basic WQ sonde • CDOM (colored dissolved organic matter) • WQ was also tested every 3 hrs over 24 hr period to check cycling	• *Daphnia* preferred deeper parts of the pools, (night and day), • Significantly more *Daphnia* found near surface at night • Pools found in two types: green and brown. • Pool could switch states (green to brown algae)

Table 3. Summary of plankton sampling methodology and results from previous rock pool studies (continued).

Citation	Zooplankton sampling method	Other parameters measured	Results
Ranta and Nuutinen 1984	• Collected *D. magna* and *D. longispina*	• Fish presence	• Predator preference structures zooplankton communities
Ranta et al. 1987	• Vertical tows with 63 um mesh: calculations for volume.	• Experimental. Pools split with curtain and predators introduced	• Fish eliminated larger cladocerans, increased smaller zooplankton • Algae response depended upon original *Daphnia* density.
Stevens and Jenkins 2000	• Water column tube sampler (40 cm diameter), pumped out and seive through 35 µm mesh • Presence/absence	• None	• Permutations testing was better than join count statistic for n <50 • Results help focus studies on which community components are distributed non-randomly
Vanschoenwinkel et al. 2007	• Composite quantitative net tow (horizontal)	• Pool depth, riparian infuence (% bordered by plants), isolation and connectedness. • Basic WQ sonde	• Mantel tests and redundancy models show local abiotic factors most dominant (vs larger scale spatial variables) • Spatial variables only important for passive dispersers

Diatoms

Diatom distribution in the environment is controlled in part by microhabitat. Thus, we sampled a single and common microhabitat among rock pools—the flocculent detrital layer immediately above the rocky substrate on the pool bottom. Other microhabitats including plankton, rocks, and plants are also present in pools and may hold additional diatom diversity. For diatom collections at each field site, six 10-mL plastic vials were prelabelled and packaged in a small Ziploc baggie along with six disposable 3 mL plastic pipettes whose mouths had been enlarged to approximately 3 mm in diameter by trimming away the tapered tip. At each pool (lichen 1, lichen 2, splash 1, splash 2, ephemeral splash, ephemeral lichen), flocculent detrital material was sampled in 2-to-3-mL “grabs” by pipette until 9 mL of material was collected. For ephemeral pool collections, as many pools were sampled as necessary to secure 9 mL of material. All diatom samples were placed on ice and preserved with a 5% formaldehyde solution upon return to the field lab. Ten percent field replicates were also taken.

Decontamination of Gear

To avoid transporting organisms between pools or study sites, all equipment was cleaned and decontaminated both between each permanent pool and between sites. Of particular concern was potential transmission of disease agents like chytrid fungus. Ephemeral pools were numerous, and equipment could not reasonably be sterilized between each pool, but equipment was decontaminated between pool types so organisms were not transported between permanent and ephemeral pools on equipment.

Two field-decontamination methods were tested during the project. At ISRO in 2009, and APIS and PIRO in 2010, gear was submerged in a 1%–5% concentration of household bleach for 10 minutes and then rinsed in a separate bucket of tap water that was carried out as waste. Three problems with this method were identified: amphibians are known to be sensitive to bleach, there was potential for spilling the bleach solution, and it became challenging to transport a large bucket of bleach water. Therefore, a second method was used in late 2009 and 2010 at ISRO. Although time consuming, a camping stove, several gallons of tap water, and large pot were brought into the field so all equipment could be boiled for five minutes between pools. Boiling was also used to decontaminate at the end of each day, along with overnight air drying. The bleach method was used at APIS and PIRO where access to sites was easier and a large amount of rinse water could be carried.

Field protocols required that personnel avoid application of sunscreen, lotions, antibacterial soaps, and insect repellents, as these substances often have chemicals that are harmful to aquatic organisms.

Laboratory Methods

Macroinvertebrates

<u>All macroinvertebrate taxa:</u> Standard operating procedures were produced prior to all lab work. See Appendix F for details. In the lab, exuviae and other aquatic insects were picked from sample material and cleaned (Appendix F, Standard Operating Procedure [SOP] 5a). Most samples from rock pool habitats lacked large amounts of detritus, though algae could be abundant in mid-to-late summer. Material of interest was sorted into vials, separating taxa by order or family. Following cleaning, non-chironomid material was progressively separated into vials based on order, then family, then genus, attempting to reach the lowest possible resolution. Vouchers of this material were then sent to experts in each taxonomic group for confirmation of identifications.

<u>Chironomid taxa:</u> Generally the entire sample was processed and all specimens identified. Chironomid exuviae were occasionally abundant in samples, so procedures were developed for assessing the need for subsampling and for subsampling itself (SOP 5b). Subsampling of dense samples utilized a grid pattern in a tray to remove half of the specimens for identification. These methods are a modification of subsampling in stream systems (Courtemanch 1996, Vinson and Hawkins 1996), where entire samples should be processed if possible. Most samples from Lake Superior coasts are likely to have low abundances, so processing the entire sample is suggested for

diversity studies in order to increase the potential to detect rare taxa. While SOP 5b (Appendix F) applied only to chironomid exuviae for the current project, it could be used for any abundant group.

Exuviae were slide mounted in euparal for identifications and to make voucher specimens (SOP 5c) and slides were labeled (SOP 5d). Exuviae were identified under compound microscopes (10X–50X magnifications) to genus using Wiederholm (1986) and Ferrington et al. (2008).

Water Quality Analysis

We processed water quality samples in field laboratories, analyzing for chlorophyll-*a* (and its breakdown derivatives), nutrients (total phosphorous [TP], total nitrogen [TN], soluble reactive phosphorous [SRP], ammonium [NH_4], nitrate-nitrite [NO_3-NO_2]), dissolved inorganic and organic carbon (DIC/DOC), anions, cations, and metals (SOP 6). From each 3-L field sample, six samples were prepared for shipment to analytical laboratories. Chlorophyll, nutrients, and DIC/DOC samples were analyzed at the Science Museum of Minnesota's St. Croix Watershed Research Station; anions, cations, and trace metal samples were analyzed at the University of Minnesota's Department of Earth Sciences Aqueous Geochemistry Laboratory. Analysis of water quality samples used the following methods and/or instrumentation:

a) Chlorophyll: APHA Standard Method 10200 H. (Chlorophyll) and EPA Methods 445.0 (Chlorophyll and Pheophytin in Algae by Fluorescence) and 446.0 (Chlorophylls and Pheopigments in Phytoplankton by Spectrophotometry).

b) TP: Standard Method 4500-P H. (Manual Digestion and Flow Injection Analysis for Total Phosphorus) and Lachat QuikChem Method 10-115-01-1F.

c) TN: Standard Method 4500-N C. (Persulfate Method), 4500-NO3 I. (Cadmium Reduction Flow Injection Method), and Lachat QuikChem Method 10-107-04-1-A.

d) SRP: Standard Method 4500-P G. (Flow Injection Analysis for Orthophosphate), and Lachat QuikChem Methods 10-115-01-1-A (high range) or 10-115-01-1-A (low range).

e) NH_4: Standard Method 4500-NH_3 F. (Phenate Method), 4500-NH_3 I. (Flow Injection Analysis), and Lachat QuikChem Method 10-107-06-1-B.

f) NO_3-NO_2: Standard Method 4500-NO_3 I. (Cadmium Reduction Flow Injection Method), and Lachat QuikChem Method 10-107-04-1-A.

g) DIC/DOC: Standard Method 5310 C.—Persulfate-Ultraviolet or Heated-Persulfate Oxidation Method, Tekmar-Dohrmann Phoenix 8000 carbon analyzer.

h) Anions: Ion chromatography system, Dionex ICS 2000 - AS19 (4 mm) column - ASRS 300 (4 mm) suppressor - NaOH eluent generator.

i) Cations: Inductively Coupled Plasma - Optical Emission Spectrometry, Thermo Scientific iCAP 6500 dual view ICP-OES.

j) Trace metals: Inductively Coupled Plasma – Mass spectrometry, Thermo Scientific XSERIES 2 ICP-MS w/ ESI PC3 Peltier cooled spray chamber, SC-FAST injection loop, and SC-4 autosampler.

All laboratory analyses included field duplicates, lab duplicates, lab spikes, and blanks as appropriate for each analysis.

Zooplankton

Zooplankton samples were transferred to 40 mL centrifuge tubes and diluted to between 20 mL and 40 mL depending on sample density. This volume was rigorously agitated, subsampled with a 1 mL Hensen-Stempel pipette, and transferred to a 1 mL Sedgwick Rafter counting slide. This process was repeated until stable variance was achieved (Colwell and Coddington 1994). After testing up to six subsamples, it was found that two rafter cell counts were sufficient. Organisms were counted and identified on an Olympus BX50F4 Microscope at several levels of magnification ranging from 40X to 400X. Standard and regionally appropriate identification keys were used (Stemberger 1979; De Melo and Hebert 1994; Lee et al. 2000; Taylor et al. 2002; Thorp and Covich 2010).

Diatoms

Diatom samples were preserved with a 5–10% formaldehyde solution upon return from the field sites (ca. 10 drops of 40% formaldehyde solution to 9 mL of field sample). To prepare diatom samples for analysis, material in a sample vial was homogenized by gentle shaking, and a 1 mL subsample was then transferred to a 50 mL snap-cap centrifuge tube. Removal of organic matter followed procedures established by Renberg (1990) and Ramstack et al. (2008a) by adding 10 mL of a 30% hydrogen peroxide solution, allowing the material to begin oxidizing overnight, and then heating the centrifuge tubes at 85°C for 3 hrs. After cooling, diatom material was rinsed five times with distilled water using centrifugation (3500 rpm for 6 min) between rinses. Subsamples of the cleaned diatom material were distributed on 22 mm × 22 mm No. 1 coverglasses, and the coverglasses were permanently mounted on microscope slides using Zrax (MicrAP, Philadelphia, Pennsylvania) high refractive index mountant (Ramstack et al. 2008b).

Slides were examined at 1000X with an Olympus BX51 compound microscope using differential interference contrast and oil immersion optics (Numerical Aperture 1.4). A minimum of 500 diatom valves were counted along one or more random transects on each slide. Only whole valves of diatoms were counted, as non-living diatoms have been shown to fragment and degrade quickly in natural settings (Kingston et al. 1983). Chrysophyte cysts (a heavily silicified resting structure produced by some species in the algal groups Chrysophyceae and Synurophyceae) were counted in addition to diatoms; however, cyst morphotypes were not counted separately. For this preliminary survey of rock pool diversity, diatoms were identified to the genus level, and analyses were limited to the permanent splash and lichen pools; samples from the ephemeral pools will be analyzed in the future. A few diatom genera were subgrouped further to capture additional ecological gradients (e.g., separation of planktonic and benthic *Fragilaria sensu lato*). Identification of diatom genera followed Spaulding et al. (2010).

Specimen Archiving and Storage

Ownership of all specimens remains with the National Park Service. Because parks generally do not have adequate facilities or staff knowledge for long-term storage and curation of specimens, macroinvertebrate material will be catalogued and housed at the University of Minnesota Insect Collection (UMSP). This arrangement, based on collection permits approved by ISRO, APIS, and PIRO, allows professional curation, along with accessibility of material to other scientists and future QA/QC. Voucher specimens for macroinvertebrates (both macroinvertebrates preserved in ethanol and slide mounted Chironomidae exuviae) will be catalogued and managed by the museum curator and director. Non-voucher material preserved in ethanol will be labeled and added to the Department of Entomology teaching collection, which is used in classroom taxonomic instruction and training. Non-voucher exuviae will be stored in the lab of Dr. Leonard C. Ferrington, Jr., and will be returned to respective parks if retention of material is no longer feasible. As described in collection permits, any non-voucher material that is not wanted by either the park of the university will be safely disposed based on university standards. ISRO Chironomidae data are part of an electronic database submitted to the National Park Service; macroinvertebrate and chironomid data from other parks are only included in this report.

Zooplankton were preserved in 80% ethanol (EtOH) because longer term preservation requires a chemical such as formalin, the use of which requires a special hazardous material permit. Further, in the amount that would be required for this study, such a chemical presents a danger to field and laboratory personnel. Unfortunately, zooplankton in EtOH do not archive well and are expected to deteriorate by late 2013. Until that time samples are stored at Northland College, in Ashland, Wisconsin, to be near the National Park Service Great Lakes Network office (GLKN) in case confirmation of taxonomic identifications is warranted. Defining morphological characters were photographed for most species, and those photos were submitted to GLKN for placement on the public server.

Diatom material from the coastal rock pools project will be housed in the Diatom Herbarium at the Science Museum of Minnesota’s St. Croix Watershed Research Station. For all diatom samples, voucher material will include one or two permanent microscope slides containing strewn mounts of peroxide-cleaned material embedded in a high refractive index mountant (Zrax; MicrAP Enterprises, Philadelphia, Pennsylvania). All slides will be assigned unique accession numbers linking them to project, park unit, and associated field data.

Analytical Methods

The macroinvertebrate community was sampled for presence and relative abundance of Chironomidae, and presence/absence for other aquatic groups. Some analyses for chironomids included only 2010 data, with habitat stratification by pool type. Most analyses included both 2009 and 2010 data, lumping 2010 data by zone for compatibility. The area sampled at each site and techniques used in both years were approximately the same, making analyses based on shoreline zonation suitable.

Accumulation Curve, Genera Estimates, and Rarefaction

Accumulation curves show how detected taxa are accrued in pooled samples over time. Estimators can be used to predict the true number of taxa based on the accumulation curve and how many were not detected. The classic Chao 1 curve (Equation 1), with 95% log-linear confidence intervals (Equation 2), is an estimator of true richness based on the rare taxa in an assemblage (Colwell and Coddington 1994). The estimator takes into account relative abundance and performs well on data that include many uncommon or rare species (Chao 1984), which describes many invertebrate communities including coastal rock pools at Isle Royale (26% of genera were considered rare). According to Chao (in Colwell 2009), the larger of the ACE (Abundance-based Coverage Estimator) and Chao 1 should be chosen.

(1) Chao 1 = S(obs) + (a^2/2b), where S(obs) is the observed number of species in a sample, a is the number of singletons (a taxon detected with only one specimen) in the same sample, and b is the number of doubletons (a taxon detected with only two specimens) in the sample

(2) $$\text{Lower 95\% Bound } = S_{obs} + \frac{T}{K}, \text{Upper 95\% Bound } = S_{obs} + \text{TK}$$

$$\text{where } T = Chao - S_{obs}\text{, and } K = \exp\left\{1.96\left[\log\left(1 + \frac{\hat{\text{var}}(\hat{S}_{Chao})}{T^2}\right)\right]^{1/2}\right\}$$

Rarefaction creates smooth curves by resampling, without replacement, all samples in the pooled dataset (right side of curve) until each one has been included (left side of curve). This creates a curve that can be used to estimate how many taxa can be expected from a certain number of samples collected (Gotelli and Colwell 2001). We calculated the Chao 1, with confidence intervals, and Coleman rarefaction curves, with standard deviations, in EstimateS (version 8.2, Colwell 2009).

Statistical Analysis of Zooplankton Communities

Zooplankton abundances were examined for regional, local, and site differences using Analysis of Variance (ANOVA) tests performed in SYSTAT and SigmaPlot (Systat Software, San Jose, California). Comparisons between means of groups showing significant differences in the ANOVA were determined using Holmes-Sidak method at $p<0.05$. Community composition data of all taxa were analyzed using standard ordination techniques in CANOCO (Ter Braak 1995, Ter Braak and Šmilauer 1998). Principal components analysis (PCA) plotted species associations and community composition organized by site and sample period with major taxonomic group and sample site independently plotted against the same axes. Higher eigenvalues indicate larger proportions of variance explained by each axis. Species vectors increase in magnitude with distance from the origin in proportion to the amount of variation explained in that species.

ANOVA tests are uncorrected for multiple comparisons. Transformations (Log + 1) were used on abundance data. Fewer than twenty total comparisons were made, only significant results are reported. Kruskall-Wallis ANOVA on ranks were used when normality or equal variance were violated. All tests are relatively conservative and should be robust despite multiple comparisons. As a consequence of both the patchiness of species data (high standard deviation) and the conservative tests chosen, the power of the tests is low (average of 0.43, range 0.41 to 0.53). Power represents the inverse of the probability of a Type II error. The relatively high probability of missing real differences is impossible to avoid, ensuring that the significant differences detected are the most robust over all the patchy and highly variable data.

Diversity Indices

Diversity can indicate community health or resilience, and can give a sense of how communities are different from each other or how they are changing (Magurran 2004). Indices will help measure and compare the species richness and evenness in the chironomid community (Legendre and Legendre 1998).

Jaccard's Index

β diversity measures the variation in diversity along a gradient (either spatially or temporally), which can assess the distinctiveness or similarity between sites (Southwood and Henderson 2000, Magurran 2004). We used Jaccard's diversity (Equation 3) in EstimateS (Colwell 2009) to analyze pool permanence (2010 data only) and chironomid community differences between zones at all 19 sites (2009 and 2010). Jaccard's index values range between 0 and 1. Significance values from Real (1999) were used to determine both differences and similarities between communities at $P \leq 0.05$.

(3) $C_{jk} = a/(a + b + c)$, where a = shared taxa, b = taxa present only in one stratification, and c = taxa present only in the other stratification.

Simpson's Index

Simpson's Reciprocal Diversity Index (Equation 4) and Simpson's Index of Diversity (Equation 5), along with bootstrap standard deviations, were calculated in EstimateS (Colwell 2009). Simpson's Index of Diversity is generally robust, describing the probability that two random individuals from a population are different taxa (Magurran 2004), and is used here to compare assemblages across sites. Simpson's Index values range from 0–1. Simpson's Reciprocal Diversity ranges from 1 to the total number of species, and measures evenness in species distributions. The higher the value, the more evenness, while lower values indicate dominance of few or a single species.

(4) $1/D$, where $D = \sum n_i(n_i-1)/N(N-1)$, n_i = abundance of the i^{th} species, and N = the total abundance

(5) $1-D$, where D is calculated as in Equation 4

Results of zooplankton counts are presented in individuals/m^3, with diversity measures of either direct counts (number of species) or indices with no units. Diversity is expressed as species richness (un-transformed number of species), generic richness (un-transformed number of genera), SRI (Simpson's Reciprocal Index), and SID (Simpson's Index of Diversity). All four measures of diversity follow the same patterns in the analyses described below, but species richness and SRI were preferred because they exhibit the least skewed distribution as measured by kurtosis.

Statistical Analysis of Diatom Communities

Diatom counts were converted to percentage abundances relative to the total count of diatoms and chrysophyte cysts. Several statistical analyses were used to explore relationships of diatom communities among parks and among pool zones. Relationships among diatom communities from all pools were explored using Detrended Correspondence Analysis (DCA) and NMDS (non-metric multidimensional scaling ordination), which are available in the software package R (Ihaka and Gentleman 1996). Ordinations were plotted separating diatom samples by park unit and pool zone. All DCAs were run on untransformed data with rare species downweighted; the NMDS were run using Bray-Curtis dissimilarity coefficients on square-root transformed data. Using procrustes rotation for the DCA and NMDS, the significance was 0.001 with an $r=0.7$, indicating the two ordinations gave the same results, and we can be confident that the groupings or trends are not statistical artifacts of the ordination techniques. Differences in utilization of pool zones by specific diatom genera at each park were visualized using box plots (Delta Graph) and tested with the t-statistic ($p<0.05$).

Statistical Analysis of Physical and Water Quality Data

Principal components analysis was implemented in the statistical software R (Ihaka and Gentleman 1996) to explore relationships among water quality and physical variables across all parks and among pool types. For this initial characterization of rock pool water quality, ordinations were limited to physical variables (temperature, pool depth, pool size, distance to lake/treeline) and a subgroup of the more than sixty water quality measures taken (chlorophyll-*a*, dissolved oxygen, TP, TN, NH_4, NO_3-NO_2, SRP, DOC, DIC, pH, specific conductivity, and the ratios of total nitrogen to total phosphorous (TN:TP) and dissolved inorganic nitrogen to total phosphorous (DIN:TP). All PCAs were scaled (centered and standardized) and used untransformed water quality and physical data. Differences between pool types within parks were further visualized using box plots (Delta Graph). Additional water quality data generated for this study are summarized in tables presented in Appendix C.

Mapping Methods

Rock pools in the area from near Datolite Mine to Passage Island on ISRO’s south shore were mapped in 2011 and 2012. This area has the densest collection of rock pool habitat in the park. Mapping consisted of documenting the location, shoreline stratum, permanence, and size of coastal rock pools, and the presence of amphibian species in those pools, using a Trimble Juno GPS and a Terrasync software data dictionary. (See SOP 8 for details regarding preparation for mapping, field techniques, and use of the Trimble GPS and Terrasync software.)

During mapping, shorelines were walked by one or more observers. In 2011, two observers were typically mapping at the same time, both for speed and safety. Sites with dangerous access due to slippery conditions (water or loose gravel) or cliffs were not mapped, but these conditions were rare and most pools that were not directly accessible were estimated from a nearby location. For example, steep cliffs on the north side of Passage Island were carefully accessed from the forest above, and both the location and size of pools on the cliffs or shoreline below were estimated. These estimated pools could not be assessed for amphibian presence, but their challenging approach made occupation unlikely.

Mapping acted as a “snapshot” of pool habitat availability and amphibian occupancy during site visits. Many factors influence pool presence and depth (evaporation, rainfall, lake height, wave direction and height, leaks in the bottom of pools), and many of these factors are irregular, so results of the 2011–2012 work may not represent annual or average pool habitat.

Amphibians were the only group included in mapping because they were of specific interest to NPS managers, and were generally simple to detect and identify to species in adult and larval stages. A field key to larval amphibians at ISRO was created for this project (Appendix E) and could be easily modified to suit other areas in the western Great Lakes. Since amphibian presence varies with life stage (adults present in spring and early summer, larvae in mid-to-late summer), mapping took place from June to mid-August. This timeframe allowed observers to assess presence of breeding adults, eggs, or larvae, and thus whether pools were used as breeding habitat for particular species. Mapping did not occur for at least 24 hours after rainfall (except for trace rain or heavy mist/fog) because of the risk of commission—pools that drain quickly, not offering adequate habitat, may have been mapped when they should not be. In addition, mapping did not occur for at least 24 hours following moderate or large wave events along the south shoreline because of the potential to include pools in the splash zone that would quickly drain or evaporate.

Results

Chironomidae

A total of 59 genera or subgenera of Chironomidae were identified from rock pools in the three parks (Figure 5). Thirty of the taxa occurred at two or more parks, and 19 taxa occupied all three parks. Twenty-nine taxa were restricted to a single park, either ISRO (18) or PIRO (11); no taxa were restricted to APIS. Forty-six of the taxa occurred in pools at ISRO, nine of which were also collected at PIRO. Forty-one taxa were detected at PIRO, and 21 taxa were detected at APIS. All 21 of the taxa collected at APIS also occurred in pools at PIRO, and 19 of the taxa also occurred in pools at ISRO.

Due to differing sample designs and pools selected for sampling, the total number of samples taken per park differed and, consequently, differing numbers of exuviae were collected. The average number of exuviae collected per sample was 40.5 for PIRO and 21.4 for APIS. Compared with the average number of exuviae collected by Anderson and Ferrington (2012) in trout streams near Duluth, Minnesota, using identical field protocols, these averages are low and reflect the less productive conditions for Chironomidae populations in pools. The low number of exuviae collected per sample also raises a question about the effectiveness of detecting taxa. In previous studies, we have tried to collect at least 100 exuviae per sample to ensure a high probability of detecting at least the common species. Bouchard and Ferrington (2011) show that two sample events spread over a ten-week period, similar to our design for assessing pools at PIRO, will only detect approximately 30% of species occurring in streams in Minnesota.

Taxonomic composition across all three parks was dominated by Orthocladiinae, with 28 taxa in this subfamily (Table 4). Orthocladiinae was also the most abundant subfamily, comprising 73.5% of pupal exuviae. Orthocladiinae was represented by 21 taxa, or 72.4% of specimens at ISRO, 21 taxa (79.6% of specimens) at PIRO, and 13 taxa (71.1% of specimens) at APIS (Table 4).

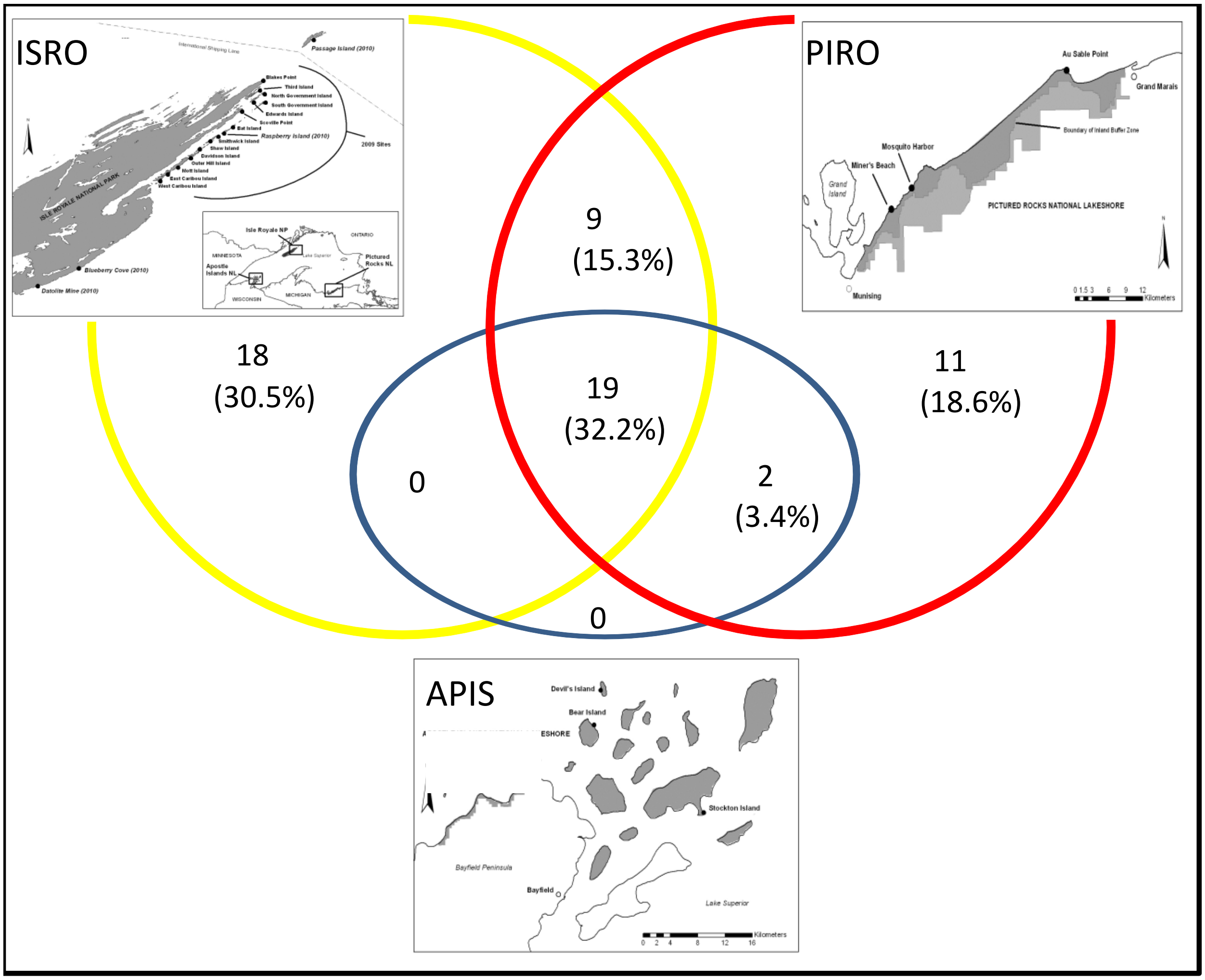

Figure 5. Numeric composition and proportional overlap of Chironomidae genera in rock pools at Isle Royale National Park, Pictured Rocks National Lakeshore, and Apostle Islands National Lakeshore, 2010–2011.

Table 4. Chironomid genera found in each park. Genera are organized by subfamily, and subgenera are shown in parentheses.

SUBFAMILY *Genus*	Park *APIS*	*ISRO*	*PIRO*
PRODIAMESINAE			
Prodiamesa		X	X
Monodiamesa		X	
Odontomesa			X
PODONOMINAE			
Parochlus		X	
TANYPODINAE			
Ablabesmyia	X	X	X
Conchapelopia		X	X
Helopelopia		X	
Labrundinia			X
Procladius		X	X
Thienemannimyia		X	
Zavrelimyia		X	
DIAMESINAE			
Diamesa	X	X	X
Pagastia		X	X
Potthastia		X	
Protanypus		X	
Pseudodiamesa		X	
ORTHOCLADIINAE			
Acricotopus			X
Brillia			X
Corynoneura	X	X	X
Cricotopus	X	X	X
Eukiefferiella	X	X	X
Heterotrissocladius	X	X	X
Limnophyes	X	X	X
Metriocnemus	X	X	X
Nanocladius		X	X
Orthocladius (*Eudactylocladius*)	X	X	X
O. (*Euorthocladius*)		X	X
O. (*Orthocladius*)	X	X	X
O. (*Pogonocladius*)		X	
Parachaetocladius			X
Paracladius		X	
Parakiefferiella	X	X	X
Parametriocnemus	X		X
Paraphaenocladius			X
Parasmittia		X	
Psectrocladius (*Allopsectrocladius*)		X	
Psectrocladius (*Psectrocladius*)	X	X	X
Pseudorthocladius		X	
Pseudosmittia		X	
Rheocricotopus			X
Synorthocladius		X	X
Thienemanniella	X	X	X
Tvetenia	X		X
Orthocladiinae genus 1		X	
CHIRONOMINAE			
Chironomus	X	X	X
Neozavrelia		X	
Cryptochironomus			X
Dicrotendipes	X	X	X
Endochironomus		X	
Glyptotendipes	X	X	X
Microtendipes			X
Parachironomus		X	X
Paratendipes			X
Polypedilum	X	X	X
Sergentia		X	
Micropsectra	X	X	X
Paratanytarsus	X	X	X
Stempellinella			X
Tanytarsus		X	X

Chironomidae – Isle Royale National Park

Subsampling

Of 285 surface-panning samples collected in 2009 and 2010, we established three categories for samples based on the need for subsampling: 1) no exuviae present (N = 38, 13%); 2) ≤20 exuviae, no subsampling needed (N = 179, 63%); 3) >20 exuviae, with subsampling protocol followed (N = 67, 24%). Of those following the subsampling protocol, only 35 (12%) required subsampling based on SOP 5b (Appendix F). One sample was not used in analyses due to an exceptionally large density of exuviae from the lowest pools, which were likely washed in from Lake Superior. In 2010, 143 macroinvertebrate and chironomid exuviae samples (one less than expected due to dry pools at Datolite in June).

Genera Richness

Forty-six Chironomidae genera (including sub-genera for the species-rich *Orthocladius* and *Psectrocladius*) were detected during 2009 and 2010 sampling (Table 5). Forty genera were detected in 2009, 37 in 2010, and 15 occurred in only one year (6 in 2010 only, and 9 in 2009 only). Sample rounds in 2010 were each within a single month, while rounds in 2009 spanned months; therefore, 2009 data in Table 5 do not come from an equivalent number of samples in each month.

Accumulation Curve and Genera Estimates

The cumulative samples (n = 246) plotted against the number of genera detected (n = 46) yields a genus accumulation curve for Isle Royale chironomids in coastal rock pool habitat (Figure 6). The Chao 1 estimator curve suggests a community of 54 genera in coastal rock pool habitat at Isle Royale, with 95% confidence intervals of 48 and 83. Fifty-four should be viewed as a low estimate (Chao 1984), even though the curve had reached an asymptote by 181 samples. Accumulation in 2009 was strongest in spring and summer, with a shallower increase in late summer and fall samples. Additions to the accumulation in 2010 were concentrated in spring and late summer, with six genera detected.

Rarefaction Curve

The rarefaction curve for expected genera detections of chironomid exuviae at Isle Royale is 33 genera detected in 50 samples (about 2 months of collecting at four sites), 39 genera detected in 100 samples (4 months of sampling), and the maximum 46 genera with 245 samples (Figure 7).

Table 5. Chironomidae genera (n = 46) identified from exuviae collected from coastal rock pools at Isle Royale National Park, Michigan. Genera are separated into subfamilies; subgenera are shown in parentheses. Forward slash separates 2009/2010 collections. Light gray fill indicates months where taxa were detected in both years.

SUBFAMILY	2009/2010						
Genus	***April***	***May***	***June***	***July***	***Aug***	***Sept–Oct***	**Totals**
PRODIAMESINAE							
Prodiamesa				0/1			(0/1) 1
Monodiamesa						2/0	(2/0) 2
PODONOMINAE							
Parochlus	0/1						(0/1) 1
TANYPODINAE							
Ablabesmyia		11/2	38/13	19/12	9/23	10/0	(87/50) 137
Conchapelopia			0/2	1/0			(1/2) 3
Helopelopia				1/3	0/1		(1/4) 5
Procladius			2/1	0/2	1/0		(3/3) 6
Thienemannimyia				1/0	0/2	6/0	(7/2) 9
Zavrelimyia		1/0		0/1			(1/1) 2
DIAMESINAE							
Diamesa	0/2						(0/2) 2
Pagastia			17/0	1/0		3/0	(21/0) 21
Potthastia			0/2			1/0	(1/2) 3
Protanypus		2/0	4/1	0/1		6/0	(12/2) 14
Pseudodiamesa			1/0	0/5			(1/5) 6
ORTHOCLADIINAE							
Corynoneura	1/5	59/8	115/3	146/12	92/29	82/31	(495/88) 583
Cricotopus		48/10	51/98	80/34	18/88	49/2	(246/232) 478
Eukiefferiella	0/5	2/13	3/2	0/1	0/10	5/2	(10/33) 43
Heterotrissocladius		5/0	41/1	0/7			(46/8) 54
Limnophyes	8/73	4/6	6/1		2/0	1/0	(21/ 80) 101
Metriocnemus	9/1	1/0		0/1	0/2	1/0	(11/4) 15
Nanocladius				2/0	0/3		(2/3) 5
Orthocladius (Eudactylocladius)	3/322	495/62	55/119	25/38	0/3	41/33	(619/577) 1196
Orthocladius (Euorthocladius)			4/0			2/0	(6/0) 6
Orthocladius (Orthocladius)		4/0	66/14	2/19	1/0	5/0	(78/33) 111
Orthocladius (Pogonocladius)						1/0	(1/0) 1
Paracladius			0/7			1/0	(1/7) 8
Parakiefferiella		0/1	170/0	3/3	1/0		(174/4) 178
Parasmittia			3/0				(3/0) 3
Psectrocadius (Allopsectrocladius)	0/1	3/2		0/19	0/3	0/5	(3/30) 33
Psectrocladius (Psectrocladius)	55/71	431/37	63/23	170/78	35/44	109/3	(863/256) 1119
Pseudorthocladius	0/1						(0/1) 1
Pseudosmittia		21/0	4/3	0/1			(25/4) 29

Table 5. Chironomidae genera (n = 46) identified from exuviae collected from coastal rock pools at Isle Royale National Park, Michigan. Genera are separated into subfamilies; subgenera are shown in parentheses. Forward slash separates 2009/2010 collections. Light gray fill indicates months where taxa were detected in both years (continued).

SUBFAMILY *Genus*	2009/2010						Totals
	April	***May***	***June***	***July***	***Aug***	***Sept–Oct***	
Synorthocladius			7/2	1/1	0/10	12/0	(20/13) 33
Thienemanniella			1/1				(1/1) 2
Orthocladiinae genus		1/0					(1/0) 1
CHIRONOMINAE Tribe Chironomimi							
Chironomus	0/3	97/17	60/11	68/16	13/2	44/2	(282/51) 333
Neozavrelia			36/39	19/0	0/16		(55/55) 110
Dicrotendipes		2/1	26/241	3/0	0/2	1/0	(32/244) 276
Endochironomus				0/1			(0/1) 1
Glyptotendipes		5/12	52/27	6/2	13/3		(76/44) 120
Parachironomus				1/0			(1/0) 1
Polypedilum				0/40	0/1		(0/41) 41
Sergentia			1/0				(1/0) 1
CHIRONOMINAE Tribe Tanytarsini							
Micropsectra	0/2	8/0	54/1				(62/3) 65
Paratanytarsus	0/1	2/0	63/125	25/24	38/51	30/1	(158/202) 360
Tanytarsus			2/0			1/0	(3/0) 3
Totals	**(76/492) 568**	**(1201/172) 1373**	**(945/737) 1682**	**(574/322) 896**	**(223/293) 516**	**(413/79) 492**	5527

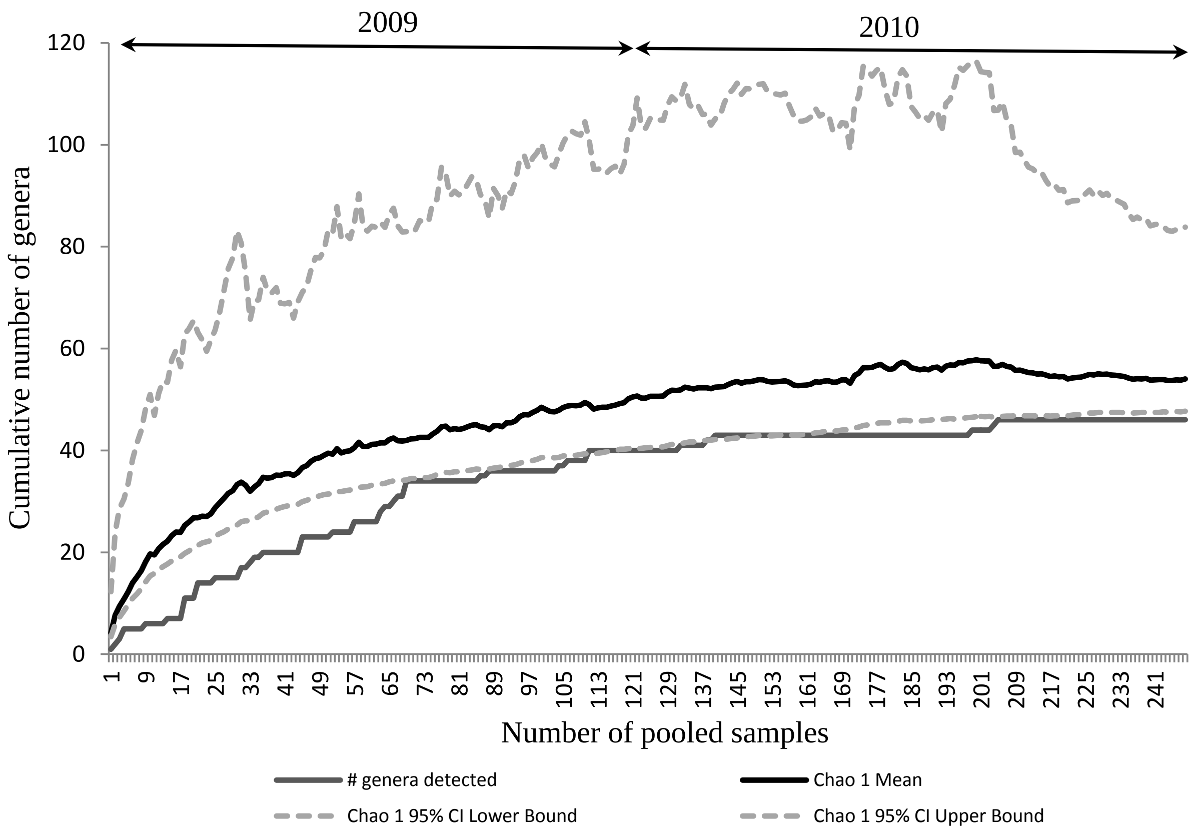

Figure 6. Pooled samples (x-axis) of Chironomidae genera (y-axis) detected, true genus richness estimate (Chao 1 mean, upper solid line), and actual number of genera detected (lower solid line) at Isle Royale National Park, 2009 and 2010.

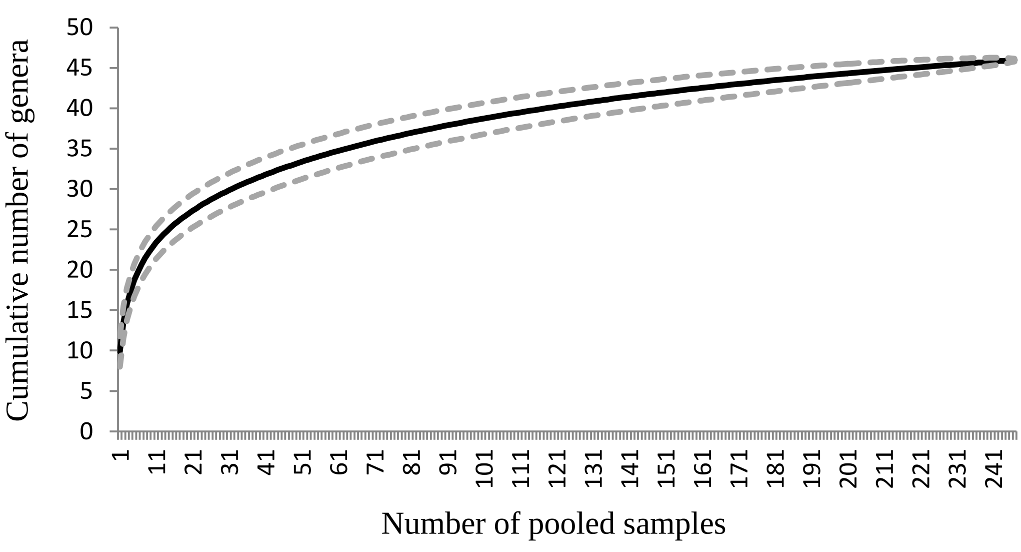

Figure 7. Rarefaction curve (solid black line) for number of genera detected for pooled samples at Isle Royale National Park, 2009-2010. Gray dashed lines are standard deviations.

Diversity Indices

<u>Jaccard's Index</u>

Jaccard's analysis does not reveal a significant community difference between zones ($P \leq 0.05$, using significance tables in Real [1999]) when all genera are included ($Cjk = 0.26$, with significance at $Cjk \leq 0.20$). Removing rare genera with only one or two individuals detected moves Cjk to 0.35. The communities between zones are also not significantly similar (with significance at $Cjk \geq 0.48$). Ten genera used the lichen zone exclusively, 24 genera used only the splash zone, and 12 genera are shared between zones (Table 6, Figure 8). However, viewing occupancy from a standpoint of preference for a zone, instead of exclusive occupancy, a significant result occurs ($Cjk = 0.15$) when 90–100% of individuals are detected in one zone. From this perspective, only seven genera do not favor a particular zone. And if the proportions are lowered to 75% of individuals favoring one zone, $Cjk = 0.04$ as only two genera (*Orthocladius* [*Eudactylocladius*] and *Corynoneura*) do not favor one zone or the other. Rare genera occurred in the lichen zone 9% of the time ($n = 4$), while rarities occurred in the splash zone 17% of the time ($n = 8$).

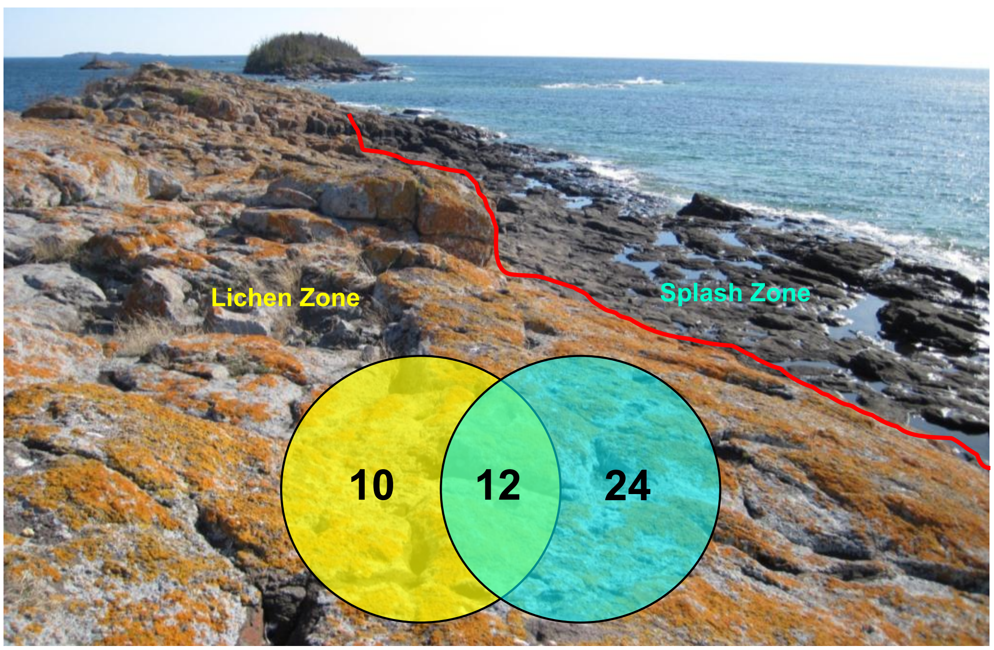

Figure 8. Chironomidae community stratification based on zones (lichen in yellow/left, splash in blue/right; red line separates the zones). Venn diagram shows number of genera exclusive to each zone and shared between zones.

Table 6. Proportional and abundance-based zonal use by Chironomidae genera in coastal rock pools at Isle Royale National Park, 2009–2010. Genera are separated into subfamilies; subgenera are shown in parentheses. Light gray fill indicates a genus only detected in one zone.

	Proportion		Abundance			
SUBFAMILY *Genus*	***Lichen***	***Splash***	***Lichen 2009***	***Lichen 2010***	***Splash 2009***	***Splash 2010***
PRODIAMESINAE						
Prodiamesa		100%	0	0	0	1
Monodiamesa		100%	0	0	2	0
PODONOMINAE						
Parochlus	100%		0	1	0	0
TANYPODINAE						
Ablabesmyia	82%	18%	73	39	14	11
Conchapelopia		100%	0	0	1	2
Helopelopia		100%	0	0	1	4
Procladius	100%		3	3	0	0
Thienemannimyia		100%	0	0	7	2
Zavrelimyia	100%		1	1	0	0
DIAMESINAE						
Diamesa		100%	0	0	0	2
Pagastia		100%	0	0	21	0
Potthastia		100%	0	0	1	2
Protanypus		100%	0	0	12	2
Pseudodiamesa		100%	0	0	1	5
ORTHOCLADIINAE						
Corynoneura	27%	73%	133	22	362	66
Cricotopus	25%	75%	74	47	172	185
Eukiefferiella		100%	0	0	10	33
Heterotrissocladius	4%	96%	2	0	44	8
Limnophyes	95%	5%	20	76	1	4
Metriocnemus	100%		11	4	0	0
Nanocladius		100%	0	0	2	3
Orthocladius (Eudactylocladius)	38%	62%	321	134	298	443
Orthocladius (Euorthocladius)		100%	0	0	6	0
Orthocladius (Orthocladius)		100%	0	0	78	33
Orthocladius (Pogonocladius)		100%	0	0	1	0
Paracladius		100%	0	0	1	7
Parakiefferiella		100%	0	0	174	4
Parasmittia		100%	0	0	3	0
Psectrocadius (Allopsectrocladius)	100%		3	30	0	0
Psectrocladius (Psectrocladius)	89%	11%	750	248	113	8
Pseudorthocladius	100%		0	1	0	0
Pseudosmittia	14%	86%	4	0	21	4
Synorthocladius		100%	0	0	20	13

Table 6. Proportional and abundance-based zonal use by Chironomidae genera in coastal rock pools at Isle Royale National Park, 2009-2010. Genera are separated into subfamilies; subgenera are shown in parentheses. Light gray fill indicates a genus only detected in one zone (continued).

	Proportion		Abundance			
SUBFAMILY *Genus*	***Lichen***	***Splash***	***Lichen 2009***	***Lichen 2010***	***Splash 2009***	***Splash 2010***
Thienemanniella		100%	0	0	1	1
Orthocladiinae genus		100%	0	0	1	0
CHIRONOMINAE Tribe Chironomimi						
Chironomus	96%	4%	270	51	12	0
Neozavrelia		100%	0	0	55	55
Dicrotendipes	99%	1%	30	244	2	0
Endochironomus	100%		0	1	0	0
Glyptotendipes	100%		76	44	0	0
Parachironomus		100%	0	0	1	0
Polypedilum	100%		0	41	0	0
Sergentia		100%	0	0	1	0
CHIRONOMINAE Tribe Tanytarsini						
Micropsectra	9%	91%	4	2	58	1
Paratanytarsus	78%	23%	101	181	57	21
Tanytarsus	100%		3	0	0	0
Annual Totals			**1879**	**1170**	**1554**	**920**
Totals			**3049**		**2474**	

Occupancy based on pool permanence was also analyzed with Jaccard's diversity for the four sites sampled in 2010. Eight genera used permanent pools exclusively, nine general used ephemeral pools exclusively, and 20 genera were shared between the two (Table 7, Figure 9). There was a significant similarity in communities between permanent and ephemeral pool types when all genera were included (Cjk = 0.54, with significance at Cjk ≤ 0.19), and when rarities were removed the level of similarity increased (Cjk = 0.77). Looking at pool use proportionally, there was no significant difference or similarity (Cjk = 0.41) in pool use when 90-100% of individuals favor either pool type. When pool use is separated into zones, the only significant result is pool use in the lichen zone, where there is a significant similarity between pool types (Cjk = 0.65 at a significance of ≥ 0.55). Rare genera did not favor either pool type, with 16% (n = 6) occurring in permanent pools and 14% (n = 5) in ephemeral pools.

Table 7. Proportional and abundance-based ephemeral and permanent pool use by Chironomidae genera in coastal rock pools at Isle Royale National Park, 2010. Genera are organized by subfamily, and subgenera are shown in parentheses. Gray fill indicates detection in only one pool type.

SUBFAMILY *Genus*	Ephemeral		Permanent	
	Proportion	**Abundance**	**Proportion**	**Abundance**
PRODIAMESINAE				
Prodiamesa	100%	1	0%	0
PODONOMINAE				
Parochlus	100%	1	0%	0
TANYPODINAE				
Ablabesmyia	20%	10	80%	40
Conchapelopia	0%	0	100%	2
Helopelopia	75%	3	25%	1
Procladius	0%	0	100%	3
Thienemannimyia	0%	0	100%	2
Zavrelimyia	100%	1	0%	0
DIAMESINAE				
Diamesa	100%	2	0%	0
Potthastia	0%	0	100%	2
Protanypus	100%	2	0%	0
Pseudodiamesa	100%	5	0%	0
ORTHOCLADIINAE				
Corynoneura	55%	48	45%	40
Cricotopus	21%	48	79%	184
Eukiefferiella	33%	11	67%	22
Heterotrissocladius	88%	7	13%	1
Limnophyes	94%	75	6%	5
Metriocnemus	75%	3	25%	1
Nanocladius	0%	0	100%	3
Orthocladius (*Eudactylocladius*)	70%	403	30%	174
Orthocladius (*Orthocladius*)	88%	29	12%	4
Paracladius	100%	7	0%	0
Parakiefferiella	50%	2	50%	2
Psectrocadius (*Allopsectrocladius*)	10%	3	90%	27
Psectrocladius (*Psectrocladius*)	39%	99	61%	157
Pseudorthocladius	0%	0	100%	1
Pseudosmittia	100%	4	0%	0
Synorthocladius	23%	3	77%	10
Thienemanniella	0%	0	100%	1
CHIRONOMINAE Tribe Chironomimi				
Chironomus	37%	19	63%	32
Neozavrelia	7%	4	93%	51
Dicrotendipes	5%	12	95%	232
Endochironomus	0%	0	100%	1

Table 7. Proportional and abundance-based ephemeral and permanent pool use by Chironomidae genera in coastal rock pools at Isle Royale National Park, 2010. Genera are organized by subfamily, and subgenera are shown in parentheses. Gray fill indicates detection in only one pool type (continued).

SUBFAMILY *Genus*	**Ephemeral** Proportion	**Ephemeral** Abundance	**Permanent** Proportion	**Permanent** Abundance
Glyptotendipes	48%	21	52%	23
Polypedilum	7%	3	93%	38
CHIRONOMINAE				
Tribe Tanytarsini				
Paratanytarsus	13%	27	87%	175
Micropsectra	100%	3	0%	0
Genera detected		**29**		**28**
Totals		**856**		**1234**

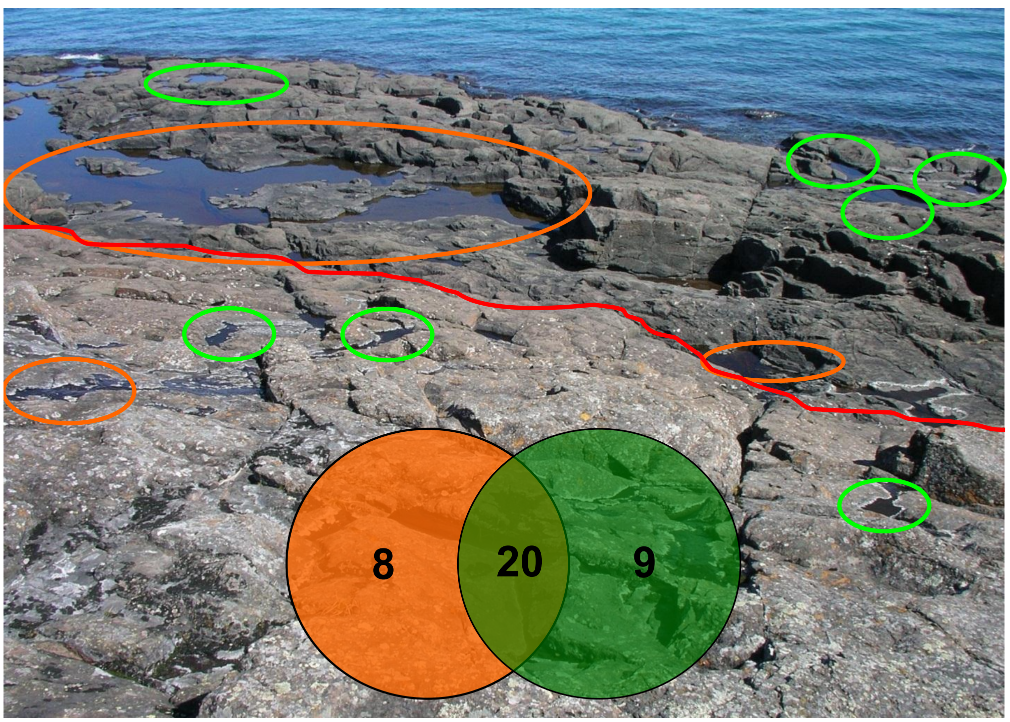

Figure 9. Chironomidae community stratification based on pool permanence (permanent in orange/left, ephemeral in green/right), with red line illustrating how samples were also separated into zones. Venn diagram shows number of genera exclusive to each pool type and shared between types.

<u>Simpson's Index</u>

Diversity for each site was also measured with genus richness and Simpson's Reciprocal Index (Figure 10). Richness was low for several sites in the western part of the Middle Islands–Blake's Point chain of islands (WC – West Caribou, EC – East Caribou, and OH – Outer Hill Island), with only 6–8 genera detected. High richness occurred at Blueberry Cove (BL) and Raspberry Island (RS) in 2010, with 25 and 27 genera detected, respectively. Simpson's Index had a generally similar pattern across sites. Comparable patterns emerge across sites when looking at individuals collected (range = 28 [at Outer Hill Island] to 711 [at Raspberry Island] in 2010) and proportion of genera detected (range = 13% [Outer Hill and East Caribou islands] to 59% [Raspberry Island] in 2010) (Figure 11).

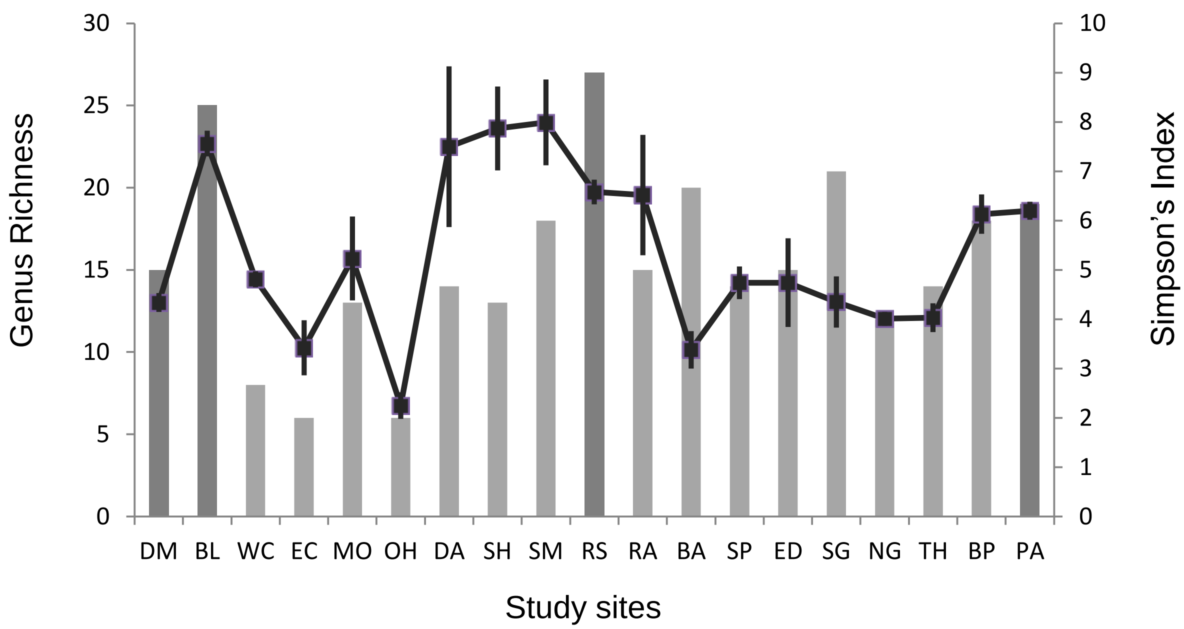

Figure 10. Chironomidae genus richness (columns) and Simpson's Index (line), with standard deviations, for study sites at Isle Royale National Park. Study sites arranged from southwest (DM – Datolite Mine) to northeast (PA – Passage Island); see Table A1 (Appendix A) for full site names. Darker columns = 2010 sites (DM, BL, RS, PA), lighter columns = 2009 sites (all others).

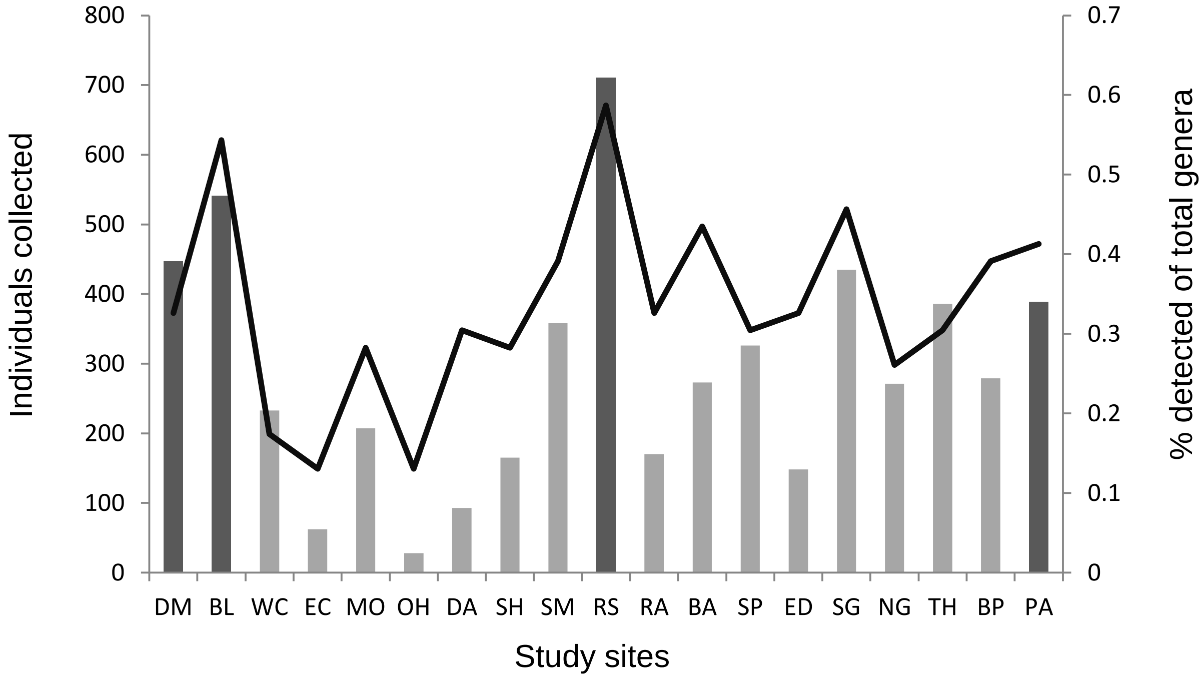

Figure 11. Total number of Chironomidae individuals (columns) collected per sample site, and proportion of total genera (line, n = 46) detected at each sample site, Isle Royale National Park. Darker columns = 2010 sites (DM, BL, RS, PA), lighter columns = 2009 sites (all others).

Collection of chironomids by month varied widely (Figure 12). The greatest number of individuals were collected in June (n = 749) and April (n = 488), while the fewest were collected in October (n = 80) and May (n = 159). The number and proportion of genera detected were both highest in July (24 genera comprising 65% of genera detected in 2010), even though moderate-to-low numbers of individuals were collected at that time. Moderate numbers of genera were detected in all other months except for October when only eight genera were detected (22% of total genera in 2010).

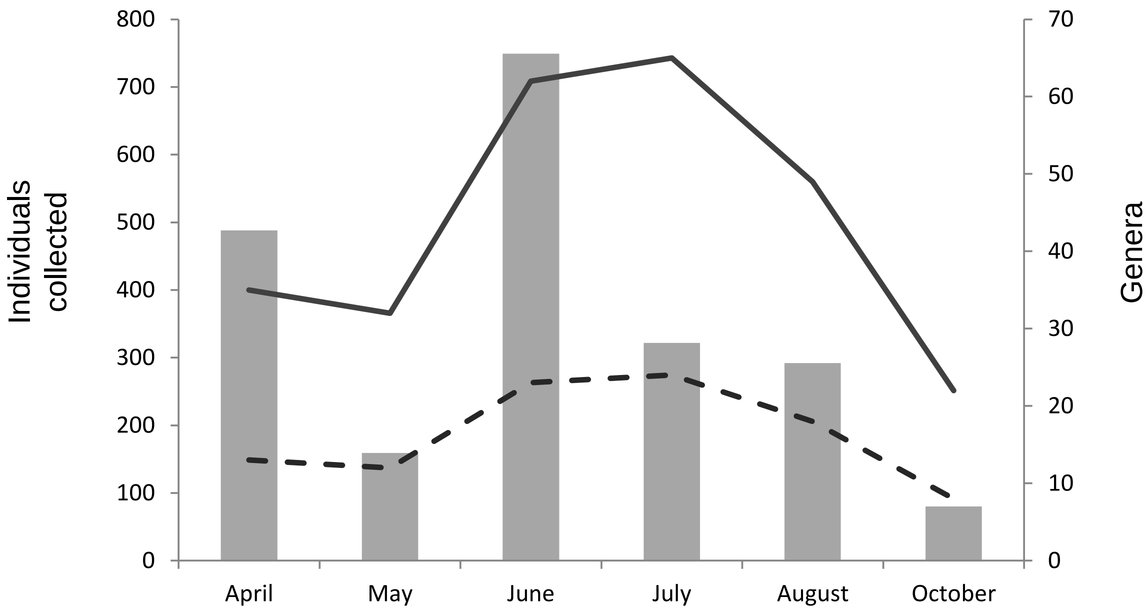

Figure 12. Numbers of Chironomidae individuals (columns) and genera (lines) collected per month, Isle Royale National Park 2010. Solid line is proportion of genera (% = n/37); dashed line is number of genera.

Four genera/subgenera (*Psectrocladius* [*Psectrocladius*], *Orthocladius* [*Eudactylocladius*], *Cricotopus*, and *Coryoneura*) were equally as abundant and widespread at all or most study sites on the southern shore of ISRO (Table 8). Fifteen genera were detected at all or most sites, but were less abundant and often had restricted emergences. The remaining 27 genera were rarely detected and at only a handful of sites. These rare genera occasionally had a wide range (e.g., *Helopelopia*, detected at four sites spanning the entire study area, but in very low abundance), but most were geographically restricted. The two most abundant genera in rock pools were *Orthocladius* (*Eudactylocladius*) and *Psectrocladius* (*Psectrocladius*) (Figure 13). Only seven genera were represented by more than 200 individuals, while half the genera were represented by fewer than 10 individuals. Ranked genera largely followed a linear trend when plotted against log abundance, which again illustrates the expected imbalance in relative generic abundances (Figure 14).

Table 8. Chironomidae genera abundance by site, arranged by site occupancy from southwest (DM) to northeast (PA), Isle Royale National Park, 2009–2010. See Appendix A for site descriptions. Gray fill indicates relative density: light gray = 1–5 individuals detected per site, medium-light gray = 6–20, medium-dark gray = 21–50, dark gray = >50.

Genus/Subgenus	Site																			
	DM	BL	WC	EC	MO	OH	DA	SH	SM	RA	RS	BA	SP	ED	SG	NG	TH	BP	PA	
Psectrocladius (Psectrocladius)	41	68	76	29	70	18	27	36	74	51	56	8	108	57	119	100	18	74	91	Abundant and widespread
Orthocladius (Eudactylocladius)	150	140	25	4	16		7	20	61	9	217	36	88	26	154	54	63	55	68	
Cricotopus	13	95	20	15	32			24	51	11	103	21	20	4	26	11	4	6	21	
Corynoneura	11	36	9		20	1	2	12	18	16	23	141	46	23	65	72	21	49	18	
Ablabesmyia	11	24	1	4	14	1	6	6	15	11	5	5	12	1	1	3	3	4	10	Common-to-uncommon and widespread
Chironomus	2	31	33	8	39	6	11	27	50	11	12	8	16	9	26	13	3	22	6	
Glyptotendipes	5	11	11	2	4		4	11	4	7	10	2	7	3	8	9	4		18	
Eukiefferiella	3	10			2				2	1	17	2	1		1		1		3	
Limnophyes	67	7			2		11	1			4			1	3			3	2	
Dicrotendipes	131	17			1					16	89		2	9	1			3	7	
Micropsectra	2						2	3	7	1	1						46	3		
Synorthocladius	1	1			1		1		2		11	15					1			
Psectrocladius (Allopsectrocladius)	8	20											3						2	
Paratanytarsus		41	58		5	1			1	31	67		16	9	1	4		34	94	
Orthocladius (Orthocladius)		19					5	3	19	2	14	9		1	3		35	1		
Pseudosmittia		1					5		2		3		2		13			1		
Parakiefferiella		1					1				3			1	1	1	170			
Neozavrelia		3						1	28		51	3	2		2	1		17	1	
Heterotrissocladius		4						16	16		3	1		1			10	2	1	
Helopelopia	1	2								1									1	Rare with limited range
Parochlus	1																			
Prodiamesa		1																		
Pseudorthocladius		1																		
Pseudodiamesa		5							1											

Table 8. Chironomidae genera abundance by site, arranged by site occupancy from southwest (DM) to northeast (PA), Isle Royale National Park, 2009–2010. See Appendix A for site descriptions. Gray fill indicates relative density: light gray = 1–5 individuals detected per site, medium-light gray = 6–20, medium-dark gray = 21–50, dark gray = >50 (continued).

	Site																		
Genus/Subgenus	***DM***	***BL***	***WC***	***EC***	***MO***	***OH***	***DA***	***SH***	***SM***	***RA***	***RS***	***BA***	***SP***	***ED***	***SG***	***NG***	***TH***	***BP***	***PA***
Protanypus		1							3		1	6			3				
Potthastia		1									1				1				
Zavrelimyia		1																1	
Tanytarsus					1									2					
Orthocladiinae genus						1													
Sergentia							1												
Metriocnemus							10			1	4								
Pagastia								5	4			1			3	1	7		
Monodiamesa										1		1							
Diamesa											2								
Paracladius											7	1							
Conchapelopia											2	1							
Thienemanniella										1			1						
Thienemannimyia											2	4			2			1	
Nanocladius											2							2	1
Orthocladius (Euorthocladius)												5			1				
Parasmittia												3				2			
Procladius													3						3
Orthocladius (Pogonocladius)															1				
Parachironomus																		1	
Polypedilum																			41
Endochironomus																			1

Rare with limited range

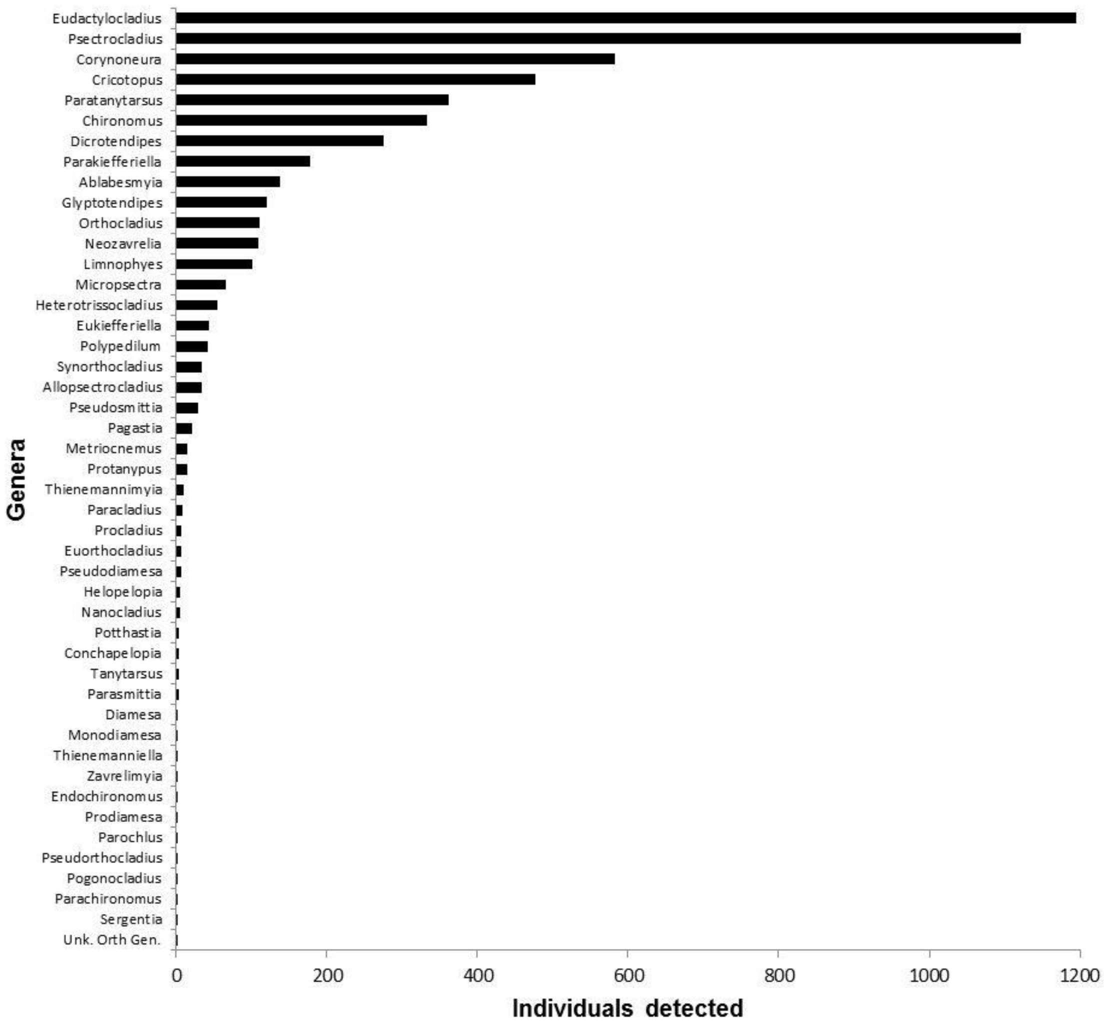

Figure 13. Proportional distribution of Chironomidae genera, Isle Royale National Park, 2009 and 2010.

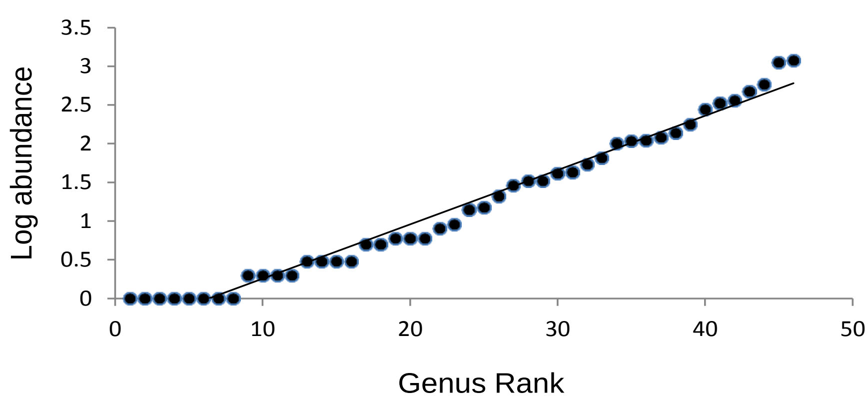

Figure 14. Chironomidae genus log abundance on rank, with linear trendline, Isle Royale National Park, 2009 and 2010.

Chironomidae – Apostle Islands National Lakeshore

Twenty-one Chironomidae taxa were collected from pools at Bear Island and Devils Island (Table 9). All 21 of the taxa at APIS were also found in pools at PIRO, and 19 taxa were also found in pools at ISRO. The subfamily Orthocladiinae dominated both richness (13 taxa) and number of specimens (71% of the total). The subfamily Chironominae was second in species richness (6 taxa), and only one taxon in each of Tanypodinae and Diamesinae was detected (Figure 15).

Table 9. Chironomidae taxa occurrence at Apostle Islands National Lakeshore, by pool type, 2010. Genera are separated into subfamilies; subgenera are shown in parentheses.

SUBFAMILY	Permanent		Ephemeral	
Genus	***Lichen Zone***	***Splash Zone***	***Lichen Zone***	***Splash Zone***
TANYPODINAE				
Ablabesmyia	X		X	
DIAMESINAE				
Diamesa	X			
ORTHOCLADIINAE				
Corynoneura	X	X	X	X
Cricotopus	X	X	X	X
Eukiefferiella	X			
Heterotrissocladius	X	X	X	X
Limnophyes	X		X	
Metriocnemus	X			
Orthocladius (Eudactylocladius)	X	X	X	X
Orthocladius (Orthocladius)	X	X	X	X
Parakiefferiella	X	X		X
Parametriocnemus	X			
Psectrocladius (Psectrocladius)	X	X	X	
Thienemanniella	X	X	X	X
Tvetenia	X			
CHIRONOMINAE				
Chironomus	X		X	
Dicrotendipes	X		X	
Glyptotendipes	X		X	
Polypedilum	X		X	X
Micropsectra	X		X	X
Paratanytarsus	X			
Total Taxa	**21**	**8**	**14**	**9**

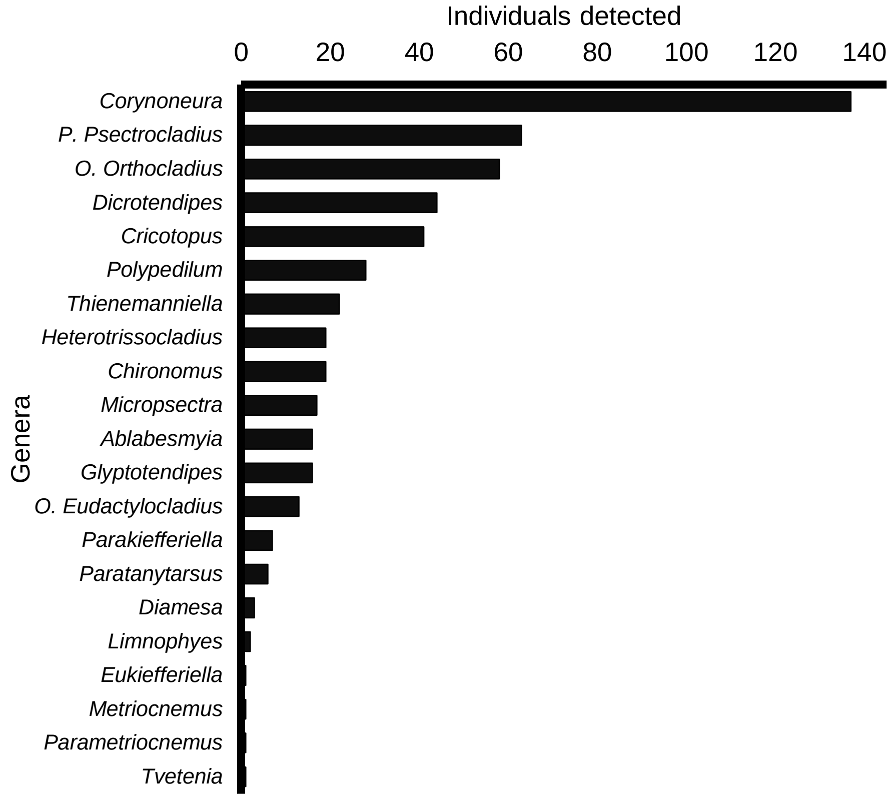

Figure 15. Taxa arrayed by abundance across all samples from all habitat types at Apostle Islands National Lakeshore, 2010.

All 21 of the taxa detected at APIS were detected in pools on Bear Island. Sixteen of the taxa were detected in pools on Devils Island. Orthocladiinae dominated both richness and abundance in pools on both islands. The Jaccard's Similarity for Chironomidae of the two islands is 0.762, which is a statistically significant departure from the value expected due to chance ($p<0.001$), indicating no significant differences in composition between the two islands.

Taxonomic richness across pool zones and types differed between the May and September collections. Twenty taxa were detected in May, including nine taxa that were not detected in September. The nine taxa only emerging in May were predominantly Diamesinae (*Diamesa*) and Orthocladiinae (*Eukiefferriella*, *Limnophyes*, *Metriocnemus*, *Orthocladius* [*Orthocladius*], *Parametriocnemus* and *Tvetenia*), which are cold-adapted and generally emerge in winter or early spring in streams. Emergence of two Tanytarsini (*Micropsectra* and *Paratanytarsus*) was also restricted to May. Some species of *Micropsectra* in the upper Midwest are also somewhat cold-adapted and can emerge in winter or early spring. All taxa with emergence restricted to May occurred exclusively, or were most abundant, in permanent lichen zone pools.

Twelve taxa were detected in September. Eleven of the 12 also emerged in May. *Glyptotendipes* was the only taxon with emergence restricted to September sample dates, but it was detected in both of the permanent lichen zone pools on both islands and in the ephemeral lichen zone pool on Bear Island. Species of *Glyptotendipes* are more abundant in lakes and ponds, and emergence is usually confined to months with warmer water temperatures. The Jaccard's Similarity for Chironomidae emerging in May versus September is 0.524, which is not a statistically significant departure from the value expected due to chance ($p>0.05$), indicating no significant differences in taxonomic richness across all pool types for the two months.

As at ISRO in 2010, pool types at APIS were divided into four categories, which were distinguished by zone and type: permanent lichen pools (two pools at each site), permanent splash pools (two pools at each site), ephemeral lichen pool zones (one sample for the zone at each site) and ephemeral splash pool zones (one sample for the zone at each site). All 21 APIS taxa were collected in permanent lichen zone pools, with the richness of individual pools ranging from 7 to 17 taxa. By contrast, only eight taxa occurred in the permanent splash zone pools, with richness in individual pools ranging from one to five taxa. The cumulative richness for ephemeral lichen zone pools was 14 taxa detected, while only nine taxa were detected in ephemeral splash zone pools (see Table 9).

With four pool categories, as listed above, there were six pair-wise comparisons of Jaccard's Similarity for chironomids at APIS. Four of the comparisons did not indicate statistically significant departures from randomness, including ephemeral splash versus ephemeral lichen ($C_{jk} = 0.533$), ephemeral splash versus permanent lichen ($C_{jk} = 0.450$), ephemeral lichen versus permanent splash ($C_{jk} = 0.467$), and permanent lichen versus permanent splash (0.381). Therefore, neither statistical similarity nor dissimilarity in chironomid composition appears to exist between the two zones, regardless of pool types being compared.

However, the other two comparisons did represent statistically significant departures from values expected due to chance. Both indicated significant similarity in composition between the two pool types compared: ephemeral splash versus permanent splash ($C_{jk} = 0.700$, $P = 0.05$) and ephemeral lichen versus permanent lichen ($C_{jk} = 0.667$, $P<0.01$). Consequently, we conclude that different pool types within zones house the same chironomid composition, making within-zone physical pool differences appear unrelated to community composition.

The Chao 1 estimator suggests a community of at least 29 genera/subgenera in coastal pools at APIS, with a maximum of 86 taxa (the upper 95% confidence interval; Figure 16). Chao (1984) advised that the estimator mean should be considered the minimum estimate. For this analysis, the curve does not appear to reach an asymptote after all samples have been analyzed; therefore, the true richness at APIS is likely higher than the current estimator mean.

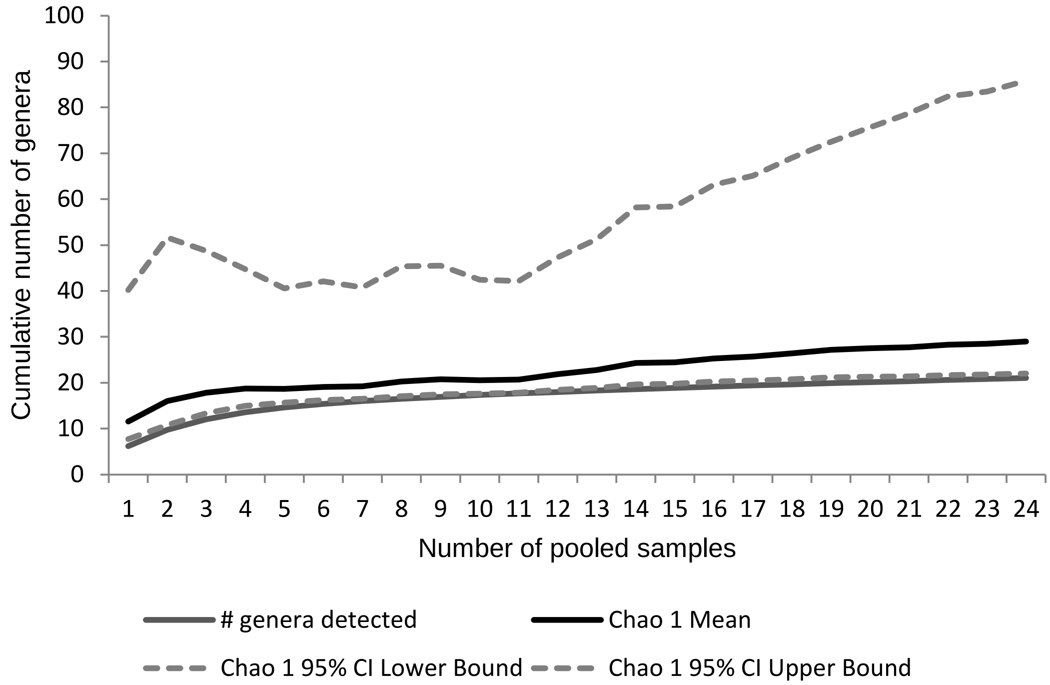

Figure 16. Pooled samples (x-axis) of Chironomidae genus/subgenus (y-axis) detection, true genus richness estimate (Chao 1 mean, upper solid line), and actual number of genera detected (lower solid line) at Apostle Islands National Lakeshore, 2010.

Chironomidae – Pictured Rocks National Lakeshore

Forty-one Chironomidae taxa were collected from pools at PIRO (Table 10, Figure 17). Eleven of the taxa collected at PIRO were not found in pools at ISRO or APIS. However, these taxa are commonly encountered in small-to-medium-sized trout streams in Minnesota, and their occurrence in pools at PIRO is probably influenced by the streams that flow into the park and act as a reserve of adults, some of which occasionally oviposit in the pools. Thirty of the taxa at PIRO also occured in pools at ISRO. All 21 of the taxa collected at APIS also occurred in pools at PIRO.

Orthocladiinae dominated both richness (21 taxa) and specimens (79.5%). Chironominae was second-most species-rich (12 taxa) in pools at PIRO. Four Tanypodinae occurred in the pools, and Diamesinae and Prodiamesinae were each represented by two genera. Analyses did not include the medicolous or cave pools, which could not be effectively compared to other parks, resulting in four genera (*Stempelinella*, *Labrundinia*, *Acricotopus*, and *Procladius*) listed in Figure 17 but not presented in other results.

Taxonomic richness across all pool types was very similar for pools at Au Sable Point (31 taxa) and Miner's Beach (32 taxa). The Jaccard's Similarity value for these two sites was 0.575, which shows statistically significant similarity of Chironomidae composition for these two areas ($p<0.01$). Forty of the 41 taxa detected at PIRO occurred at these two sites. Au Sable Point and Mosquito Harbor shared only 15 of the 30 taxa that were detected at these two sites, and the Jaccard's Similarity value was not statistically significant ($C_{jk} = 0.428$, $p>0.05$). Similarly, Miner's Beach and Mosquito Harbor shared 16 of 30 taxa, and the Jaccard's Similarity value also was not significant (Cjk = 0.457, P>0.05).

Table 10. Chironomidae taxa occurrence at Pictured Rocks National Lakeshore by pool type and zone, 2010. Genera are separated into subfamilies; subgenera are shown in parentheses.

SUBFAMILY Genus	Permanent *Lichen Zone*	Permanent *Splash Zone*	Ephemeral *Lichen Zone*	Ephemeral *Splash Zone*
PRODIAMESINAE				
Prodiamesa	X			
Odontomesa	X			
TANYPODINAE				
Ablabesmyia	X		X	
Conchapelopia	X	X	X	
DIAMESINAE				
Diamesa	X			
Pagastia	X		X	
ORTHOCLADIINAE				
Brillia	X			
Corynoneura	X	X	X	X
Cricotopus	X		X	X
Eukiefferiella	X		X	X
Heterotrissocladius		X		X
Limnophyes	X		X	X
Metriocnemus	X			
Nanocladius	X	X		X
Orthocladius (Eudactylocladius)	X	X	X	X
O. (Euorthocladius)	X		X	
O. (Orthocladius)	X		X	X
Parachaetocladius			X	
Parakiefferiella	X	X		X
Parametriocnemus	X			
Paraphaenocladius				X
Psectrocladius (Psectrocladius)	X		X	
Rheocricotopus	X			
Synorthocladius	X			
Thienemanniella	X	X	X	X

Table 10. Chironomidae taxa occurrence at Pictured Rocks National Lakeshore by pool type and zone, 2010. Genera are separated into subfamilies; subgenera are shown in parentheses (continued).

	Permanent		Ephemeral	
SUBFAMILY *Genus*	***Lichen Zone***	***Splash Zone***	***Lichen Zone***	***Splash Zone***
CHIRONOMINAE				
Tvetenia	X			
Chironomus	X			
Cryptochironomus	X			
Dicrotendipes	X		X	
Glyptotendipes	X			
Microtendipes	X			
Parachironomus	X			
Paratendipes	X			
Polypedilum	X			
Micropsectra	X		X	
Paratanytarsus	X		X	
Tanytarsus	X		X	
Total Taxa	**34**	**7**	**17**	**11**

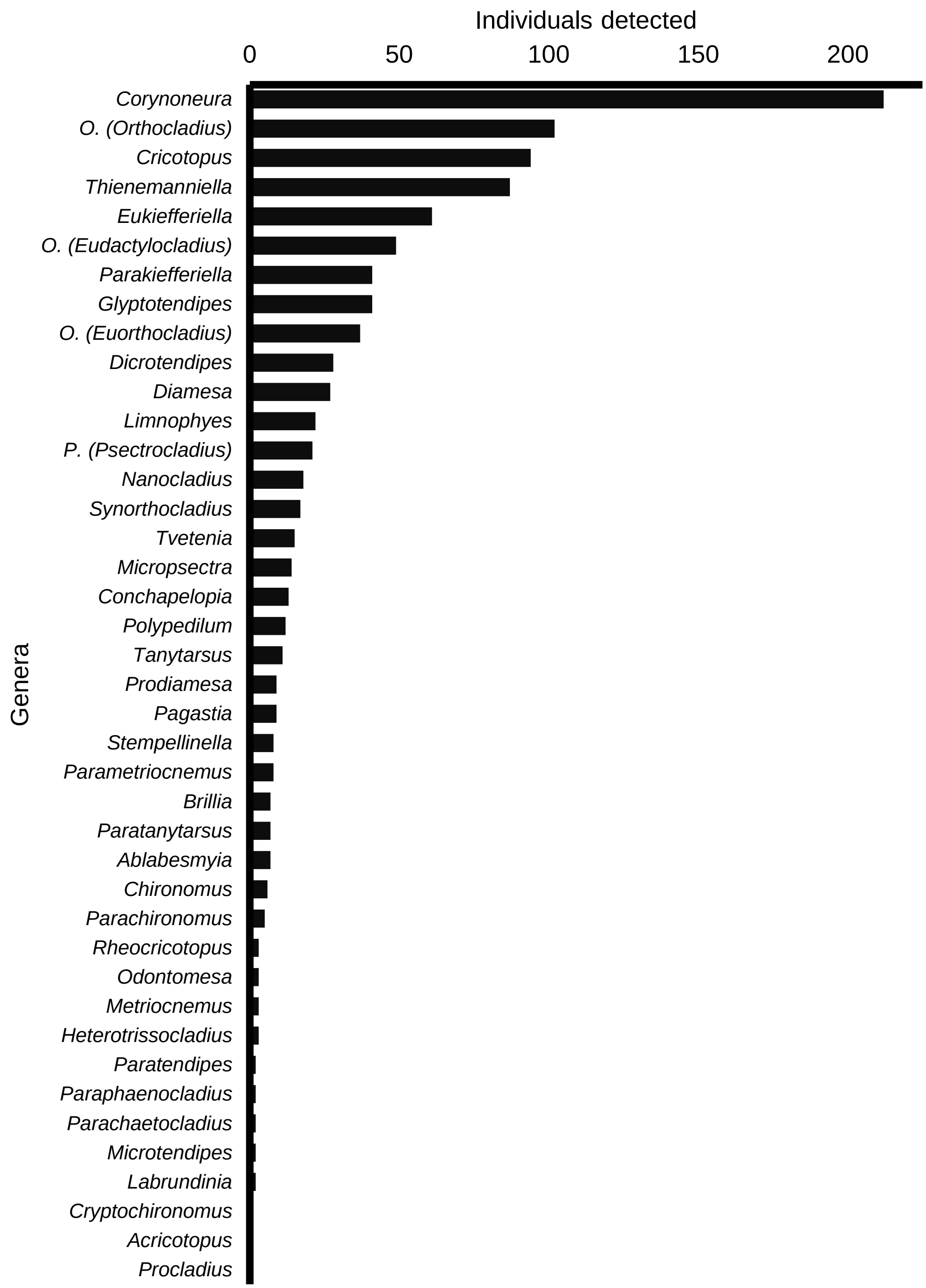

Figure 17. Taxa arrayed by abundance across all samples from all habitat types at Pictured Rocks National Lakeshore, 2010.

Taxonomic richness across all pool types differed between the May and August sample dates. Twenty-nine taxa were detected in May, including 17 taxa that were not detected in August. The taxa only emerging in May were predominantly Prodiamesinae (*Prodiamesa*, *Odontomesa*), Diamesinae

(*Diamesa*, *Pagastia*), and Orthocladiinae (*Brillia*, *Heterotrissocladius*, *Metriocnemus*, *Orthocladius* [*Euorthocladius*], *Parachaetocladius*, *Parametriocnemus* and *Tvetenia*) that are cold-adapted and generally emerge in late winter or early spring in streams. All taxa with emergence restricted to May occurred exclusively, or were most abundant, in permanent lichen zone pools. By contrast, taxa that were only detected as emerging in August consisted predominantly of Chironominae that are more commonly detected emerging during summer in streams (e.g., *Chironomus*, *Dicrotendipes*, *Glyptotendipes*, *Microtendipes*, *Parachironomus*, and *Polypedilum*).

The Jaccard's Similarity for Chironomidae emerging in May versus September is 0.293, which, although low, is not a statistically significant departure from the value expected due to chance ($p>0.05$), indicating no significant differences in taxonomic richness across all pool types for the two months. However, the differences in known emergence times and the tendency of cooler-adapted taxa emerging in May, combined with the predominance of Chironominae in August, suggests a seasonal difference in emergence influenced by water temperatures.

Pool types at PIRO are structurally more diverse than at ISRO and were initially divided into six categories. Four categories were similar to pool zones and types on ISRO, and they consisted of four permanent lichen pools, four permanent splash pools, two ephemeral lichen pools, and two ephemeral splash pools. Thirty-four of the 41 taxa detected at PIRO were collected in at least one of the permanent lichen pools, with richness from individual collections in permanent lichen pools ranging from 7 to 15 taxa. By contrast, only seven taxa occurred in permanent splash zone pools, with richness in individual collections ranging from three to six taxa. Cumulative taxa richness for ephemeral lichen zone pools ($n = 17$) was higher than splash zone pools ($n = 11$). Richness totals for individual collections from ephemeral lichen pools ranged from 8 to 11 taxa, while sample richness from ephemeral splash pools ranged from five to nine taxa.

Jaccard's similarity revealed significantly different community compositions between permanent lichen zone pools and permanent splash zone pools ($C_{jk} = 0.1714$, $P = 0.05$). All other comparisons of pool zones and types were non-significant. The Chao 1 estimator suggests a community of at least 43 genera in coastal pools at PIRO, with an upper 95% confidence interval of 53 (Figure 18). Forty-three genera should be viewed as the low estimate (Chao 1984), even though the curve appears to reach an asymptote by about 18–20 samples. With a quickly declining upper confidence interval and closely matching detected-to-estimated community richness, PIRO samples appear to be the most representative of the actual chironomid rock pool community.

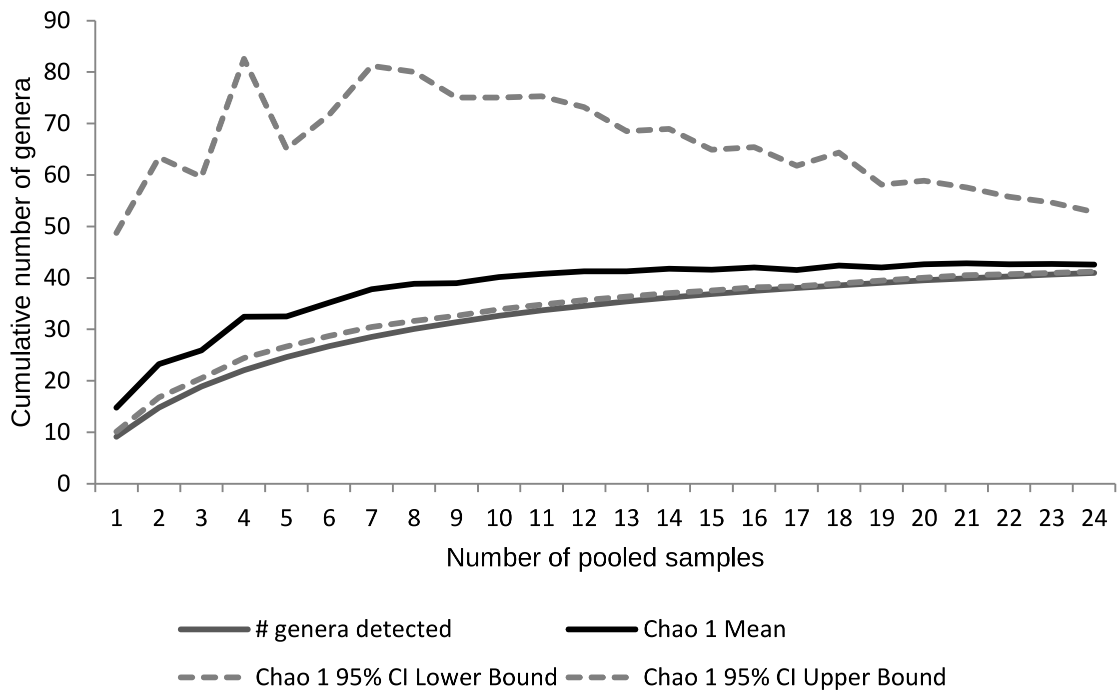

Figure 18. Pooled samples (x-axis) of Chironomidae genus/subgenus (y-axis) detection, true genus richness estimate (Chao 1 mean, upper solid line), and actual number of genera detected (lower solid line) at Pictured Rocks National Lakeshore, 2010.

Non-Chironomidae – Isle Royale National Park

Table 11 shows the geographic range of non-chironomid aquatic macroinvertebrates detected on the surface of or in ISRO pools. Non-chironomid macroinvertebrates were not collected at APIS and PIRO. Using Merrit et al. (2008) and descriptions in McAlpine (1987), families and genera were only included if they have represenatives that are known to be aquatic. Orders in Table 11 confirmed or likely to be using the pools, based on collection of larvae or active adults, include: Collembola (springtails), Odonata (dragonflys and damselflys), Hemiptera (true bugs), Coleoptera (beetles, not including Staphylinidae), Trichoptera (caddisflies), and Diptera (flies).

Many taxa had an unknown relationship to pool habitats, with apparent use of pools being incidental or the result of entrapment and death on pool surfaces. Ephemeroptera and Plecoptera appeared to use pool habitat incidentally, and Staphylinidae, which were detected at nearly all sites, may have been using pool edges while searching for prey. Plecoptera did not appear to be using the pools for larval development, but were swarming on the rocks and pool surfaces after emerging from Lake Superior. Particularly common plecopterans were *Paracapnia* (Family Capniidae) and *Arcynopteryx* (Family Perlodidae), and these are probably common nearshore Lake Superior genera.

At least one hydrophilid beetle was likely to be semi-aquatic and foraging along the edges of pools, a strategy that may be employed by additional taxa (Table 11). Most dytiscid beetles were detected as larvae in pools. Some Collembola (only 2010 samples identified for this order) and Trichoptera may have been incidentally active on pool surfaces, yet some genera from each order were clearly common on and in pools. Many dipteran families listed are aquatic, though not all are expected to have used pool habitats (specifically, Ceratopogonidae, Simuliidae and Tipulidae) and adults likely died on the pools' surface. Other dipterans were clearly active in pools (Chaoboridae, Culicidae) or on the surface (Dolichopodidae, Phoridae). Many Hymenopteran families have aquatic parasitoids, but an anonymous expert established that none of our hymenopterans were aquatic (samples included Ichneumonidae, Braconidae, Scelionidae, Mymaridae, Pteromalidae, Trichogrammatidae, and Eulophidae).

Among the commonly collected Trichoptera, *Apatania zonella* is likely emerging from Lake Superior instead of splash zone pools. *Hydropsyche* larvae, which are sometimes rheophilic (preferring flowing water), have been observed in stationary retreats along cracks and crevices of the lowest pools that receive regular wave wash. Limnephilidae larvae were fairly common in larger lichen zone pools.

Based on frequency of detection in both 2009 and 2010 samples, along with known or likely aquatic habitat use, 17 families across six the most common families and genera that characterized the ISRO coastal rock pool community included:

Collembola	
Hypogastruridae	
Isotomidae	*Semicerura*
Poduridae	*Podura aquatica*
Sminthuridae	
Odonata	
Aeshnidae	*Aeshna*
Hemiptera	
Corixidae	*Callicorixa*, *Sigara*
Gerridae	*Aquarius*
Notonectidae	*Notonecta*
Coleoptera	
Dytiscidae	*Agabus, Liodessus, Rhantus, Stictotarsus*
Trichoptera	
Apataniidae	*Apatania zonella*
Hydropsychidae	*Hydropsyche*
Limnephilidae	*Limnephilus*, *Psychoglypha*
Diptera	
Culicidae	*Aedes*
Phoridae	*Dohrniphora*

Table 11. Non-Chironomidae aquatic macroinvertebrates detected in Isle Royale National Park rock pools, 2009–2010. Sites are arranged from southwest (DM) to northeast (PA). See Appendix A for site descriptions. "‡" indicates specimens sent to experts for confirmation but not returned and may be considered tentatively identified; all other taxa were confirmed.

ORDER		Site																		
Family	***Genus***	***DM***	***BL***	***WC***	***EC***	***MO***	***OH***	***DA***	***SH***	***SM***	***RA***	***RS***	***BA***	***SP***	***ED***	***SG***	***NG***	***TH***	***BP***	***PA***
AMPHIPODA																				
	Hyalella azteca													X						
COLLEMBOLA																				
Entomobryidae																				
	‡ *Harlomillsia*	X																		
	‡ *Tomocerus*		X																	
Hypogastruridae																				
	Genus 1	X	X									X								X
Isotomidae																				
	Semicerura	X	X									X								X
	Genus 1	X										X								X
Poduridae																				
	Podura aquatica L	X	X																	X
Sminthuridae																				
	Sminthurides											X								X
	Sminthurus											X								
	Genus 1	X	X									X								X
EPHEMEROPTERA																				
Baetidae																				
	‡ *Baetis*																X			
	‡ *Camelobaetidus?*																		X	
	‡ Genus 1		X									X								
Caenidae																				
	‡ *Amercaenis or Caenis*?																			X
Heptageniidae																				
	‡ *Heptagenia*										X									
	‡ *Leucrocuta*	X	X																	
	‡ Genus 1											X								
Leptophlebiidae																				
	‡ *Leptophlebia*							X					X							
	‡ Genus 1						X			X						X				
ODONATA																				
Aeshnidae																				
	Aeshna	X	X									X								X
	Triacanthagyna			X					X	X	X			X	X					

Table 11. Non-Chironomidae aquatic macroinvertebrates detected in Isle Royale National Park rock pools, 2009-2010. Sites are arranged from southwest (DM) to northeast (PA). See Appendix A for site descriptions. "‡" indicates specimens sent to experts for confirmation but not returned and may be considered tentatively identified; all other taxa were confirmed (continued).

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Family	***Genus***	***DM***	***BL***	***WC***	***EC***	***MO***	***OH***	***DA***	***SH***	***SM***	***RA***	***RS***	***BA***	***SP***	***ED***	***SG***	***NG***	***TH***	***BP***	***PA***
Libellulidae																				
	‡ *Erythrodiplax*										X								X	
	‡ *Libellula*																			X
	‡ Genus 1																		X	
PLECOPTERA																				
Capniidae																				
	Paracapnia	X	X	X		X		X	X	X	X	X	X	X	X	X	X	X	X	X
	‡ *Capnia*					X														
	‡ *Allocapnia*	X																		
Chloroperlidae																				
	‡ *Haploperla*																	X	X	
	Genus 1		X	X		X					X	X	X	X	X	X	X		X	
Perlidae																				
	‡ Genus 1																			X
Perloididae																				
	Arcynopteryx		X		X	X		X		X		X	X		X	X			X	
	‡ *Diura or Isoperla?*		X																	
	‡ *Osobenus*																			X
	Skwala										X									X
	‡ Genus 1		X																	
HEMIPTERA																				
Corixidae																				
	Callicorixa	X	X	X	X	X	X			X	X	X	X		X	X	X	X	X	X
	‡ *Corisella*	X																		
	Sigara			X	X	X	X	X		X	X		X	X	X	X	X	X	X	
Gerridae																				
	Aquarius	X	X	X		X					X		X	X	X	X	X	X	X	X
	‡ *Gerris*					X	X													
	‡ *Limnoporus*					X						X								
Notonectidae																				
	‡ *Buenoa*																		X	
	‡ *Notonecta*	X	X																	
Saldidae																				
	‡ *Rupisalda*															X				
	‡ *Salda*																		X	
	‡ *Saldula*	X																		

Table 11. Non-Chironomidae aquatic macroinvertebrates detected in Isle Royale National Park rock pools, 2009-2010. Sites are arranged from southwest (DM) to northeast (PA). See Appendix A for site descriptions. "‡" indicates specimens sent to experts for confirmation but not returned and may be considered tentatively identified; all other taxa were confirmed (continued).

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Family	***Genus***	***DM***	***BL***	***WC***	***EC***		***MO***	***OH***	***DA***	***SH***	***SM***	***RA***	***RS***	***BA***	***SP***	***ED***	***SG***	***NG***	***TH***	***BP***	***PA***
COLEOPTERA																					
Carabidae																					
	Genus 1												X								
Dytiscidae																					
	Acilius			X											X						
	Agabus			X			X				X		X		X			X		X	
	Copelatus?											X									
	Hydroporus						X				X			X							
	Hydrotrupes?																		X		
	Hygrotus																				X
	Laccophilus					X															
	Liodessus		X	X																	X
	Nebrioporus			X					X												
	Neoporus			X					X		X								X		
	Oreodytes																				X
	Rhantus		X	X	X	X					X		X		X		X			X	
	Stictotarsus		X	X	X		X	X		X	X				X	X	X	X	X	X	X
	Genus 1			X																	
Gyrinidae																					
	Gyrinus		X	X													X			X	
Hydrophilidae																					
	Helophorus							X			X	X		X	X	X	X			X	X
	Helocombus												X								
	Hydrobius?		X	X																X	X
	Hydrochus		X										X								
	Paracymus?			X																	
	Genus 1																				X
Scirtidae																					
	Ora?		X																		
	Prionocyphon										X										
TRICHOPTERA																					
Apataniidae																					
	Apatania zonella		X	X	X			X	X	X	X	X	X			X		X	X	X	X
Limnephilidae																					
	Frenesia																			X	
	Glyphopsyche			X	X																
	Grammotaulis		X	X																	

Table 11. Non-Chironomidae aquatic macroinvertebrates detected in Isle Royale National Park rock pools, 2009-2010. Sites are arranged from southwest (DM) to northeast (PA). See Appendix A for site descriptions. "‡" indicates specimens sent to experts for confirmation but not returned and may be considered tentatively identified; all other taxa were confirmed (continued).

ORDER		**Site**																		
Family	***Genus***	***DM***	***BL***	***WC***	***EC***	***MO***	***OH***	***DA***	***SH***	***SM***	***RA***	***RS***	***BA***	***SP***	***ED***	***SG***	***NG***	***TH***	***BP***	***PA***
	Limnephilus			X					X		X		X	X		X	X	X	X	X
	Psycoglypha			X	X	X		X	X								X			
	Hesperophylax			X																
Hydropsychidae																				
	Hydropsyche			X		X			X		X	X		X	X	X		X	X	X
Hydroptilidae																				
	Genus 1														X					
	Genus 2			X							X	X				X				X
Lepidostomatidae																				
	Lepidostoma togatum				X	X					X									
	Genus 1			X		X	X		X	X			X	X						
Leptoceridae																				
	Oecetis			X										X						
	Ceraclea			X	X						X	X		X				X	X	
Phrygaenidae																				
	Agrypnia			X																
	Genus 1		X	X							X	X	X						X	
DIPTERA																				
Ceratopogonidae																				
	‡ *Echinohelea lanei* Wirth															X				
	‡ *Stilobezzia elegantula* (Johannsen)														X					
	‡ *Stilobezzia sp.*								X											
Chaoboridae																				
	‡ *Chaoborus (Chaoborus)*																X			
Culididae																				
	Aedes		X	X	X	X	X	X	X	X	X	X	X	X	X	X	X	X	X	X
	Anopheles		X	X																
	‡ *Psorophora?*																		X	
Dolichopodidae																				
	‡ *Campsicnemus*										X									
	‡ *Dolichopus*																		X	
	‡ *Diostracus?*																			X
	‡ *Liancalus*								X											
	‡ *Paraphrosylus*											X								
	‡ *Pelastoneurus*		X																	
	‡ *Tachytrechus*											X								

Table 11. Non-Chironomidae aquatic macroinvertebrates detected in Isle Royale National Park rock pools, 2009-2010. Sites are arranged from southwest (DM) to northeast (PA). See Appendix A for site descriptions. "‡" indicates specimens sent to experts for confirmation but not returned and may be considered tentatively identified; all other taxa were confirmed (continued).

ORDER		Site																		
Family	***Genus***	***DM***	***BL***	***WC***	***EC***	***MO***	***OH***	***DA***	***SH***	***SM***	***RA***	***RS***	***BA***	***SP***	***ED***	***SG***	***NG***	***TH***	***BP***	***PA***
	Telmaturgus parvus (Van Duzee)		X	X					X		X	X		X					X	
	Thinophilus?		X	X																
	Xanthochlorus helvinus									X										
	‡ Genus 1															X				
Empididae																				
	Hilara		X	X																
Phoridae																				
	Dohrniphora				X	X			X		X		X		X			X	X	
	‡ *Megaselia*			X																
Psychodidae																				
	‡ *Telmatoscopus*			X																
Sciomyzidae																				
	‡ *Colobaea*																			X
Simuliidae																				
	Helodon		X	X																
	‡ *Parasimulium*																	X		
	Prosimulium		X	X																
	Simulium		X									X	X							
	‡ Genus 1																			X
Stratiomyidae																				
	Allognosta?			X																
Tipulidae																				
	‡ *Antocha*			X																
	Elliptera?			X										X						
	Pedicia					X			X	X	X	X		X	X	X	X		X	X
	Limonia								X				X		X	X			X	
	‡ *Phalacrocera*		X																	
	Tipula		X		X															X
	‡ Genus 1																		X	
	‡ Genus 2																X			

Zooplankton

We defined zooplankton as any animal over 30 µm (our mesh size) up to but not including insects. A total of 177 zooplankton taxa were counted (Table 12). Representatives of major groups are shown in Figure 19. Diversity and density of organisms in the samples were so high that counting took more than four times longer than expected based on previously reported densities and diversity (VanBuskirk and Smith 1991). In total, 115 samples were counted out of 215 total samples taken. All samples were counted from APIS and PIRO from the two visits (spring and late summer). Passage Island (ISRO) samples were counted for all dates, but only the July sample was counted for other ISRO sites so as to capture communities at peak productivity for that region (based on results of the Passage Island counts). Results documented below include summary measures of the entire data set, analyses of regional differences in zooplankton communities, characteristics of ecological zones or pool types, and detailed results from Passage Island.

Table 12. Zooplankton taxa present in rock pools sampled at Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010. Names in parentheses are tentative identifications (75%–95% certain).

	APIS	ISRO	PIRO
COPEPODA (20 taxa)			
Acanthocyclops capillatus	X		
Cyclops sp.	X	X	
Diacyclops albus		X	
Diacyclops langoidus			X
Diacyclops nanus		X	X
Diacyclops thomasi	X	X	
Diacyclops sp.	X	X	X
Eucyclops elegans	X		
Microcyclops rubellus	X	X	X
Microcyclops vericans	X		
Paracyclops (chiltoni)			X
cyclopoid adult, unidentified		X	
Harpacticoid	X	X	X
Epischura lacustris		X	X
Leptodiaptomus sicilis	X	X	X
Leptodiaptomus sp.	X	X	
Limnocalanus macrurus			X
Senecella calanoides	X		
Skistodiaptomus oregonensis	X		
Skistodiaptomus reighardi		X	
CLADOCERA (28 taxa)			
Acroperus harpae		X	
Alona sp.	X	X	
Alona bicolor		X	
Alona circumfimbriata		X	X
Alona costata	X	X	
Alona gutatta	X	X	X
Alona quadrangula		X	
Alona rectangula	X	X	
Alonella nana	X	X	
Biapertura (Alona) affinis	X	X	
Chydorus sp.	X	X	X
Chydorus faviformis		X	
Chydorus sphaericus	X	X	
Eurycercus longirostris	X		
Kurzia (latissima)	X		
(Paralona pigra)		X	
Ceriodaphnia sp.		X	X
Ceriodaphnia lacustris		X	
Ceriodaphnia quadrangula		X	
Diaphanosoma sp.		X	
Simocephalus sp.		X	
Scapholeberis mucronata		X	
Bosmina longirostrus	X		
Bosmina spp.	X	X	
Daphnia ambigua		X	
Daphnia pulex catawba		X	
Daphnia mendotae dentifera		X	
Holopedium gibberum	X	X	
ROTIFERA (96 taxa)			
Bdelloid rotifer	X	X	X
Adineta sp.	X	X	X
Anuraeopsis fissa	X		
Ascomorpha sp.	X	X	X
Asplanchna herricki	X		
Asplanchna priodonta	X	X	X
Asplanchna sp.	X	X	X
Cephalodella sp.	X		X
Collotheca mutabilis	X		
Collotheca pelagica		X	X
Colurella sp.	X	X	X
Conochilus sp.	X	X	X
Conochilus hippocrepis	X		
Conochilus unicornis	X	X	
Conochiloides dossarius	X	X	X
Dicranophorus sp.	X	X	X
Dissotrocha sp.	X	X	
Encentrum sp.	X	X	
Euchlanis calpidia		X	
Euchlanis dilatata		X	
Euchlanis triquetra		X	
Euchlanis spp.		X	X
Gastropus stylifer	X	X	X
Habrotrocha sp.			X
Harringia sp.	X		
Hexarthra mira		X	X
Kellicottia bostoniensis	X	X	X
Kellicottia longispina	X	X	X
Keratella cochlearis cochlearis	X	X	X
Keratella cochlearis robusta		X	
Keratella cochlearis tecta	X	X	X
Keratella crassa		X	
Keratella earlinae	X	X	X
Keratella hiemalis	X		X
Keratella quadrata	X		
Lecane candida		X	
Lecane crepida	X		

Table 12. Zooplankton taxa present in rock pools sampled at Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010. Names in parentheses are tentative identifications (75%–95% certain) (continued).

	APIS	ISRO	PIRO
ROTIFERA (continued)			
Lecane flexilis	X	X	
Lecane inermis	X	X	X
Lecane luna		X	X
Lecane mira	X	X	X
Lecane mucronata	X		
Lecane ovalis		X	
Lecane stenroosi	X		
Lecane (tenuiseta)	X	X	
Lecane tudicola			X
Lepadella patella	X	X	X
Lepadella ovalis	X	X	X
Lepadella triptera	X		
Lophocharis sp.		X	
Monostyla sp.	X		
Monostyla bulla	X	X	X
Monostyla closterocerca	X	X	
Monostyla copeis	X	X	
Monostyla cornuta	X		
Monostyla crenata	X		
Monostyla lunaris	X	X	X
Monostyla obtusa	X	X	
Monostyla quadridentata		X	
Mytilina ventralis		X	
Notholca acuminata	X		X
Notholca caudate	X		
Notholca labis	X		
Notholca laurentiae	X		
Notholca squamula	X		X
Notholca sp.	X		
Notomata sp.	X		
Philodina sp.	X	X	X
Ploesoma sp.	X		X
Ploesoma hudsoni	X		
Ploesoma truncata	X		
Polyarthra dolichoptera	X	X	
Polyarthra major		X	
Polyarthra remata	X	X	X
Polyarthra vulgaris	X	X	X
Polyarthra spp.	X	X	X
Pompholyx sulcata	X	X	
Proales sp.	X	X	X
Rotaria sp.	X		
Schwabia sp.	X		
Synchaeta sp.	X	X	X
Synchaeta grandis			X
Synchaeta kitina			X
Synchaeta tremula			X
Squatinella sp.		X	
Testudinella sp.	X		
Trichocerca caputina			X
Trichocerca cylindrica	X	X	X
Trichocerca elongata		X	
Trichocerca iernis	X		
Trichocerca porcellus			X
Trichocerca pusilla	X	X	
Trichocerca rousseleti	X		
Trichotria tetractis	X	X	
Wierzyskiella velox			X
unidentified rotifer	X	X	X
TESTATE PROTISTA (23 taxa)			
Arcella gibbosa	X		X
Arcella sp.	X		
Centropyxis constricta aerophila	X	X	X
Centropyxis constricta spinosa	X	X	X
Codonella sp.	X	X	X
Cucurbitella (tricuspis)			X
Cyclopyxis spp.	X	X	X
Difflugia bacillaliarum			X
Difflugia (lucida)		X	
Difflugia (oblonga)	X		
Difflugia urceolata	X		
Difflugia sp.	X	X	
Euglypha sp.	X	X	X
Geopyxella sp.	X	X	
Hyalosphenia papilio	X		
Lesquereusia spiralis		X	X
Nadinella sp.			X
Nebellidae	X		X
Phryganella sp.	X		
Trinema sp.	X	X	
Wailesella eboracensis	X		X
unidentified testate			X
unidentified protist	X	X	X
OSTRACODA (7 taxa)			
Candoninae		X	
Cypridopsinae	X		
Cypridopsis sp.			X
Potamocypris unicaudata	X		
Potamocypris sp.	X	X	
Scottia pseudobrowniana (?)	X	X	
unidentified ostracod		X	
juvenile ostracod		X	X
OTHER			
Hydrachnidiae	X	X	X
Tardigrada			X

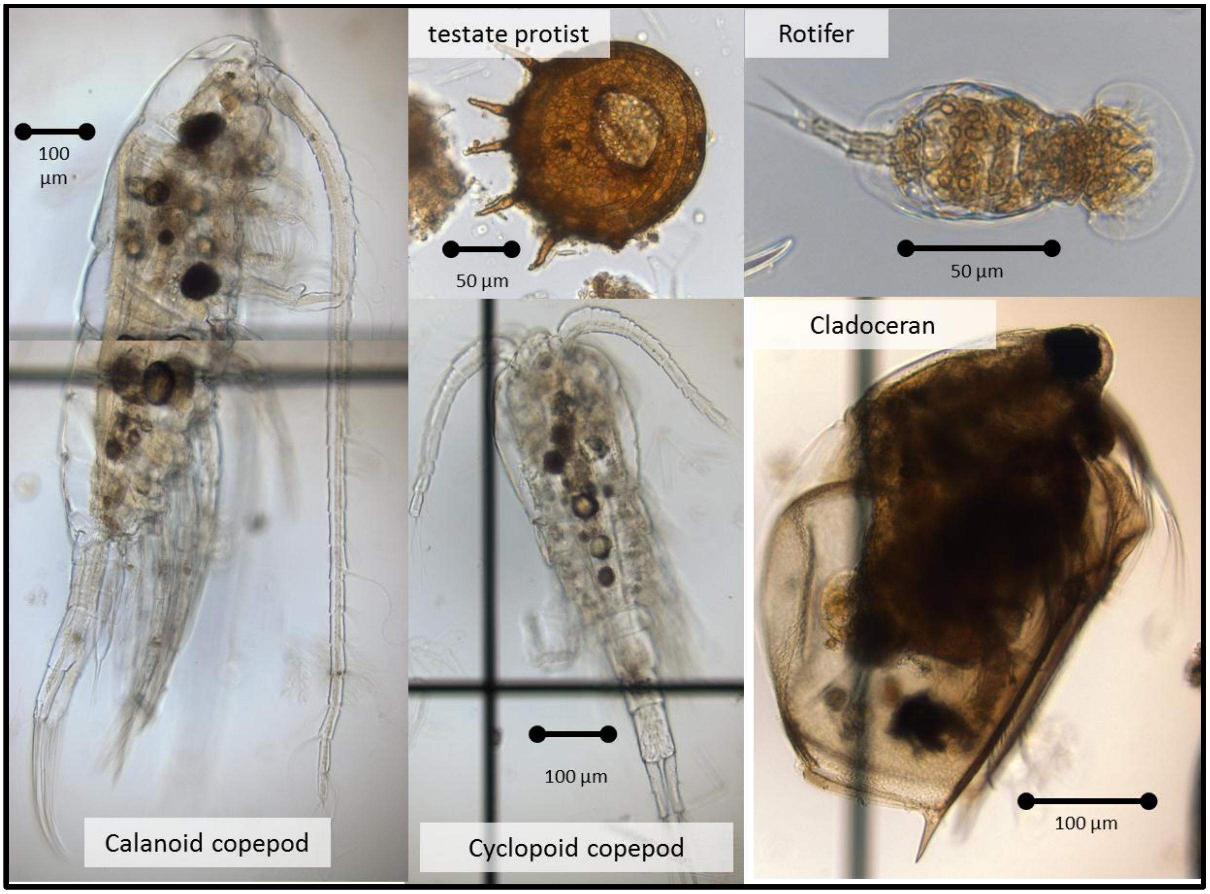


Figure 19. Representatives of major zooplankton groups from rock pools sampled in Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010.

Summary Measures of Zooplankton Community Structure Across All Sites

Statistical summaries of diversity, total zooplankton abundance, and abundance of the primary groups of zooplankton are shown in Table 13. Basic characteristics of the zooplankton abundances are remarkable in both value and range. Maximum densities in a rock pool could reach 80,000 individuals per cubic meter with up to 26 species. Minimum values of zero show how variable the zooplankton densities are in these systems, with standard deviations greater than the means for all taxonomic groups and no single species appearing in all or even most of the pools. No pools sampled were empty.

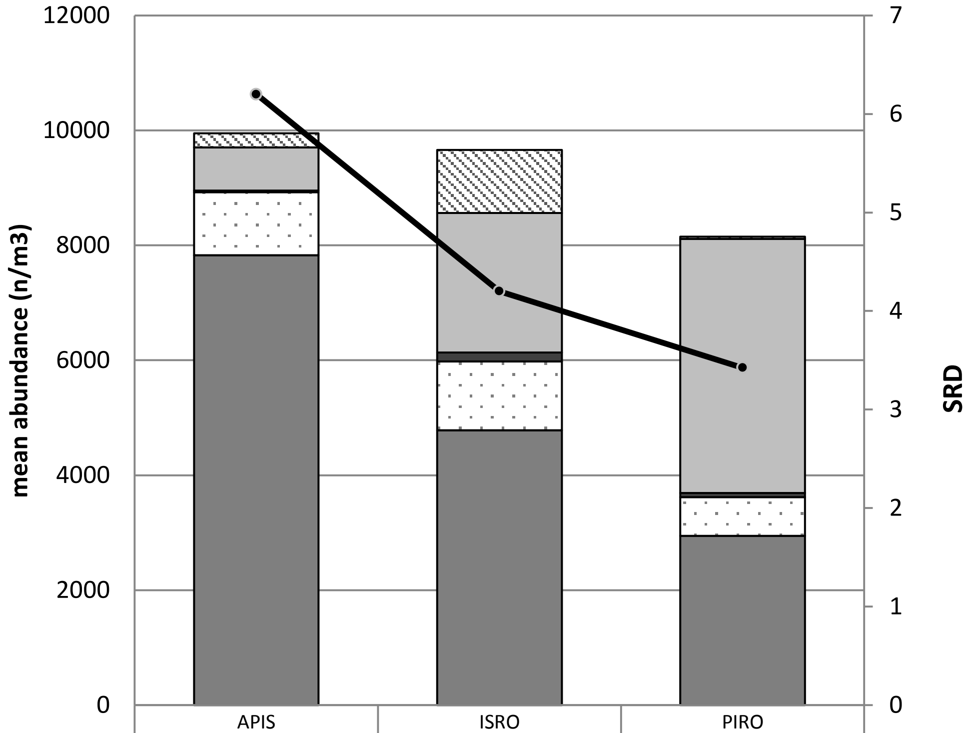
Regional and Site Specific Zooplankton Composition and Abundance

Zooplankton communities at APIS, PIRO, and ISRO were different from one another, as shown by the abundances of major zooplankton groups, taxa unique to each park, and differences between particular sites (Figure 20). The analyses in this section lump all pool types to examine park and site differences. Total zooplankton abundance and species richness were significantly higher at APIS than other parks ($F(2,112) = 3.563$, $p = 0.032$; $F(2,112) = 5.784$, $p = 0.004$ respectively). This is remarkable because if sample bias exists, it should be toward ISRO where several sites were counted only in July when highest abundance and diversity are expected. Total abundances and proportion of some taxa may therefore be over-represented in the data presentation, and separate analyses are described below to circumvent that problem.

Comparing parks, ISRO had significantly greater cladoceran abundance ($F(2,112) = 5.155$, $p = 0.007$). September productivity at APIS is responsible for much of its high abundance results (Figure 20), so more abundant cladocerans at ISRO is not likely an artifact of July results, but shows true community difference. All parks together show a general zooplankton phenology of increased richness and abundance as summer progresses. Combining all sites, species richness was higher in late summer (July through September) than other months. Total zooplankton abundance was significantly higher in July ($F(7, 65) = 7.667$, $p < 0.001$; $H(7) = 40.64$, $p < 0.001$ respectively).

Table 13. Summary statistics for the total zooplankton data set from rock pools sampled in Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010, including Simpson's Index of Diversity (SID) and Simpson's Reciprocal Diversity Index (SRD).

	Mean	Standard deviation	Standard error	Confidence interval	Range	Maximum	Minimum	Median
Species richness	9.761	6.155	0.579	1.147	25	26	1	9
Generic richness	8.115	4.906	0.462	0.914	22	23	1	8
SRD	4.538	2.939	0.276	0.548	15.538	16.538	1	4.273
SID	0.662	0.243	0.0229	0.0453	0.94	0.94	0	0.766
Total zooplankton (n/m^3)	9510.475	13396.386	1260.226	2496.976	78262.932	78422.003	159.071	4931.201
Testate abundance (n/m^3)	1064.227	2141.356	201.442	399.131	12407.538	12407.538	0	159.071
Rotifer abundance (n/m^3)	5163.473	10224.788	961.867	1905.816	73968.015	73968.015	0	1908.852
Copepod abundance (n/m^3)	2417.035	7425.757	698.556	1384.1	71104.737	71104.737	0	318.142
Cladoceran abundance (n/m^3)	672.884	1920.417	180.658	357.95	17020.597	17020.597	0	0
Ostracod abundance (n/m^3)	108.394	336.277	31.634	62.679	2226.994	2226.994	0	0



	APIS	ISRO	PIRO
Cladocera	244.2876071	1100.668694	41.49678261
Copepoda	749.9061429	2427.115581	4419.407348
Ostracoda	28.40553571	156.5053387	76.07743478
Protista (testate)	1096.453679	1195.598161	670.8646522
Rotifera	7828.565643	4782.392645	2946.271565
SRD	6.199321081	4.200917357	3.424690564

Figure 20. Mean zooplankton abundance in proportion to major taxonomic groups at Apostle Islands (APIS), Isle Royale (ISRO), and Pictured Rocks (PIRO), 2010. Shannon's Reciprocal Index (SRD) is shown as a line and represents diversity weighted by evenness (higher values = more diverse and more even).

Of the 177 taxa (primarily species) of zooplankton identified, 42 were common to all parks, 47 were common to two parks, and 89 taxa were unique to one park (Figure 21). A list of taxa distribution by park is given in Table 12. Copepod nauplii and copepodids were counted separately and identified to major group (calanoid or cyclopoid), for a total of 182 analytes. These life stages are important indicators of pool colonization, egg dispersal, and possibly of permanent use by the taxa. These data will be part of future analyses. The nauplii and copepodids, as well as some other unidentifiable organisms, were left out of analyses discussed below, consequently taxa numbers in any given analysis may not add up to 177.

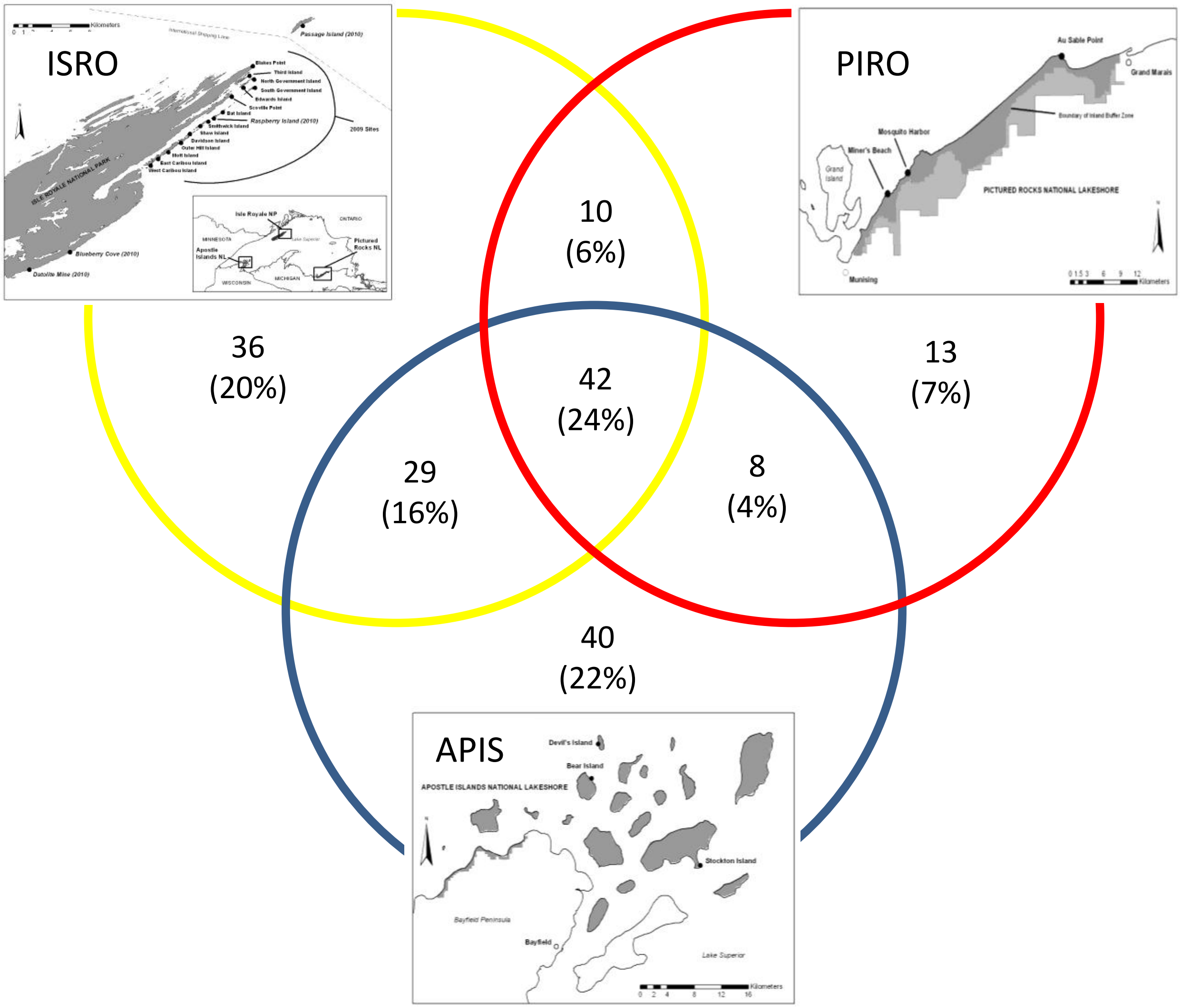
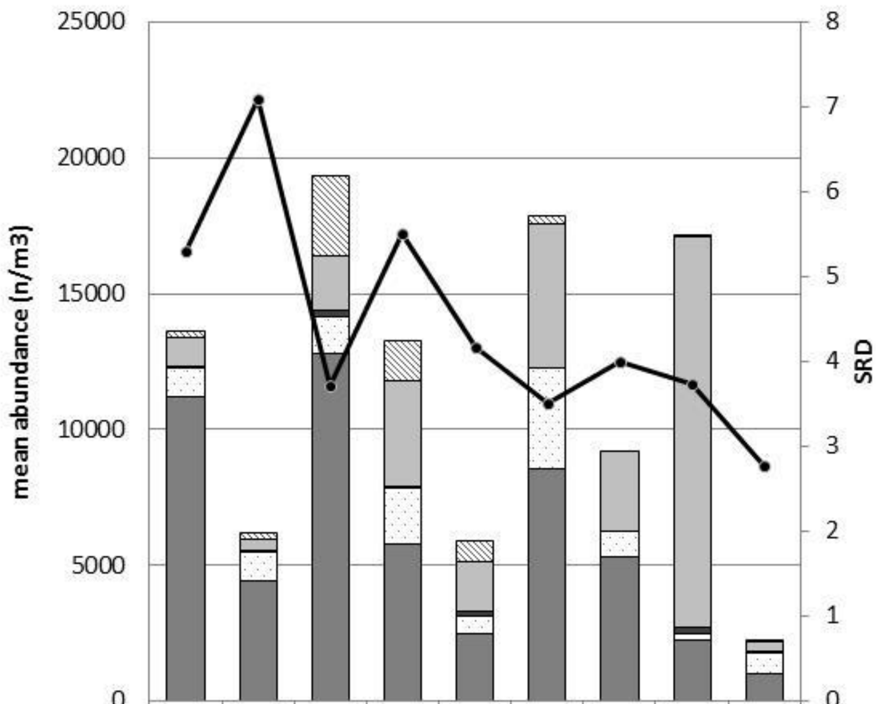


Figure 21. Numbers and relative proportions of zooplankton taxa (primarily species) shared and unique to rock pools studied in Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010.

PIRO's pools were dominated by cosmopolitan species shared with all or one of the other parks. Forty-two taxa were common to all three parks, eight were shared with APIS, ten were shared with ISRO, and only 13 taxa were unique to PIRO. More species were shared between APIS and ISRO than between either of those parks and PIRO. APIS and ISRO contained similar numbers of unique species (40 and 36, respectively), with correspondingly greater overall taxonomic diversity. Many of the cosmopolitan species were rotifers, but that did not exclude specific rotifers from being unique to an individual park. Many of the rock pool zooplankton species are considered rare or incidental catches in Lake Superior, with 60.7% of cladoceran species and roughly 80% of rotifer species from our samples described as incidental by Stemberger (1979) and Balcer et al. (1984).

In addition to differences between parks, there were site-specific differences in zooplankton communities (Figure 22). Devils Island (DI) and Bear Island (BI) at APIS, and Blueberry Cove (BL) and Raspberry Island (RS) at ISRO, each had significantly higher species richness than other sites, (H(9) = 20.991, p = 0.013). Along with Au Sable (AS) from PIRO, these sites had significantly higher Simpsons Reciprocal Diversity values (H(9) = 17.838, p = 0.037). At least some of this relationship (i.e., BL and RS from ISRO) must have been an artifact of counting only July samples, since early and late season samples collected at Passage Island (PA) had lower mean values. Datolite Mine (DM) at ISRO was significantly less rich, even when including July samples.



	BI	DI	BL	DM	PA	RS	AS	MB	MH
Cladocera	227	261	2923	1454	799	318	0	32	80
Copepoda	1079	420	2008	3931	1827	5329	2969	14348	358
Ostracoda	45	11	278	45	175	0	0	286	40
Protista (testate)	1091	1102	1332	2068	652	3712	937	223	736
Rotifera	11226	4431	12825	5795	2487	8563	5338	2259	1054
SRD	5.30	7.10	3.71	5.51	4.17	3.51	4.00	3.74	2.78

Figure 22. Mean zooplankton abundance in proportion to major taxonomic groups at nine pool sites—Bear Island (BI) and Devils Island (DI) from Apostle Islands National Lakeshore; Blueberry Island (BL), Datolite Mines (DM), Passage Island (PA), and Raspberry Island (RI) from Isle Royale National Park; and pools near Au Sable lighthouse (AS), Miners Beach (MB), and Mosquito Harbor (MH) from Pictured Rocks National Lakeshore. Shannon's Reciprocal Index (SRD) is shown as a line and represents diversity weighted by evenness (higher values = more diverse and more even).

Sampling by date did not directly influence the results for AS, BI, and DI. Consequently, site-to-site differences confirmed the trend in park differences described above–the highest abundance and diversity were found at APIS, followed closely by ISRO. The AS site at PIRO was the only pool system at that park that was structured into two zones like APIS and ISRO sites. The other PIRO sites were outside the study design due to geomorphological conditions. Statistically significant differences between major taxonomic groups did not show up at the regional or site level, but the differences were more important in tests between pool types.

Zooplankton Composition and Abundance Across Permanent and Ephemeral Pools from Lichen and Splash Zones

Habitat types used in this study are described in the List of Terms and Acronyms section above. Zooplankton abundance measures for permanent pools from both lichen and splash zones were quantitative, but the ephemeral pools from both zones were sampled by timed search, so results from ephemeral pools should be considered an index and not an inference to population numbers.

There were no statistically significant differences in total zooplankton abundance or diversity between the four pool types in or across zones (ephemeral pools from splash zone, ephemeral pools from the lichen zone, permanent pools from the splash zone, and permanent pools from the lichen zone). Principal components analysis (PCA) showed that, in taking the entire data set together, pool type explains 21.1% of variation in zooplankton species level community structure on the F1 axis and an additional 12.1% more on axis F2 (cumulative 33.2%). These axes represent a spread of pool types roughly containing each of the four pool types in a different quadrant. This plot was too dense to show graphically, and these findings should be considered preliminary. Further work is needed to find a proper transformation of the data to better sort out community composition using this tool. One problem is that more than one-third of species, even if extremely abundant, were found in only one or two pools.

The patchiness of the zooplankton distribution can be broken down in a few ways (Table 14). Cladocera and ostracods have the highest percentage of taxa unique to the lichen zones and to permanent pools. There was also a large percentage of testate protist species unique to permanent pools (number of species was an order of magnitude greater than in ephemeral pools). This comparison treated uniqueness separately in the two categories—lichen against splash and ephemeral against permanent. Species were considered unique to Lake Superior only if they did not occur in the pool samples.

Species or taxa that were unique to either lichen vs splash, permanent vs ephemeral, or Lake Superior (vs pools) are listed in Table 15. Species without a label were found in lichen and splash zones and in both permanent and ephemeral pools. Some qualitative patterns are based on the biology of the organisms. Species unique to the splash zone are characterized by a large group of rotifers and copepods, although both also prefer permanent pools. There is a special community of organisms unique to permanent splash pools. Ephemeral splash pools have the fewest unique species. Lichen zone and permanent pools have a large number of unique cladocerans that are characteristic of littoral areas (Dodson et al. 2010) and support quite a few unique testate protists and rotifers.

Table 14. Zooplankton species distribution compared over pool habitat types from rock pools sampled in Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010. Number and percent of zooplankton taxa unique to lichen vs splash pools, ephemeral vs permanent pools, and taxa unique to Lake Superior (found nearshore but not in pools) are compared separately.

	Pool Type				
	Lichen	***Splash***	***Ephemeral***	***Permanent***	**Lake Superior**
Total taxa unique to each zone:	42	40	15	66	12
	23.2%	22.1%	8.3%	36.5%	6.6%
Cyclopoid taxa	3	2	1	3	4
	21.4%	14.3%	7.1%	21.4%	28.6%
Calanoid taxa	1	2	0	2	2
	14.3%	28.6%	0.0%	28.6%	28.6%
Cladoceran taxa	12	6	5	12	0
	42.9%	21.4%	17.9%	42.9%	0.0%
Rotifer taxa	14	29	7	30	6
	14.6%	30.2%	7.3%	31.3%	6.3%
Testate protist taxa	6	4	1	11	0
	26.1%	17.4%	4.3%	47.8%	0.0%
Ostracod taxa	3	0	0	5	0
	42.9%	0.0%	0.0%	71.4%	0.0%

Table 15. Zooplankton species or taxa unique to either lichen vs splash pools, permanent vs ephemeral pools, or Lake Superior (vs pools) at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore. Species in **bold** font showed no affinities and were found in lichen and splash zones and in both permanent and ephemeral pools. Names in parentheses are tentative identifications (75%–95% certain).

	Pool Type				
	Lichen	***Splash***	***Ephemeral***	***Permanent***	**Lake Superior**
CRUSTACEA (20 taxa)					
Cyclopoid nauplii					
Cyclopoid copepodid					
Calanoid nauplii					
Calanoid copepodid				P	
Acanthocyclops capillatus					
Cyclops sp.	L		E		
Diacyclops albus		S		P	
Diacyclops langoidus					Lsup
Diacyclops nanus					Lsup
Diacyclops thomasi		S			
***Diacyclops* sp.**					
Eucyclops elegans					Lsup
Microcyclops rubellus	L			P	

Table 15. Zooplankton species or taxa unique to either lichen vs splash pools, permanent vs ephemeral pools, or Lake Superior (vs pools) at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore. Species in **bold** font showed no affinities and were found in lichen and splash zones and in both permanent and ephemeral pools. Names in parentheses are tentative identifications (75%–95% certain) (continued).

	Pool Type				Lake Superior
	Lichen	***Splash***	***Ephemeral***	***Permanent***	
Microcyclops vericans					Lsup
Paracyclops (chiltoni)	L			P	
cyclopoid adult, unidentified					
Harpacticoid					
Epischura lacustris				P	
Leptodiaptomus sicilis					
Leptodiaptomus sp.		S			
Limnocalanus macrurus					Lsup
Senecella calanoides					Lsup
Skistodiaptomus oregonensis		S		P	
Skistodiaptomus reighardi	L			P	
CLADOCERA (28 taxa)					
Acroperus harpae	L			P	
Alona **sp.**					
Alona bicolor	L			P	
Alona circumfimbriata	L				
Alona costata				P	
Alona gutatta				P	
Alona quadrangula		S		P	
Alona rectangula				P	
Alonella nana				P	
Biapertura (Alona) affinis					
Chydorus **sp.**					
Chydorus faviformis					
Chydorus sphaericus					
Eurycercus longirostris	L			P	
Kurzia (latissima)	L			P	
(Paralona pigra)	L			P	
Ceriodaphnia sp.					
Ceriodaphnia lacustris				P	
Ceriodaphnia quadrangula					
Diaphanosoma sp.	L				
Simocephalus sp.	L				
Scapholeberis mucronata					
Bosmina longirostrus		S	E		
Bosmina spp.	L			P	

Table 15. Zooplankton species or taxa unique to either lichen vs splash pools, permanent vs ephemeral pools, or Lake Superior (vs pools) at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore. Species in **bold** font showed no affinities and were found in lichen and splash zones and in both permanent and ephemeral pools. Names in parentheses are tentative identifications (75%–95% certain) (continued).

	Pool Type				Lake Superior
	Lichen	***Splash***	***Ephemeral***	***Permanent***	
Daphnia ambigua	L		E		
Daphnia pulex catawba	L		E		
Daphnia mendotae dentifera	L		E		
Holopedium gibberum			E		
ROTIFERA (96 taxa)					
Bdelloid rotifer					
***Adineta* sp.**					
Anuraeopsis fissa					
Ascomorpha sp.				P	
Asplanchna herricki					
Asplanchna priodonta					
***Asplanchna* sp.**					
Cephalodella sp.				P	
Collotheca mutabilis					Lsup
Collotheca pelagica					
***Colurella* sp.**					
***Conochilus* sp.**					
Conochilus hippocrepis		S	E		
Conochilus unicornis					
Conochiloides dossarius					
***Dicranophorus* sp.**					
Dissotrocha sp.	L			P	
***Encentrum* sp.**					
Euchlanis calpidia		S			
Euchlanis dilatata					
Euchlanis triquetra					
Euchlanis spp.		S			
Gastropus stylifer					
Habrotrocha sp.		S		P	
Harringia sp.	L				
Hexarthra mira		S		P	
Kellicottia bostoniensis		S			
Kellicottia longispina					
Keratella cochlearis cochlearis					
Keratella cochlearis robusta		S	E		
Keratella cochlearis tecta		S			

Table 15. Zooplankton species or taxa unique to either lichen vs splash pools, permanent vs ephemeral pools, or Lake Superior (vs pools) at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore. Species in **bold** font showed no affinities and were found in lichen and splash zones and in both permanent and ephemeral pools. Names in parentheses are tentative identifications (75%–95% certain) (continued).

	Pool Type				
	Lichen	***Splash***	***Ephemeral***	***Permanent***	**Lake Superior**
Keratella crassa	L		E		
Keratella earlinae					
Keratella hiemalis		S	E		
Keratella quadrata		S		P	
Lecane candida	L		E		
Lecane crepida	L			P	
Lecane flexilis					
Lecane inermis					
Lecane luna					
Lecane mira					
Lecane mucronata		S		P	
Lecane ovalis		S	E		
Lecane stenroosi		S	E		
Lecane (tenuiseta)					
Lecane tudicola		S		P	
Lepadella patella					
Lepadella ovalis					
Lepadella triptera		S		P	
Lophocharis sp.		S		P	
Monostyla sp.	L			P	
Monostyla bulla					
Monostyla closterocerca					
Monostyla copeis					
Monostyla cornuta		S		P	
Monostyla crenata	L			P	
Monostyla lunaris					
Monostyla obtusa				P	
Monostyla quadridentata	L			P	
Mytilina ventralis		S		P	
Notholca acuminata		S			
Notholca caudata					Lsup
Notholca labis		S		P	
Notholca laurentiae					
Notholca squamula				P	
Notholca sp.	L			P	
Notomata sp.		S			
***Philodina* sp.**					

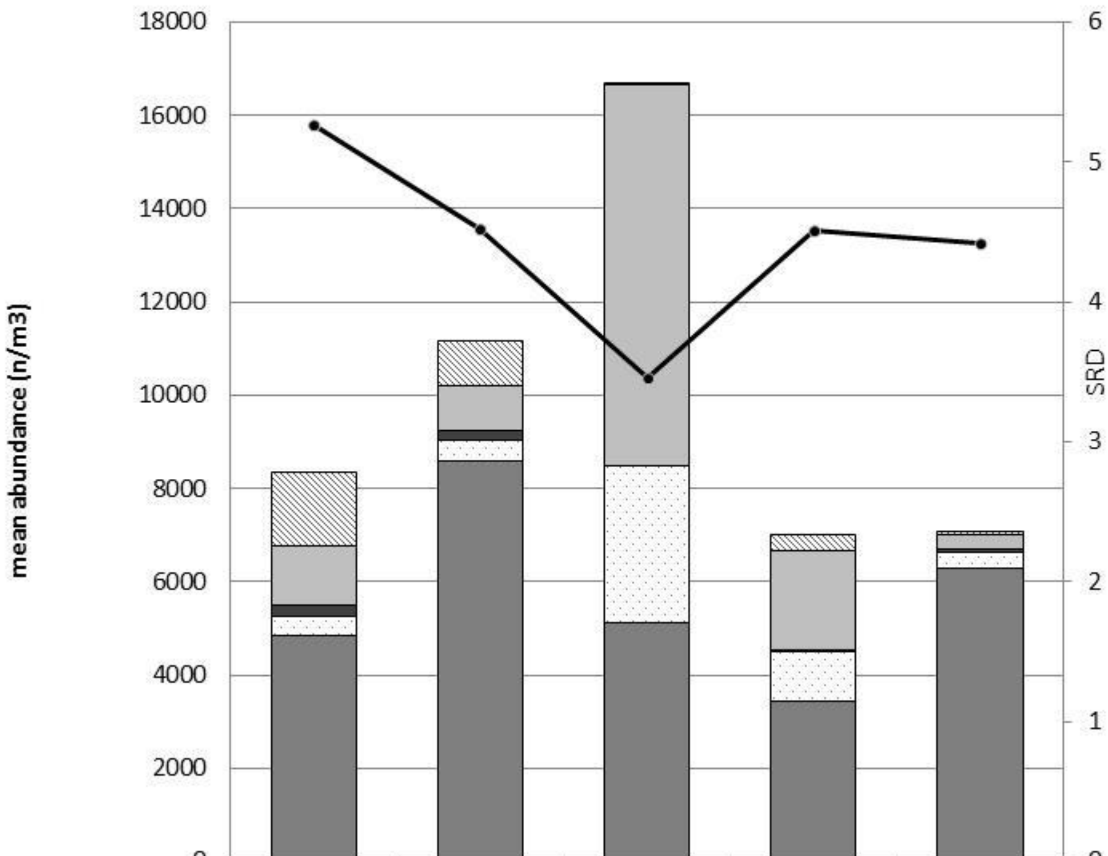
Table 15. Zooplankton species or taxa unique to either lichen vs splash pools, permanent vs ephemeral pools, or Lake Superior (vs pools) at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore. Species in **bold** font showed no affinities and were found in lichen and splash zones and in both permanent and ephemeral pools. Names in parentheses are tentative identifications (75%–95% certain) (continued).

	Pool Type				Lake Superior
	Lichen	***Splash***	***Ephemeral***	***Permanent***	
Ploesoma sp.		S			
Ploesoma hudsoni	L				
Ploesoma truncata					Lsup
Polyarthra dolichoptera		S			
Polyarthra major		S		P	
Polyarthra remata					
Polyarthra vulgaris		S			
***Polyarthra* spp.**					
Pompholyx sulcata					
***Proales* sp.**					
Rotaria sp.	L			P	
Schwabia sp.		S		P	
***Synchaeta* sp.**					
Synchaeta grandis					Lsup
Synchaeta kitina					Lsup
Synchaeta tremula					Lsup
***Squatinella* sp.**					
Testudinella sp.		S		P	
Trichocerca caputina		S		P	
Trichocerca cylindrica				P	
Trichocerca elongata	L			P	
Trichocerca iernis		S		P	
Trichocerca porcellus	L			P	
Trichocerca pusilla					
Trichocerca rousseleti	L			P	
Trichotria tetractis					
Wierzyskiella velox					
unidentified rotifer					
TESTATE PROTISTS (23 taxa)					
Arcella gibbosa					
Arcella sp.	L			P	
Centropyxis constricta aerophila					
Centropyxis constricta spinosa					
***Codonella* sp.**					
Cucurbitella (tricuspis)		S		P	

Table 15. Zooplankton species or taxa unique to either lichen vs splash pools, permanent vs ephemeral pools, or Lake Superior (vs pools) at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore. Species in **bold** font showed no affinities and were found in lichen and splash zones and in both permanent and ephemeral pools. Names in parentheses are tentative identifications (75%–95% certain) (continued).

	Pool Type				
	Lichen	***Splash***	***Ephemeral***	***Permanent***	**Lake Superior**
Difflugia bacillaliarum		S		P	
Difflugia (lucida)	L		E		
Difflugia (oblonga)				P	
Difflugia urceolata		S		P	
***Difflugia* sp.**					
***Euglypha* sp.**					
***Geopyxella* sp.**					
Hyalosphenia papilio	L			P	
Lesquereusia spiralis	L			P	
Nadinella sp.	L			P	
Nebellidae				P	
Phryganella sp.		S		P	
Trinema sp.	L			P	
(Wailesella eboracensis)					
unidentified testate					
unidentified protist					
OSTRACODA (7 taxa)					
Candoninae				P	
Cypridopsinae	L			P	
Cypridopsis sp.	L			P	
Potamocypris unicaudata				P	
Potamocypris sp.	L				
Scottia pseudobrowniana (?)				P	
unidentified ostracod					
juvenile ostracod					
OTHER					
Hydrachnidiae					
Tardigrada	L			P	
Collembola	L		E		

A second pattern emerges from the distribution of major taxonomic groups of zooplankton (Figure 23). As above, the high standard deviation, which is greater than the mean, makes statistical comparison difficult. Qualitatively three features are notable: 1) the permanent lichen pools supported the highest diversity, 2) testate protists were more abundant in Lake Superior than in the rock pools, and 3) rotifers dominated pool abundance in ephemeral pools. These relationships are statistically significant if the data set is limited to individual parks instead of lumped (below), indicating that regional variation may be overwhelming local ecological factors that structure the community. One important relationship is significant at the regional level. The cladoceran zooplankton were strongly associated with the lichen zone ($F(5, 112) = 2.127$, $p = 0.069$). Species of the common cladoceran *Daphnia* were only found in ephemeral lichen zone pools, a surprising result given their dominance in many deep pelagic systems (see Table 15). Other cladocerans characteristic of littoral areas were found in the more permanent lichen zone pools.



	lichen zone, permanent	lichen zone, ephemeral	Lake Superior, nearshore	splash zone, permanent	splash zone, ephemeral
Cladocera	1580	974	37	358	74
Copepoda	1278	965	8187	2130	318
Ostracoda	249	175	0	40	53
Protista (testate)	408	473	3350	1069	361
Rotifera	4846	8594	5137	3438	6289
SRD	5.27	4.52	3.46	4.52	4.42

Figure 23. Zooplankton abundance, by major taxonomic group, compared across pool habitat types from Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010. Simpson's reciprocal diversity is also shown (line), a measure of diversity and evenness.

While there was a nearly even distribution of zooplankton taxa between lichen and splash zone pools, pool permanence appeared to shape the most unique zooplankton community. Seventy-five zooplankton species were common to lichen and splash pools (Figure 24), about the same that were common between permanent and ephemeral pools (Figure 25). However, 66 species were unique to permanent pools and 15 were unique to ephemeral pools, compared to a more even spread of 42 unique to lichen and 40 to splash zones. In general, pool zone and permanence both structured the zooplankton community composition, though a large number of shared species appeared to colonize any of the pools.

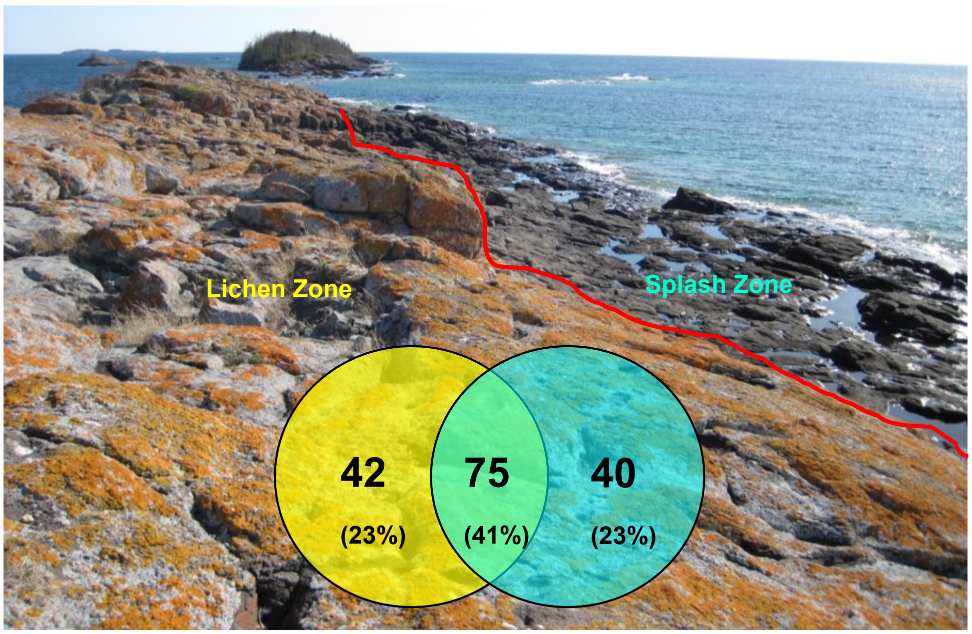


Figure 24. Zonal distribution of zooplankton community at Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010. Stratification is based on pools from two zones (lichen in yellow/left, splash in blue/right; red line separates the zones). Venn diagram shows number of genera exclusive to each zone and shared between zones. Percent of taxa out of total taxa will not add to 100% due to exclusion of unidentified species.

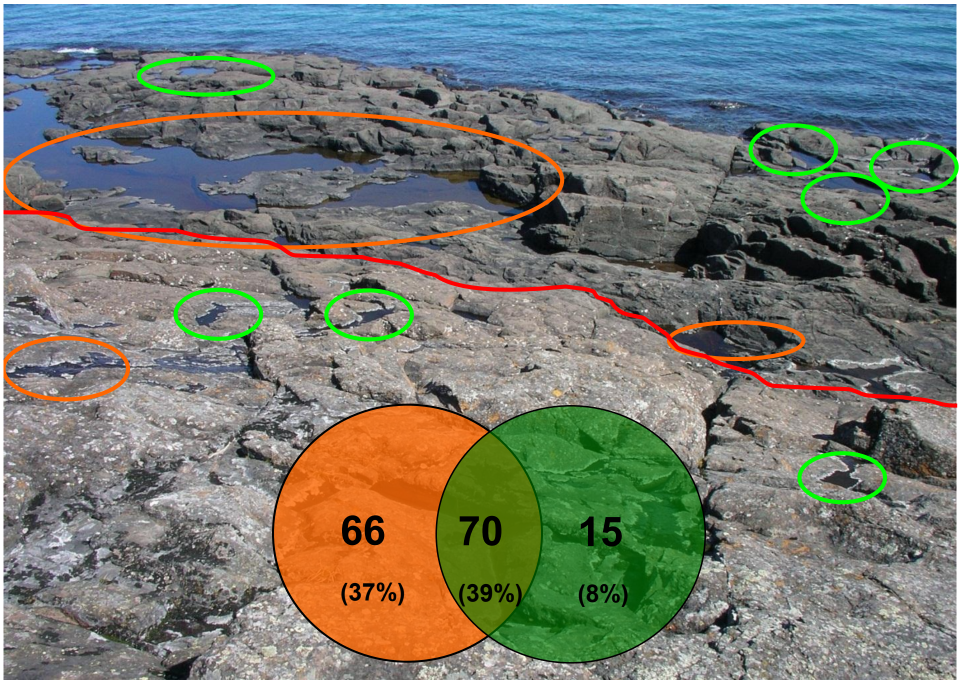


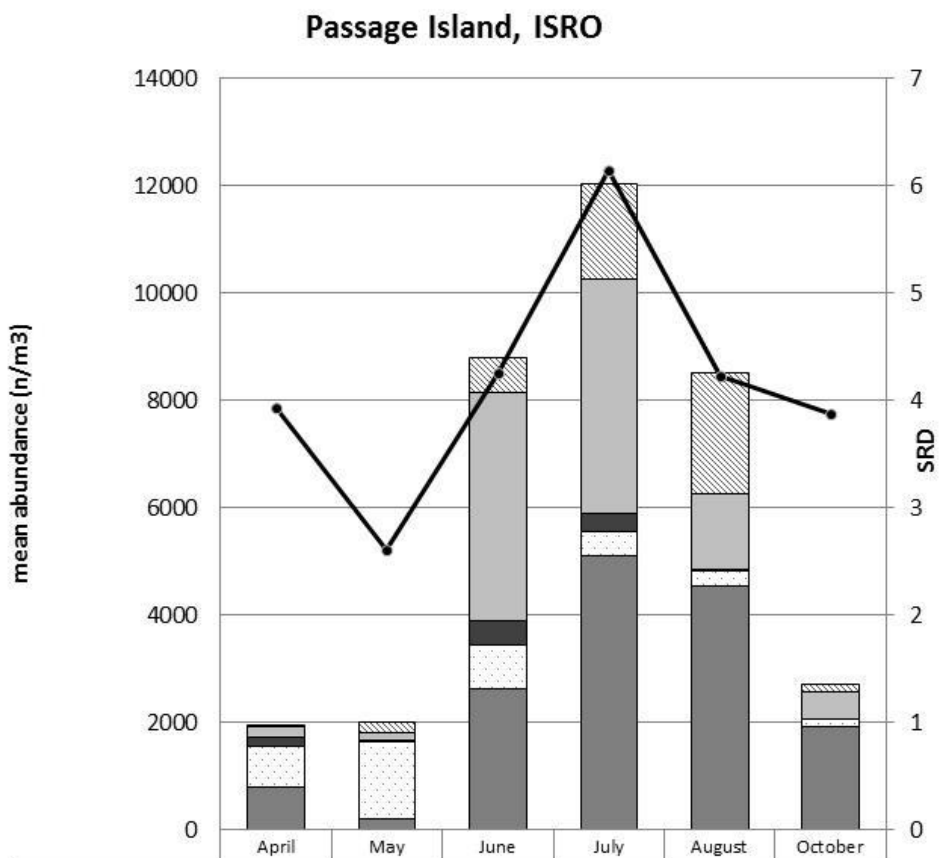
Figure 25. Stratification of zooplankton community based on pool permanence at Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010. Permanent pools are shown in orange/left, ephemeral pools in green/right; red line separates the zones.The number indicates unique or shared species with percent of total species unique or shared below. Percent of taxa out of total taxa will not add to 100% due to exclusion of unidentified species.

Zooplankton Communities of Passage Island, Isle Royale National Park

The most complete zooplankton data (collected monthly through the entire 2010 sampling period) are from Passage Island, ISRO. While sampling at APIS captured a period of high productivity, those pools were not sampled monthly. The physical structure of the rock pool systems at PIRO was unique and did not lend itself to study design. The one site at PIRO that was structured like the other systems had its splash zone pools buried in sand during the September visit. The 2010 ISRO sites were selected in a stratified random manner to ensure sites from different geographic areas in the park were included. Passage Island is a distant island near an international shipping lane, has a high density of pools (45,164 pools were mapped in 2011, which is 63% of all pools between Passage and Malone Bay on the south shore of ISRO), and has a unique coastal plant community. Consquently, Passage Island was chosen as its own stratum within the random sampling design.

Although assemblages differ across parks, the community dynamics at Passage Island may be representative of the other study locations and clarify the factors that structure the zooplankton ecology. Seasonally, the lowest overall abundance occurred in April and May, with a dip in species evenness in May due to dominance of testate protozoa (Figure 26). Taxa abundance and diversity peaked in July due to a balanced increase of rotifers, copepods, and cladocera, with a decrease in October to spring levels but with rotifer rather than testate protist dominance.

The seasonal trends in zooplankton community composition and abundance are statistically significant. Species richness was significantly higher in July than all other months ($F(5, 40) = 5.875$, $p < 0.001$). Cladocera, copepods, and ostracods were all significantly more abundant in July and August (respectively, $F(5,40) = 4.461$, $p = 0.004$; $F(5,40) = 2.902$, $p = 0.029$; and $F(5,40) = 3.329$, $p = 0.016$). Rotifers were significantly less abundant in May ($F(5,40) = 7.717$, $p < 0.001$).

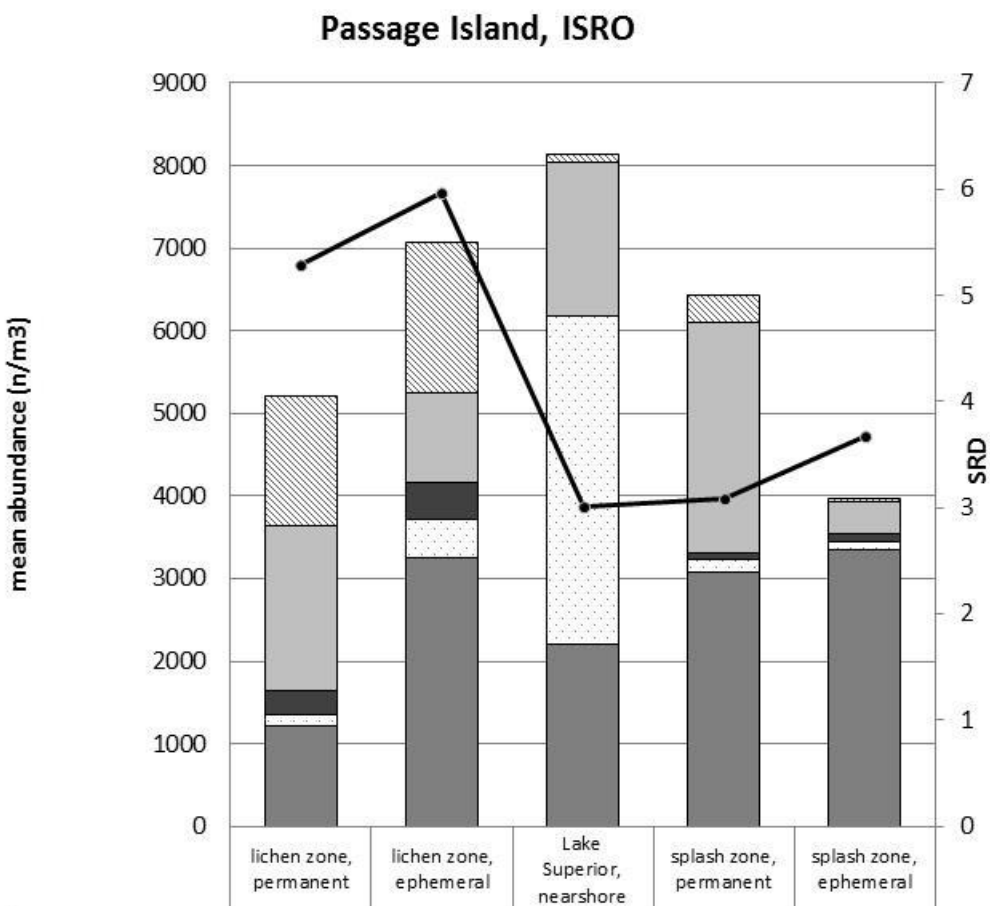


	April	May	June	July	August	October
Cladocera	23	182	636	1773	2254	136
Copepoda	205	159	4249	4386	1405	500
Ostracoda	182	23	477	318	27	0
Protista (testate)	750	1432	795	454	292	136
Rotifera	795	205	2636	5113	4534	1932
SRD	3.93	2.61	4.25	6.15	4.23	3.88

Figure 26. Seasonal trends of zooplankton composition, diversity, and abundance at Passage Island, Isle Royale National Park, 2010. Line indicates Simpson's Reciprocal Diversity, a measure of diversity and evenness.

Significant differences existed in zooplankton taxonomic groups among pool types on Passage Island (Figure 27). Lake Superior had the highest total abundance but the least amount of evenness (SRI). Species evenness-weighted diversity was significantly higher in lichen pools, both permanent and ephemeral ($F(4,40) = 2.206$, $p = 0.091$). Community composition was also different among pool types, with cladoceran abundance significantly higher in permanent and ephemeral lichen pools

(F4,40) = 9.184, $p < 0.001$). Finally, testate protists were significantly more abundant in Lake Superior than in any of the rock pools ($F(5,40) = 10.779$, $p < 0.001$).



	lichen zone, permanent	lichen zone, ephemeral	Lake Superior, nearshore	splash zone, permanent	splash zone, ephemeral
Cladocera	1577	1813	95	331	45
Copepoda	1988	1082	1877	2797	386
Ostracoda	292	445	0	66	91
Protista (testate)	146	477	3977	159	114
Rotifera	1206	3245	2195	3075	3340
SRD	5.30	5.97	3.01	3.08	3.67

Figure 27. Mean zooplankton abundance against pool type, with Simpson's Reciprocal Index, for Passage Island, Isle Royale National Park, 2010.

The factors that structure the zooplankton community in the rock pools of Passage Island are representative of the other sites, seen qualitatively in trends for those sites. Statistical power (1-β) is too low with the larger data set to detect the differences seen in graphic representations. Results from Passage Island indicate that for management and monitoring purposes, site-specific scale will most clearly detect processes with the most direct impact on zooplankton distribution and abundance

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Diatoms

Genera Richness and Pool Communities

The richness of diatom genera varied slightly among parks. A total of 82 diatom genera plus chrysophyte cysts were identified from pools in the three parks (Table 16). The highest diatom taxa richness (68 genera) was found at PIRO, likely a result of the varying types of pools. At PIRO, 57 genera were identified from lichen pools, 45 genera from splash pools, and 43 genera from other pools (Tables 16 and 17). Sixty-one taxa were identified from APIS pools, with 53 genera present in lichen pools and 49 genera present in splash pools (Tables 16 and 17). ISRO taxa richness totaled 59 genera, including 45 genera in lichen pools and 40 genera in splash pools (Tables 16 and 17). Rock pool diatom communities also varied among parks and among pool types. Rock pool communities within parks and among pool types were characterized by their dominant taxa (>5% abundance) (Table 18).

Table 16. Diatom genera and subgroups identified in rock pool samples from Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010, including representative species for each genus.

Genus or taxonomic group	Code	ISRO	APIS	PIRO	Description and representative species
Chrysophyte cysts	Cyst	x	x	x	All chrysophyte cyst morphotypes grouped
Achnanthidium	Achn	x	x	x	*Achnanthidium minutissimum* and vars., *A. exiguum*
Adlafia	Adla			x	*Adlafia bryophila*
Amphipleura	Ampl		x	x	*Amphipleura pellucida*
Amphora	Amph	x	x	x	*Amphora perpusilla, A. ovalis, A. libyca, A. fogediana*
Aneumastus	Aneu		x	x	*Aneumasus minor, A. tusculus*
Asterionella	Aste	x	x		*Asterionella formosa*
Aulacoseira	Aula	x	x	x	*Aulacoseira islandica, A. ambigua, A. alpigena*
Brachysira	Brac	x	x	x	*Brachysira microcephala, B. rossii*
Caloneis	Calo	x	x	x	*Caloneis bacillum*
Cavinula	Cavi		x	x	*Cavinula pseudoscutiformis, C. scutelloides*
Chamaepinnularia	Cham	x	x	x	*Chamaepinnularia hassiaca*
Cocconeis	Cocc	x	x	x	*Cocconeis placentula* and vars.
Cyclotella	Cycl	x	x	x	*Cyclotella comensis, C. delicatula, C. ocellata*
Cymbella	Cymb	x	x	x	*Cymbella cistula, C. proxima, C. affinis*
Cymbopleura	Cymp		x	x	*Cymbopleura subcuspidata*
Decussata	Decu			x	*Decussata placenta*
Delicata	Deli	x	x	x	*Delicata delicatula*, undescribed species
Denticula	Dent	x	x	x	*Denticula tenuis*
Diadesmis	Diad	x	x	x	*Diadesmis contenta*
Diatoma	Diat	x	x	x	*Diatoma tenue, D. vulgare, D. ehrenbergii*
Diatoma mesodon	Dime			x	*Diatoma mesodon*
Diploneis	Dipl	x	x	x	*Diploneis marginestriata*
Discostella	Disc	x	x		*Discostella stelligera, D. pseudostelligera*
Distrionella	Dist		x		*Distrionella incognita*
Encyonema	Ency	x	x	x	*Encyonema minutum*

Table 16. Diatom genera and subgroups identified in rock pool samples from Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010, including representative species for each genus (continued).

Genus or taxonomic group	Code	ISRO	APIS	PIRO	Description and representative species
Encyonopsis	Encp	x	x	x	*Encyonopsis microcephala*
Eolimna	Eoli	x		x	*Eolimna minima*
Epithemia	Epit	x	x	x	*Epithemia cistula, E. smithii*
Eucocconeis	Euco	x	x	x	*Eucocconeis flexella, E. laevis*
Eunotia	Euno	x	x	x	*Eunotia bilunaris, E. rhombica*
Fistulifera	Fist			x	*Fistulifera pelliculosa*
Fragilaria (plankton)	Frpl	x	x	x	True planktonic *Fragilaria* species, *Fragilaria crotonensis, F. capucina*
Fragilariforma	Frfo			x	*Fragilariforma virescens* and vars.
Frustulia	Frus	x	x	x	*Frustulia saxonica*
Geissleria	Geis		x	x	*Geissleria schoenfeldtii*
Gomphonema	Gomp	x	x	x	*Gomphonema angustatum, G. geitleri, G. intricatum, G. gracile*
Halamphora	Hala			x	*Halamphora veneta*
Hannaea	Hann	x			*Hannaea superiorensis*
Hantzschia	Hant				*Hantzschia amphioxys*
Hippodonta	Hipp			x	*Hippodonta capitata, H. hungarica*
Karayevia	Kara	x	x	x	*Karayevia clevei, K. laterostrata, K. amoena*
Kobayasiella	Koba	x	x	x	*Kobayasiella subtilissima*
Krasskella	Kras	x		x	*Krasskella kriegeriana*
Luticola	Luti	x	x		*Luticola mutica*
Martyana	Mart		x	x	*Martyana martyii*
Mastogloia	Mast			x	*Mastogloia lacustris, M. grevillei*
Meridion	Meri			x	*Meridion circulare*
Microcostatus	Micr	x			*Microcostatus krasskei*
Navicula	Navi	x	x	x	*Navicula radiosa, N. cryptotenella*
Navicula (small)	Nvsm		x	x	Group includes the many small "naviculoid" taxa whose generic placement is uncertain
Navicula schmassmannii	Nschm	x		x	*Navicula schmassmannii*
Neidiopsis	Neip	x			*Neidiopsis*
Neidium	Neid	x	x	x	*Neidium ampliatum, N. cf. hankensis*
Nitzschia	Nitz	x	x	x	*Nitzschia dissipata,*
Nitzschia (plankton)	Nzpl	x	x	x	Group includes several *Nitzschia* species most often found in Great Lakes plankton
Nupela	Nupe		x		*Nupela vitiosa, N. impexiformis*
pennate GV unid	Pund	x	x	x	Group includes girdle views of pennate taxa that could not be determined to genus
Pinnularia	Pinn	x	x	x	*Pinnularia biceps* group
Planothidium	Plan	x		x	*Planothidium dubium, P. frequentissimum*
Platessa	Plat		x	x	*Platessa conspicua*
Psammothidium	Psam	x	x	x	*Psammothidium altaicum*
Pseudostaurosira	Psst	x	x		*Pseudostaurosira brevistriata, P. microstriata, P. elliptica, P. parasitica*

Table 16. Diatom genera and subgroups identified in rock pool samples from Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010, including representative species for each genus (continued).

Genus or taxonomic group	Code	ISRO	APIS	PIRO	Description and representative species
Puncticulata (large)	Punl	x	x	x	*Puncticulata bodanica*
Puncticulata (small)	Puns		x	x	*Puncticulata radiosa, P. comta*
Reimeria	Reim	x	x	x	*Reimeria sinuata*
Rhopalodia	Rhop	x		x	*Rhopalodia gibba*
Rossithidium	Ross	x	x	x	*Rossithidium linearis, R. pusillum, R duthii*
Sellaphora	Sell	x	x	x	*Sellaphora pupula* vars., *S. vitabunda*
Stauroforma	Stfo	x			*Stauroforma exigua*
Stauroneis	Stau	x			*Stauroneis cf. anceps*
Staurosira	Stsa	x	x	x	*Staurosira venter, S. construens*
Staurosirella	Stsl	x	x	x	*Staurosirella pinnata, S. lapponica, S. leptostauron*
Stenopterobia	Sten	x			*Stenopterobia curvula*
Stephanodiscus (large)	Stel	x	x	x	*Stephanodiscus niagarae, S. theriotensis*
Stephanodiscus (small)	Stes	x	x	x	*Stephanodiscus transylvanicus, minutus, parvus, hantzschii*
Surirella	Suri			x	*Surirella angusta, S. Iowensis*
Synedra	Synd	x	x	x	Group includes unicellular and rosette colony formers, *S. rumpens, S. subrhombica, Fragilaria capucina* v. *amphicephala, F. capucina* v. *austriaca*
Synedra cyclopum	Sync		x	x	*Synedra cyclopum*
Tabellaria (long)	Tabl	x	x	x	*Tabellaria quadriseptata, T. fenestrata, T. flocculosa IIIp*
Tabellaria (small)	Tabs	x	x	x	*Tabellaria flocculosa IV*
Ulnaria	Ulna	x	x	x	*Ulnaria ulna, U. ulna* var. *danica, U. ulna* var. *chaseana*
Urosolenia	Uros	x	x	x	*Urosolenia eriensis*

Table 17. Mean percent abundance of diatom genera by pool type in Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010.

		ISRO		APIS		PIRO			
Taxon	**Code**	***Lichen***	***Splash***	***Lichen***	***Splash***	***Lichen***	***Splash***	***Cave***	***Medicolous***
Chrysophyte cysts	Cyst	2.99	0.18	10.25	0.20	0.75	0.04	0.00	0.10
Achnanthidium	Achn	22.54	21.67	27.78	31.68	38.27	30.94	54.57	47.73
Adlafia	Adla	0.00	0.00	0.00	0.00	0.08	0.04	0.00	0.09
Amphipleura	Ampl	0.00	0.00	0.02	0.00	0.00	0.00	0.10	0.00
Amphora	Amph	0.01	0.02	0.22	0.32	0.32	0.31	0.00	0.00
Aneumastus	Aneu	0.00	0.00	0.02	0.00	0.00	0.08	0.00	0.00
Asterionella	Aste	0.00	0.05	0.13	0.17	0.00	0.00	0.00	0.00
Aulacoseira	Aula	0.00	0.90	0.89	0.00	0.06	0.08	0.00	0.00
Brachysira	Brac	15.26	2.31	2.68	8.25	1.47	3.15	0.00	8.28
Caloneis	Calo	0.04	0.01	0.04	0.00	0.11	0.00	0.09	0.18
Cavinula	Cavi	0.00	0.00	0.10	0.00	0.04	0.00	0.10	0.00
Chamaepinnularia	Cham	0.06	0.00	0.10	0.07	0.49	0.00	0.00	0.18
Cocconeis	Cocc	0.00	0.06	0.12	0.05	0.09	0.04	0.00	0.00
Cymbella	Cymb	2.20	5.55	0.57	0.49	2.18	3.75	0.67	6.02
Cyclotella	Cycl	0.07	3.93	5.77	3.69	0.55	1.04	0.74	0.00
Cymbopleura	Cymp	0.00	0.00	0.05	0.02	0.42	0.00	0.00	0.00
Decussata	Decu	0.00	0.00	0.00	0.00	0.04	0.00	0.00	0.00
Delicata	Deli	0.05	30.56	4.19	15.07	3.10	15.42	5.88	1.32
Denticula	Dent	0.00	1.05	2.41	0.78	0.26	0.51	0.29	0.19
Diadesmis	Diad	0.01	0.00	0.10	0.00	0.02	0.00	0.00	0.00
Diatoma	Diat	0.03	0.02	0.17	0.44	0.02	0.08	0.00	0.00
Diatoma mesodon	Dime	0.00	0.00	0.00	0.00	0.00	0.00	0.00	0.28
Diploneis	Dipl	0.00	0.00	0.02	0.00	0.00	0.04	0.00	0.00
Discostella	Disc	0.02	0.17	0.00	0.02	0.00	0.00	0.00	0.00
Distrionella	Dist	0.00	0.00	0.00	0.02	0.00	0.00	0.00	0.00
Encyonema	Ency	7.21	2.23	1.63	1.55	2.56	4.48	1.69	0.27
Encyonopsis	Encp	3.86	19.48	10.68	10.46	7.42	18.62	7.56	3.11
Eolimna	Eoli	0.03	0.01	0.00	0.00	0.09	0.00	3.51	0.00

Table 17. Mean percent abundance of diatom genera by pool type in Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010 (continued).

		ISRO		APIS		PIRO			
Taxon	**Code**	***Lichen***	***Splash***	***Lichen***	***Splash***	***Lichen***	***Splash***	***Cave***	***Medicolous***
Epithemia	Epit	0.50	0.00	0.00	0.02	4.38	0.00	0.00	0.46
Eucocconeis	Euco	0.02	0.29	0.29	0.64	0.04	0.80	0.09	0.10
Eunotia	Euno	0.57	0.02	4.39	0.25	0.42	0.08	0.39	0.00
Fistulifera	Fist	0.00	0.00	0.00	0.00	0.04	0.00	0.00	0.00
Fragilaria (plankton)	Frpl	0.05	0.21	0.26	0.34	0.04	0.00	0.00	0.00
Fragilariforma	Frfo	0.00	0.00	0.00	0.00	1.11	0.00	0.00	0.00
Frustulia	Frus	0.27	0.00	0.04	0.10	0.32	0.00	0.00	0.00
Geissleria	Geis	0.00	0.00	0.18	0.02	0.04	0.00	0.18	0.00
Gomphonema	Gomp	1.36	0.80	0.64	0.63	6.51	3.06	0.58	6.99
Halamphora	Hala	0.00	0.00	0.00	0.00	0.00	0.00	0.18	0.00
Hannaea	Hann	0.00	0.08	0.00	0.00	0.00	0.00	0.00	0.00
Hantzschia	Hant	0.00	0.00	0.00	0.00	0.00	0.00	0.00	0.00
Hippodonta	Hipp	0.00	0.00	0.00	0.00	0.00	0.04	0.00	0.00
Karayevia	Kara	0.00	0.01	0.11	0.12	0.06	0.00	0.00	0.10
Kobayasiella	Koba	0.26	0.40	0.06	0.52	1.25	0.91	3.77	0.19
Krasskella	Kras	0.10	0.05	0.00	0.00	0.00	0.00	0.00	0.09
Luticola	Luti	0.01	0.00	0.00	0.02	0.00	0.00	0.00	0.00
Martyana	Mart	0.00	0.00	0.05	0.00	0.00	0.08	0.18	0.00
Mastogloia	Mast	0.00	0.00	0.00	0.00	0.92	0.00	0.00	0.00
Meridion	Meri	0.00	0.00	0.00	0.00	0.28	0.08	0.00	0.47
Microcostatus	Micr	0.01	0.00	0.00	0.00	0.00	0.00	0.00	0.00
Navicula	Navi	1.98	0.59	1.92	2.09	1.59	1.32	2.28	0.28
Navicula (small)	Nvsm	0.00	0.00	0.00	0.05	0.02	0.00	0.00	0.00
Navicula schmassmannii	Nschm	0.06	0.00	0.00	0.00	4.90	0.15	0.00	0.46
Neidiopsis	Neip	0.01	0.00	0.00	0.00	0.00	0.00	0.00	0.00
Neidium	Neid	0.03	0.00	0.11	0.02	0.02	0.11	0.00	0.00
Nitzschia	Nitz	25.07	0.83	4.00	2.97	6.39	0.97	10.64	0.47
Nitzschia (plankton)	Nzpl	0.04	0.00	0.12	0.29	0.00	0.04	0.00	0.00
Nupela	Nupe	0.00	0.00	0.00	0.05	0.00	0.00	0.00	0.00

Table 17. Mean percent abundance of diatom genera by pool type in Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010 (continued).

		ISRO		**APIS**		**PIRO**			
Taxon	**Code**	***Lichen***	***Splash***	***Lichen***	***Splash***	***Lichen***	***Splash***	***Cave***	***Medicolous***
pennate GV unid	Pund	0.00	0.04	0.37	0.71	0.00	0.32	0.75	0.10
Pinnularia	Pinn	0.08	0.00	0.25	0.00	0.46	0.00	0.00	0.00
Planothidium	Plan	0.00	0.01	0.00	0.00	0.08	0.11	0.10	0.00
Platessa	Plat	0.00	0.00	0.02	0.00	0.06	0.00	0.00	0.00
Psammothidium	Psam	0.17	0.00	7.64	0.22	1.83	0.61	0.29	0.10
Pseudostaurosira	Psst	0.52	0.00	0.05	0.07	0.00	0.00	0.00	0.00
Puncticulata (large)	Punl	0.00	0.02	0.06	0.07	0.02	0.00	0.00	0.00
Puncticulata (small)	Puns	0.00	0.00	0.17	0.07	0.02	0.00	0.00	0.00
Reimeria	Reim	0.00	0.01	0.05	0.02	0.02	0.04	0.18	0.00
Rhopalodia	Rhop	0.41	0.00	0.00	0.00	1.53	0.04	0.28	0.09
Rossithidium	Ross	1.28	0.02	0.17	0.15	0.15	0.04	2.15	0.46
Sellaphora	Sell	0.02	0.00	0.16	0.00	0.04	0.12	0.10	0.00
Stauroforma	Stfo	4.99	0.00	0.00	0.00	0.00	0.00	0.00	0.00
Stauroneis	Stau	0.01	0.00	0.00	0.00	0.00	0.00	0.00	0.00
Staurosira	Stsa	0.41	0.07	0.49	0.12	0.02	0.57	0.20	0.00
Staurosirella	Stsl	0.00	0.02	0.04	0.05	0.07	0.23	0.10	0.00
Stenopterobia	Sten	0.08	0.00	0.00	0.00	0.00	0.00	0.00	0.00
Stephanodiscus (large)	Stel	0.00	0.06	0.00	0.07	0.02	0.08	0.00	0.00
Stephanodiscus (small)	Stes	0.00	0.02	0.00	0.07	0.00	0.04	0.00	0.00
Surirella	Suri	0.00	0.00	0.00	0.00	0.02	0.00	0.00	0.00
Synedra	Synd	1.14	7.96	9.72	16.02	7.66	10.85	2.29	18.86
Synedra cyclopum	Sync	0.00	0.00	0.05	0.00	0.00	0.04	0.00	0.00
Tabellaria (long)	Tabl	0.83	0.11	0.45	0.61	0.02	0.27	0.09	0.18
Tabellaria (small)	Tabs	5.29	0.00	0.02	0.10	0.94	0.08	0.00	2.09
Ulnaria	Ulna	0.04	0.18	0.07	0.15	0.33	0.27	0.00	0.81
Urosolenia	Uros	0.00	0.02	0.09	0.05	0.00	0.08	0.00	0.00

Table 18. Dominant taxa (>5% abundance) of rock pool communities within parks and among pools types.

Park and Pool Type	Dominant Taxa
ISRO splash pools	*Achnanthidium, Delicata, Encyonopsis, Synedra, Cyclotella, Aulacoseira, Cymbella, Brachysira, Encyonema, Gomphonema, Denticula*
ISRO lichen pools	*Nitzschia, Brachysira, Achnanthidium, Encyonema, Stauroforma, Encyonopsis, Tabellaria (small), Navicula, Gomphonema, Rossithidium, chrysophyte cysts, Cymbella, Pseudostaurosira, Tabellaria (large), Rhopalodia, Synedra, Staurosira*
APIS splash pools	*Achnanthidium, Delicata, Brachysira, Synedra, Encyonopsis, Cyclotella, Navicula*
APIS lichen pools	*Achnanthidium, Psammothidium, chrysophyte cysts, Encyonopsis, Eunotia, Synedra, Cyclotella, Delicata, Nitzschia, Aulacoseira, Navicula, Denticula, Brachysira*
PIRO splash pools	*Achnanthidium, Delicata, Encyonopsis, Synedra, Cymbella, Brachysira, Encyonema, Gomphonema*
PIRO lichen pools	*Achnanthidium, Epithemia, Encyonopsis, Gomphonema, Synedra, Navicula schmassmannii, Nitzschia, Delicata, Rhopalodia, Psammothidium, Mastogloia, Navicula, Fragilariforma, Cymbella, Encyonema, Brachysira*
PIRO cave pool	*Achnanthidium, Nitzschia, Delicata, Encyonopsis, Eolimna, Kobayasiella*
PIRO medicolous pool	*Achnanthidium, Synedra, Brachysira, Gomphonema, Cymbella*

Accumulation Curve and Genera Estimates

The Chao 1 estimator curve was calculated for each park's samples to estimate potential diatom genera richness in rock pools. Direct counts on the 48 samples from ISRO reported 59 genera; Chao 1 suggests 61 genera with 95% confidence intervals of 59 and 71 genera. Chao 1 estimates for APIS suggest 66 potential diatom genera (95% confidence intervals of 62 to 84 genera) based on 16 samples that produced 61 genera in direct counts. Chao 1 for PIRO is 79 genera with a 95% confidence interval extending from 72 to 106 genera. Counts of 18 samples from PIRO rock pools produced 68 genera.

Rarefaction Curve

The Cole rarefaction curve for expected genera was used to determine how many genera were encountered in each pool sample. At ISRO, three sampling periods (May, July, October) each resulted in 16 samples. The expected number of genera from 16 samples was 50; from 32 samples, the expected number of genera was 56; and from 48 samples, 59 genera were expected. Eight samples were collected during two sampling events at APIS (May, September). The rarefaction curve estimated that 55 genera would be encountered after eight samples, and 61 genera would be counted after 16 samples. Two sampling events (May, August) occurred at PIRO rock pools, generating 10 and 8 samples, respectively. Cole rarefaction estimates 63 genera encountered after 10 samples and 69 genera encountered after 18 samples.

Diversity Indices

<u>Jaccard's Index</u>

Jaccard's Index was calculated for diatoms from lichen and splash zones within each park but showed no significant dissimilarity (at $P \leq 0.05$) in genus use between pool zones when all genera were included in the analysis. At ISRO, the Jaccard's index (Cjk) was 0.44, at APIS Cjk = 0.67, and at PIRO Cjk = 0.55, with significance ($P \leq 0.05$) at Cjk ≤ 0.22.

Because Jaccard's Index is based on presence-absence of genera, it does not fully account for relative use of pool zones by genera. To ascertain pool selectivity among genera, boxplots and the t-test were used to illustrate and test for significant differences of mean percent genera abundance between pool zones (parks treated separately) (Figures 28–30; also see Table 18). For each park, boxplots show taxa that were present at >5% abundance in two or more samples and genera that showed a significant t-test statistic.

Three trends in pool selectivity by diatom taxa are apparent in this analysis: taxa that are found in both pool zones, taxa that selectively inhabit lichen pools, and taxa that selectively inhabit splash pools. At ISRO the genera *Achanthidium*, *Gomphonema*, *Navicula*, and large *Tabellaria* were found in high abundance and with no selectivity between pool zones. Groups of genera that had significantly higher abundance in lichen pools included chrysophyte cysts, *Nitzschia*, *Encyonema*, *Brachysira*, *Eunotia*, *Pinnularia*, small *Tabellaria* species, *Stauroforma*, and *Rossithidium* species (Figure 28). Genera that had significantly higher abundance in ISRO splash pools included *Synedra*, *Encyonopsis*, *Denticula*, *Cyclotella*, *Delicata*, *Cymbella*, *Discostella*, *Eucocconeis*, and *Ulnaria* species.

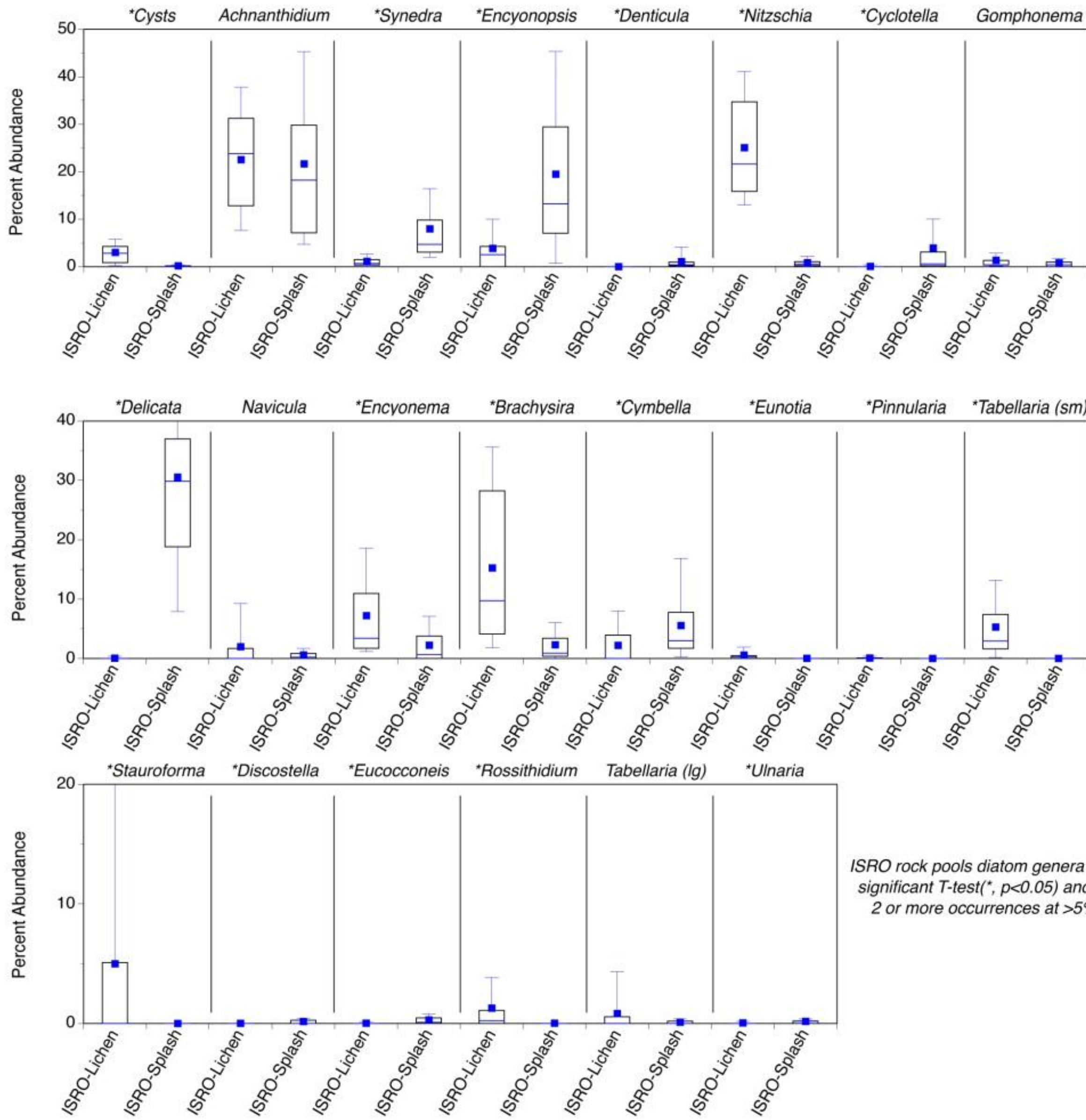


Figure 28. Boxplots of all taxa that were present at >5% abundance in two or more samples, and genera that showed a significant t-test statistic (marked with *, $p<0.05$) in lichen and splash pools at Isle Royale National Park, 2010. The top and bottom lines correspond to the 75th percentile (top quartile) and 25th percentile (bottom quartile), respectively, and the line through the middle of the box corresponds to the 50th percentile (median). Whiskers extend from the 10th percentile (bottom decile) to the 90th percentile (top decile). The blue square symbol represents the mean value.

Few genera in APIS pools showed strong trends in selectivity by pool type. Chrysophyte cysts and the diatom genus *Denticula* were the only two taxa that showed significant differences in abundance between pool types; in this case, both taxa were more abundant in lichen pools (Figure 29). Other abundant genera in APIS pools showed no significant selectivity for pool zones: *Achnanthidium*, *Synedra*, *Encyonopsis*, *Cyclotella*, *Delicata*, *Navicula*, *Brachysira*, *Psammothidium*, and *Eunotia* species.

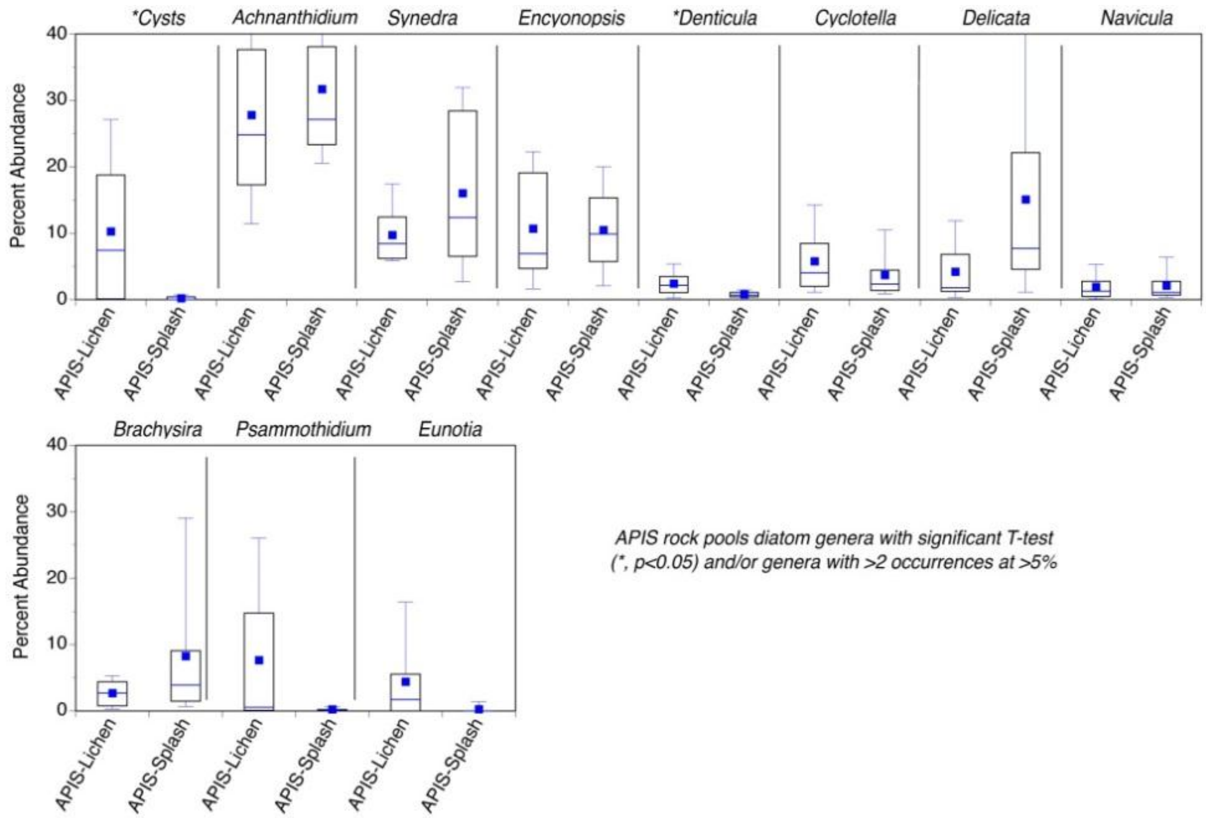


Figure 29. Boxplots of all taxa that were present at >5% abundance in two or more samples, and genera that showed a significant t-test statistic (marked with *, $p<0.05$) in lichen and splash pools at Apostle Islands National Lakeshore, 2010. See Figure 28 for description of boxplots.

Two diatom groups exhibited selectivity for lichen pools over splash pools at PIRO—*Nitzschia* spp. and *Navicula schmassmannii* (Figure 30). No diatom groups showed selectivity for splash pools at PIRO. The diatom groups *Achnanthidium*, *Synedra*, *Encyonopsis*, *Gomphonema*, *Delicata*, *Encyonema*, *Brachysira*, *Cymbella*, and *Psammothidium* species were abundant in both splash and lichen pools but showed no significant selectivity (Figure 30). Boxplots also included taxa that were abundant in PIRO's other pool types (Figure 30). The genera *Synedra*, *Brachysira*, and *Cymbella* had notable high abundance in the medicolous zone pools, whereas *Nitzschia* and *Achnanthidium* species had high relative abundance in PIRO's cave pool (Figure 30). T-tests were not calculated using the "other" pool types at PIRO.

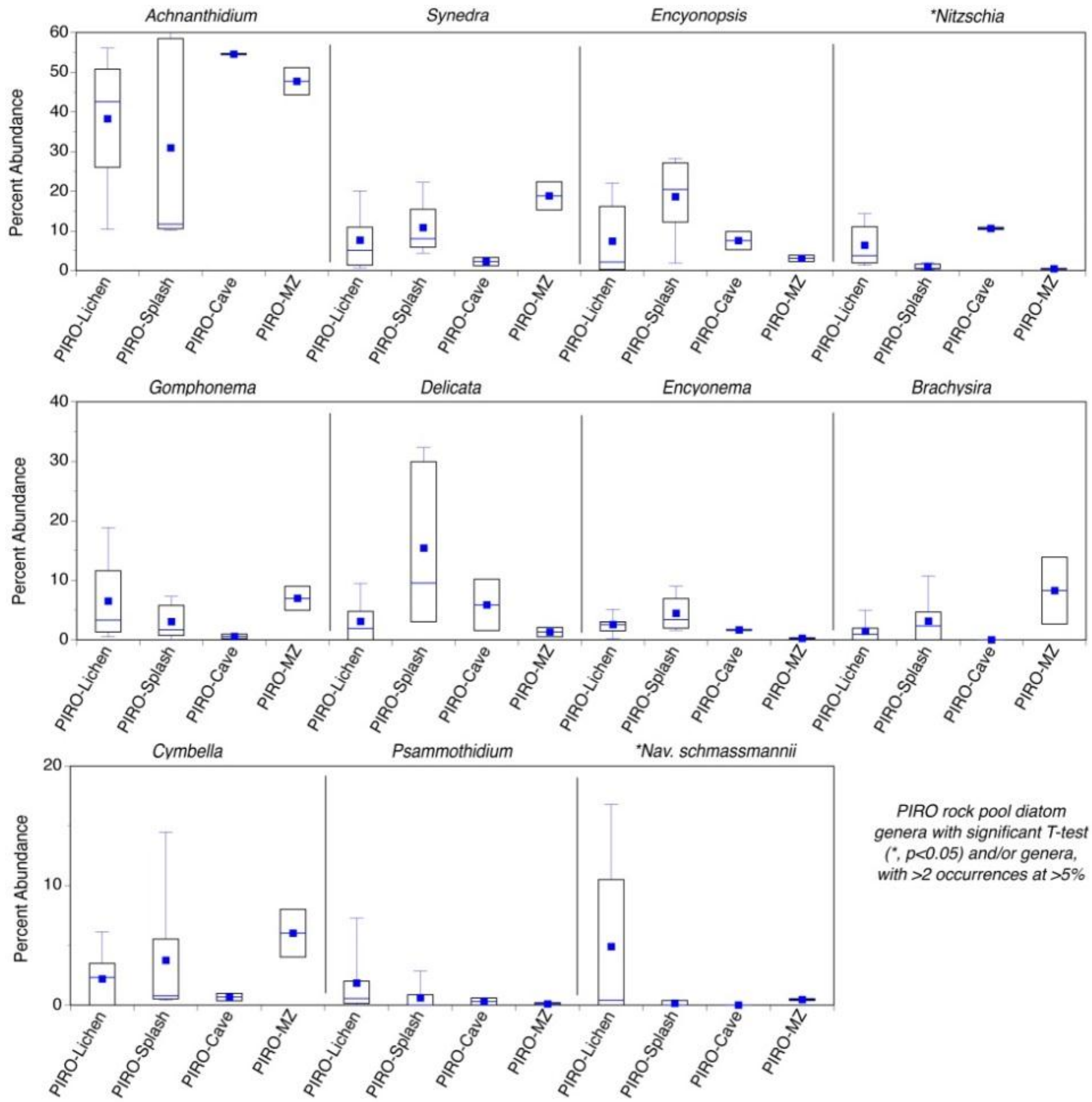


Figure 30. Boxplots of all taxa that were present at >5% abundance in two or more samples, and genera that showed a significant t-test statistic (marked with *, p<0.05) in lichen, splash, cave, and medicolous zone pools at Pictured Rocks National Lakeshore, 2010. See Figure 28 for description of boxplots.

Venn diagrams were constructed for each park to determine diatom community stratification among all genera based on pool type (Figure 31; also see Table 18). Of the 59 genera found at ISRO, 26 taxa were shared between splash and lichen pools, with eight genera found only in splash pools and 12 genera found only in lichen pools. At APIS, 41 taxa from among 61 genera were shared between splash and lichen pools; eight genera were found only in splash pools, and 12 genera were found only in lichen pools (Figure 31). Sixty-eight genera were encountered among the three pool types (lichen, splash, other) at PIRO. Thirty-two genera were shared among all pool types, 13 genera were only found in lichen pools, seven genera were found only in splash pools, and three genera were limited to PIRO's other pool types (Figure 31).

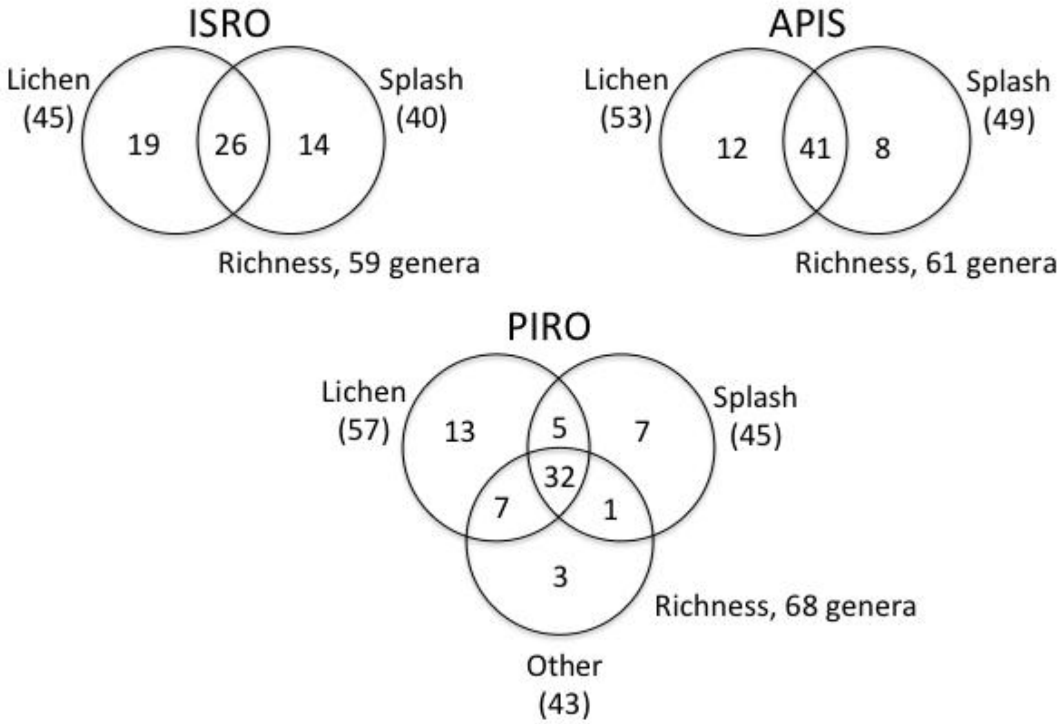


Figure 31. Diatom community stratification based on rock pool zones (lichen, splash, and other) at Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010. Venn diagrams show number of genera exclusive to each zone and shared between zones.

Simpson's Index

Simpson's Reciprocal Index (see Equation 4) and genus richness were calculated for each sampling site (Tables 19 and 20). Richness among sites in 2010 ranged from 34 taxa at ISRO's Raspberry Island to 59 genera at PIRO's AuSable Point. Among parks, ISRO's sites had the lowest richness (range 34–39 genera). Simpson's Reciprocal Index ranged from 3.18 at PIRO Mosquito Harbor to 9.03 at ISRO Blueberry Cove. Other sites had Simpson's index values between 5.7 and 7.5. Although pool sites had relatively high genus richness, dominance by a few taxa produced relatively high community unevenness and only moderate levels of diversity.

Table 19. Genus richness, Simpson's Reciprocal Index, and Shannon Diversity by pool site at Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010.

	ISRO				APIS		PIRO		
	Blueberry Cove	***Datolite Mine***	***Passage Island***	***Raspberry Island***	***Bear Island***	***Devils Island***	***AuSable Point***	***Miners Bay***	***Mosquito Harbor***
Richness	39	35	35	34	47	52	59	49	45
Simpson's Reciprocal Index (1/D)	9.03	5.70	7.48	6.87	6.04	7.52	5.72	6.35	3.18
Shannon (H')	2.50	2.22	2.32	2.25	2.24	2.48	2.36	2.39	1.90

Table 20. Mean percent abundance of diatom genera by pool site at Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010.

		Mean Percent Abundance								
		ISRO				***APIS***		***PIRO***		
Taxon	**Code**	*Blueberry Cove*	*Datolite Mine*	*Passage Island*	*Raspberry Island*	*Bear Island*	*Devils Island*	*AuSable Point*	*Miners Bay*	*Mosquito Harbor*
Chrysophyte cysts	Cyst	2.36	0.55	2.04	1.39	0.15	10.29	0.96	0.19	0.03
Achnanthidium	Achn	19.87	29.62	15.20	23.74	31.15	28.32	40.36	30.03	46.90
Adlafia	Adla	0.00	0.00	0.00	0.00	0.00	0.00	0.12	0.03	0.03
Amphipleura	Ampl	0.00	0.00	0.00	0.00	0.00	0.02	0.00	0.00	0.03
Amphora	Amph	0.00	0.00	0.02	0.05	0.17	0.37	0.22	0.52	0.00
Aneumastus	Aneu	0.00	0.00	0.00	0.00	0.02	0.00	0.03	0.00	0.03
Asterionella	Aste	0.00	0.00	0.03	0.06	0.12	0.18	0.00	0.00	0.00
Aulacoseira	Aula	0.00	0.00	0.02	1.78	0.89	0.00	0.06	0.10	0.00
Brachysira	Brac	8.71	4.17	18.59	3.68	2.18	8.75	2.84	0.19	4.55
Caloneis	Calo	0.00	0.00	0.09	0.02	0.00	0.04	0.03	0.13	0.09
Cavinula	Cavi	0.00	0.00	0.00	0.00	0.00	0.10	0.00	0.06	0.03
Chamaepinnularia	Cham	0.06	0.00	0.06	0.00	0.02	0.15	0.73	0.00	0.06
Cocconeis	Cocc	0.02	0.00	0.00	0.10	0.12	0.05	0.03	0.13	0.00
Cyclotella	Cycl	1.05	2.30	0.64	4.03	6.11	3.35	1.15	0.25	0.54
Cymbella	Cymb	8.20	3.68	1.77	1.85	0.41	0.64	0.16	5.69	2.78

Table 20. Mean percent abundance of diatom genera by pool site at Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010 (continued).

		Mean Percent Abundance								
		ISRO				***APIS***		***PIRO***		
Taxon	**Code**	*Blueberry Cove*	*Datolite Mine*	*Passage Island*	*Raspberry Island*	*Bear Island*	*Devils Island*	*AuSable Point*	*Miners Bay*	*Mosquito Harbor*
Cymbopleura	Cymp	0.00	0.00	0.00	0.00	0.07	0.00	0.60	0.03	0.00
Decussata	Decu	0.00	0.00	0.00	0.00	0.00	0.00	0.06	0.00	0.00
Delicata	Deli	17.37	10.70	13.75	19.41	16.24	3.02	5.39	10.01	4.50
Denticula	Dent	0.35	0.77	0.89	0.10	1.77	1.42	0.42	0.25	0.29
Diadesmis	Diad	0.00	0.01	0.00	0.00	0.00	0.10	0.03	0.00	0.00
Diatoma	Diat	0.00	0.03	0.00	0.06	0.10	0.51	0.07	0.03	0.00
Diatoma mesodon	Dime	0.00	0.00	0.00	0.00	0.00	0.00	0.00	0.00	0.09
Diploneis	Dipl	0.00	0.00	0.00	0.00	0.02	0.00	0.00	0.00	0.03
Discostella	Disc	0.06	0.10	0.05	0.16	0.00	0.02	0.00	0.00	0.00
Distrionella	Dist	0.00	0.00	0.00	0.00	0.02	0.00	0.00	0.00	0.00
Encyonema	Ency	4.44	1.81	1.53	11.11	2.15	1.03	3.45	2.73	2.04
Encyonopsis	Encp	10.13	9.40	20.88	6.27	14.33	6.81	3.91	15.40	10.89
Eolimna	Eoli	0.00	0.02	0.00	0.06	0.00	0.00	0.00	0.13	1.17
Epithemia	Epit	0.08	0.93	0.00	0.00	0.00	0.02	0.03	6.54	0.15
Eucocconeis	Euco	0.11	0.20	0.28	0.03	0.49	0.43	0.47	0.06	0.25
Eunotia	Euno	0.59	0.06	0.44	0.08	0.00	4.64	0.63	0.07	0.13
Fistulifera	Fist	0.00	0.00	0.00	0.00	0.00	0.00	0.00	0.06	0.00
Fragilaria (plankton)	Frpl	0.02	0.26	0.00	0.25	0.37	0.24	0.00	0.06	0.00
Fragilariforma	Frfo	0.00	0.00	0.00	0.00	0.00	0.00	1.67	0.00	0.00
Frustulia	Frus	0.08	0.02	0.45	0.00	0.00	0.14	0.47	0.00	0.00
Geissleria	Geis	0.00	0.00	0.00	0.00	0.02	0.18	0.06	0.00	0.06
Gomphonema	Gomp	2.08	0.90	0.72	0.63	0.69	0.59	5.57	6.45	2.81
Halamphora	Hala	0.00	0.00	0.00	0.00	0.00	0.00	0.00	0.00	0.06
Hannaea	Hann	0.00	0.02	0.00	0.14	0.00	0.00	0.00	0.00	0.00
Hantzschia	Hant	0.00	0.00	0.00	0.00	0.00	0.00	0.00	0.00	0.00
Hippodonta	Hipp	0.00	0.00	0.00	0.00	0.00	0.00	0.00	0.03	0.00
Karayevia	Kara	0.00	0.00	0.00	0.02	0.12	0.11	0.03	0.06	0.03

Table 20. Mean percent abundance of diatom genera by pool site at Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010 (continued).

		Mean Percent Abundance								
		ISRO				***APIS***		***PIRO***		
Taxon	**Code**	*Blueberry Cove*	*Datolite Mine*	*Passage Island*	*Raspberry Island*	*Bear Island*	*Devils Island*	*AuSable Point*	*Miners Bay*	*Mosquito Harbor*
Kobayasiella	Koba	0.61	0.17	0.49	0.06	0.37	0.21	0.00	2.63	1.32
Krasskella	Kras	0.11	0.12	0.05	0.00	0.00	0.00	0.00	0.00	0.03
Luticola	Luti	0.00	0.00	0.00	0.01	0.00	0.02	0.00	0.00	0.00
Martyana	Mart	0.00	0.00	0.00	0.00	0.05	0.00	0.06	0.00	0.06
Mastogloia	Mast	0.00	0.00	0.00	0.00	0.00	0.00	0.00	1.39	0.00
Meridion	Meri	0.00	0.00	0.00	0.00	0.00	0.00	0.13	0.36	0.16
Microcostatus	Micr	0.02	0.00	0.00	0.00	0.00	0.00	0.00	0.00	0.00
Navicula	Navi	1.11	3.34	0.25	0.45	2.99	1.03	0.61	2.45	1.28
Navicula (small)	Nvsm	0.00	0.00	0.00	0.00	0.00	0.05	0.00	0.03	0.00
Navicula schmassmannii	Nschm	0.00	0.00	0.00	0.11	0.00	0.00	7.28	0.13	0.22
Neidiopsis	Neip	0.00	0.00	0.03	0.00	0.00	0.00	0.00	0.00	0.00
Neidium	Neid	0.02	0.00	0.05	0.00	0.00	0.13	0.10	0.03	0.00
Nitzschia	Nitz	8.92	18.45	9.01	15.40	4.41	2.56	5.82	4.15	4.12
Nitzschia (plankton)	Nzpl	0.08	0.00	0.00	0.00	0.27	0.15	0.03	0.00	0.00
Nupela	Nupe	0.00	0.00	0.00	0.00	0.05	0.00	0.00	0.00	0.00
pennate GV unid	Pund	0.08	0.00	0.00	0.00	0.58	0.49	0.07	0.20	0.28
Pinnularia	Pinn	0.14	0.00	0.00	0.02	0.00	0.25	0.53	0.16	0.00
Planothidium	Plan	0.00	0.00	0.02	0.00	0.00	0.00	0.13	0.10	0.03
Platessa	Plat	0.00	0.00	0.00	0.00	0.00	0.02	0.03	0.06	0.00
Psammothidium	Psam	0.00	0.00	0.33	0.02	0.12	7.74	3.07	0.19	0.13
Pseudostaurosira	Psst	0.00	1.04	0.00	0.00	0.12	0.00	0.00	0.00	0.00
Puncticulata (large)	Punl	0.00	0.05	0.00	0.00	0.02	0.11	0.03	0.00	0.00
Puncticulata (small)	Puns	0.00	0.00	0.00	0.00	0.22	0.02	0.03	0.00	0.00
Reimeria	Reim	0.02	0.00	0.00	0.00	0.05	0.02	0.03	0.03	0.06
Rhopalodia	Rhop	0.17	0.64	0.01	0.00	0.00	0.00	0.00	2.30	0.15
Rossithidium	Ross	0.86	0.06	0.05	1.64	0.20	0.12	0.03	0.22	0.87
Sellaphora	Sell	0.03	0.00	0.00	0.00	0.12	0.04	0.10	0.07	0.03

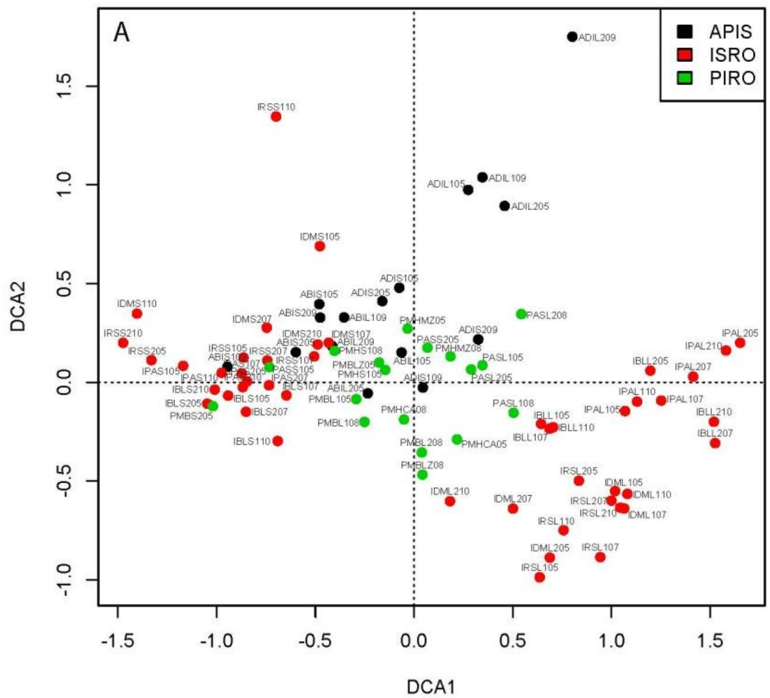
Table 20. Mean percent abundance of diatom genera by pool site at Isle Royale National Park and Apostle Islands and Pictured Rocks national lakeshores, 2010 (continued).

		Mean Percent Abundance								
		ISRO				*APIS*		*PIRO*		
Taxon	**Code**	*Blueberry Cove*	*Datolite Mine*	*Passage Island*	*Raspberry Island*	*Bear Island*	*Devils Island*	*AuSable Point*	*Miners Bay*	*Mosquito Harbor*
Stauroforma	Stfo	3.16	0.00	6.82	0.00	0.00	0.00	0.00	0.00	0.00
Stauroneis	Stau	0.00	0.00	0.03	0.00	0.00	0.00	0.00	0.00	0.00
Staurosira	Stsa	0.02	0.79	0.00	0.14	0.14	0.47	0.38	0.00	0.20
Staurosirella	Stsl	0.00	0.00	0.00	0.04	0.07	0.02	0.19	0.03	0.10
Stenopterobia	Sten	0.00	0.00	0.16	0.00	0.00	0.00	0.00	0.00	0.00
Stephanodiscus (large)	Stel	0.00	0.11	0.00	0.00	0.02	0.05	0.06	0.03	0.00
Stephanodiscus (small)	Stes	0.02	0.00	0.02	0.00	0.00	0.07	0.00	0.03	0.00
Surirella	Suri	0.00	0.00	0.00	0.00	0.00	0.00	0.03	0.00	0.00
Synedra	Synd	2.56	8.36	1.88	5.40	11.74	13.99	9.88	5.61	12.09
Synedra cyclopum	Sync	0.00	0.00	0.00	0.00	0.05	0.00	0.03	0.00	0.00
Tabellaria (long)	Tabl	0.24	0.17	1.37	0.09	0.44	0.62	0.19	0.03	0.12
Tabellaria (small)	Tabs	6.13	0.95	1.95	1.54	0.02	0.09	1.44	0.00	0.73
Ulnaria	Ulna	0.11	0.20	0.06	0.07	0.12	0.10	0.06	0.49	0.43
Urosolenia	Uros	0.02	0.02	0.00	0.00	0.05	0.09	0.06	0.00	0.00

Community Analysis

Diatom communities from all 82 rock pools sampled were ordinated using a detrended correspondence analysis to resolve relationships among samples based on their diatom assemblages. Each park's samples grouped differently in the ordination (Figure 32a). Pools from ISRO perfectly separated into splash zone pools and lichen zone pools along DCA axis 1. Several ISRO splash pool samples from October are notably absent from the main samples group, reflecting some of the instances of seasonality in ISRO's pools (Table 19). PIRO's samples grouped near the origin of the ordination and did not clearly differentiate into pool zones or types; two PIRO splash zone pools (PMBS205 and PASS105) plotted separately within the cluster of ISRO splash pools. APIS pools arrayed primarily along DCA axis 2 with Devils Island lichen pool samples plotting strongly positive on axis 2. APIS pools secondarily arranged along DCA axis 1 with Bear Island samples plotted left of the origin.

Vectors representing the rock pool diatom genera separated into four clusters (Figure 32b). At the center of the ordination are the most common genera that were found in all parks and all pool types including *Achnanthidium* and *Gomphonema*; also found near the origin are several taxa that were limited to the groundwater-fed PIRO pools (*Meridion*, *Diatoma mesodon*). The upper right of the ordination contains species that were most characteristic of lichen pools including the low pH-softwater taxa (*Stauroforma*, *Stenopterobia*, *Frustulia*, *Pinnularia*, *Eunotia*, *Psammothidium*, and chrysophyte cysts). Toward the upper left of the ordination are taxa that characterized many of the splash pools such as *Cyclotella*, *Discostella*, *Eucocconeis*, *Synedra*, and *Denticula*. This group includes additional species that are more characteristic of planktonic diatom communities in Lake Superior such as *Aulacoseira*, *Hannaea*, *Asterionella*, *Urosolenia*, *Puncticulata*, and planktonic *Fragilaria* species that appear able to colonize the splash zone pools. Toward the bottom of the ordination is the last group of diatom genera. Many of these taxa characterize the PIRO lichen pools (*Navicula schmassmannii*, *Mastogloia*, *Epithemia*, *Rhopalodia*, *Fragilariforma*) and cave pools (*Kobayasiella*, *Eolimna*), but are also well-represented in some ISRO lichen pools (*Rhopalodia*, *Nitzschia*, *Rossithidium*, *Encyonema*, *Pseudostaurosira*). Finally, it should be noted that plotted along axis 1 are those taxa that very clearly separate the lichen and splash pool diatom communities. These include *Brachysira* and small *Tabellaria* species, which plotted positive on axis 1, and *Delicata* and *Encyonopsis* species that plotted negative on axis 1.



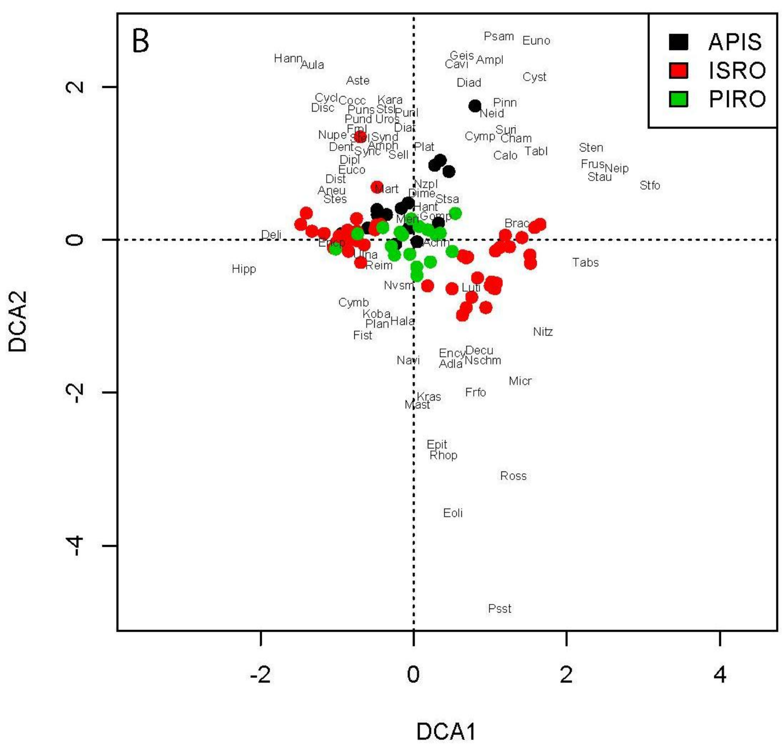


Figure 32. Detrended correspondence analysis of diatom communities in rock pools from Apostle Islands (black symbols), Isle Royale (red symbols), and Pictured Rocks (green symbols), 2010. **A.** Sample scores on DCA axes 1 and 2. See Table Appendix C-1 for sample codes. **B.** Taxon scores on DCA axes 1 and 2. See Table 18 for taxon codes. Note different scaling of axes on Figures A and B.

Table 21. Mean percent abundance of diatom genera (arranged from highest to lowest maximum abundance) during three seasonal sample periods at Isle Royale National Park, 2010.

		Lichen pools			Splash pools		
Taxon	**Code**	***May***	***July***	***October***	***May***	***July***	***October***
Chrysophyte cysts	Cyst	3.94	2.49	2.56	0.05	0.02	0.47
Achnanthidium	Achn	24.14	23.30	20.19	23.91	32.48	8.62
Delicata	Deli	0.11	0.00	0.05	32.17	27.56	31.95
Nitzschia	Nitz	23.35	28.40	23.44	0.89	1.27	0.32
Encyonopsis	Encp	2.72	3.01	5.85	20.19	17.18	21.08
Brachysira	Brac	13.78	15.18	16.82	1.82	1.52	3.58
Cyclotella	Cycl	0.22	0.00	0.00	0.64	0.38	10.78
Synedra	Synd	1.48	1.05	0.89	10.10	8.63	5.14
Encyonema	Ency	9.40	6.07	6.15	1.26	1.78	3.66
Tabellaria (small)	Tabs	3.92	5.16	6.78	0.00	0.00	0.00
Cymbella	Cymb	2.05	2.14	2.41	5.66	4.47	6.51
Stauroforma	Stfo	5.21	4.87	4.89	0.00	0.00	0.00
Navicula	Navi	1.75	1.32	2.87	0.07	0.73	0.98
Aulacoseira	Aula	0.00	0.00	0.00	0.00	0.02	2.67
Gomphonema	Gomp	1.99	1.10	1.01	0.56	0.62	1.21
Denticula	Dent	0.00	0.00	0.00	1.32	1.62	0.22
Rossithidium	Ross	1.60	1.44	0.82	0.00	0.07	0.00
Tabellaria (long)	Tabl	0.54	1.01	0.94	0.07	0.05	0.20
Eunotia	Euno	0.38	0.36	0.97	0.00	0.05	0.00
Pseudostaurosira	Psst	0.97	0.37	0.22	0.00	0.00	0.00
Rhopalodia	Rhop	0.16	0.22	0.85	0.00	0.00	0.00
Kobayasiella	Koba	0.30	0.36	0.11	0.27	0.26	0.68
Staurosira	Stsa	0.20	0.68	0.34	0.00	0.00	0.21
Frustulia	Frus	0.11	0.05	0.67	0.00	0.00	0.00
Epithemia	Epit	0.59	0.57	0.34	0.00	0.00	0.00
Fragilaria (plankton)	Frpl	0.15	0.00	0.00	0.02	0.41	0.21
Discostella	Disc	0.05	0.00	0.00	0.00	0.10	0.40
Eucocconeis	Euco	0.02	0.05	0.00	0.34	0.38	0.15
Psammothidium	Psam	0.37	0.10	0.05	0.00	0.00	0.00
Ulnaria	Ulna	0.00	0.00	0.12	0.07	0.27	0.19
Hannaea	Hann	0.00	0.00	0.00	0.00	0.02	0.21
Stephanodiscus (large)	Stel	0.00	0.00	0.00	0.17	0.00	0.00
Cocconeis	Cocc	0.00	0.00	0.00	0.02	0.00	0.15
Krasskella	Kras	0.08	0.13	0.07	0.15	0.00	0.00
Stenopterobia	Sten	0.05	0.04	0.14	0.00	0.00	0.00
Asterionella	Aste	0.00	0.00	0.00	0.00	0.00	0.14
Pinnularia	Pinn	0.09	0.05	0.10	0.00	0.00	0.00
Eolimna	Eoli	0.00	0.10	0.00	0.00	0.00	0.02
Navicula schmassmannii	Nschm	0.02	0.10	0.05	0.00	0.00	0.00
Diatoma	Diat	0.10	0.00	0.00	0.00	0.00	0.05
Nitzschia (plankton)	Nzpl	0.00	0.09	0.03	0.00	0.00	0.00
Caloneis	Calo	0.04	0.00	0.09	0.00	0.02	0.00

Table 21. Mean percent abundance of diatom genera (arranged from highest to lowest maximum abundance) during three seasonal sample periods at Isle Royale National Park, 2010 (continued).

		Lichen pools			**Splash pools**		
Taxon	**Code**	***May***	***July***	***October***	***May***	***July***	***October***
Chamaepinnularia	Cham	0.05	0.05	0.09	0.00	0.00	0.00
Puncticulata (large)	Punl	0.00	0.00	0.00	0.00	0.00	0.07
Amphora	Amph	0.02	0.00	0.00	0.07	0.00	0.00
pennate GV unid	Pund	0.00	0.00	0.00	0.07	0.05	0.00
Staurosirella	Stsl	0.00	0.00	0.00	0.00	0.00	0.06
Sellaphora	Sell	0.00	0.00	0.05	0.00	0.00	0.00
Neidiopsis	Neip	0.00	0.04	0.00	0.00	0.00	0.00
Neidium	Neid	0.03	0.04	0.03	0.00	0.00	0.00
Stauroneis	Stau	0.00	0.04	0.00	0.00	0.00	0.00
Urosolenia	Uros	0.00	0.00	0.00	0.02	0.00	0.02
Reimeria	Reim	0.00	0.00	0.00	0.02	0.00	0.00
Planothidium	Plan	0.00	0.00	0.00	0.02	0.00	0.00
Stephanodiscus (small)	Stes	0.00	0.00	0.00	0.00	0.02	0.02
Microcostatus	Micr	0.02	0.00	0.00	0.00	0.00	0.00
Karayevia	Kara	0.00	0.00	0.00	0.00	0.02	0.00
Diadesmis	Diad	0.00	0.02	0.00	0.00	0.00	0.00
Luticola	Luti	0.02	0.00	0.00	0.00	0.00	0.00

Water Quality

Physical Characteristics of Pools

Establishing study sites based on two lichen, two splash, and ephemeral pool presence was easy at ISRO, limited to very few sites at APIS, and not possible at PIRO. Many potential sites with pools at APIS were dry due to a very dry winter and spring in 2010. At PIRO, only likely sites for rock pools were selected (Au Sable Point, Miners Beach, Mosquito Harbor); however, these sites also contained pools fed by groundwater seeps. Particularly interesting are the medicolous zone (MZ) pool and cave (CA) pool at PIRO's Mosquito Harbor site.

Measures of pool dimensions (length and width), pool depth, and location of pool relative to Lake Superior and treeline were taken for each established permanent pool. Box plots of data presented by park unit and separated by pool zone (lichen, splash, other) highlight differences among parks and among pool types (Figure 33).

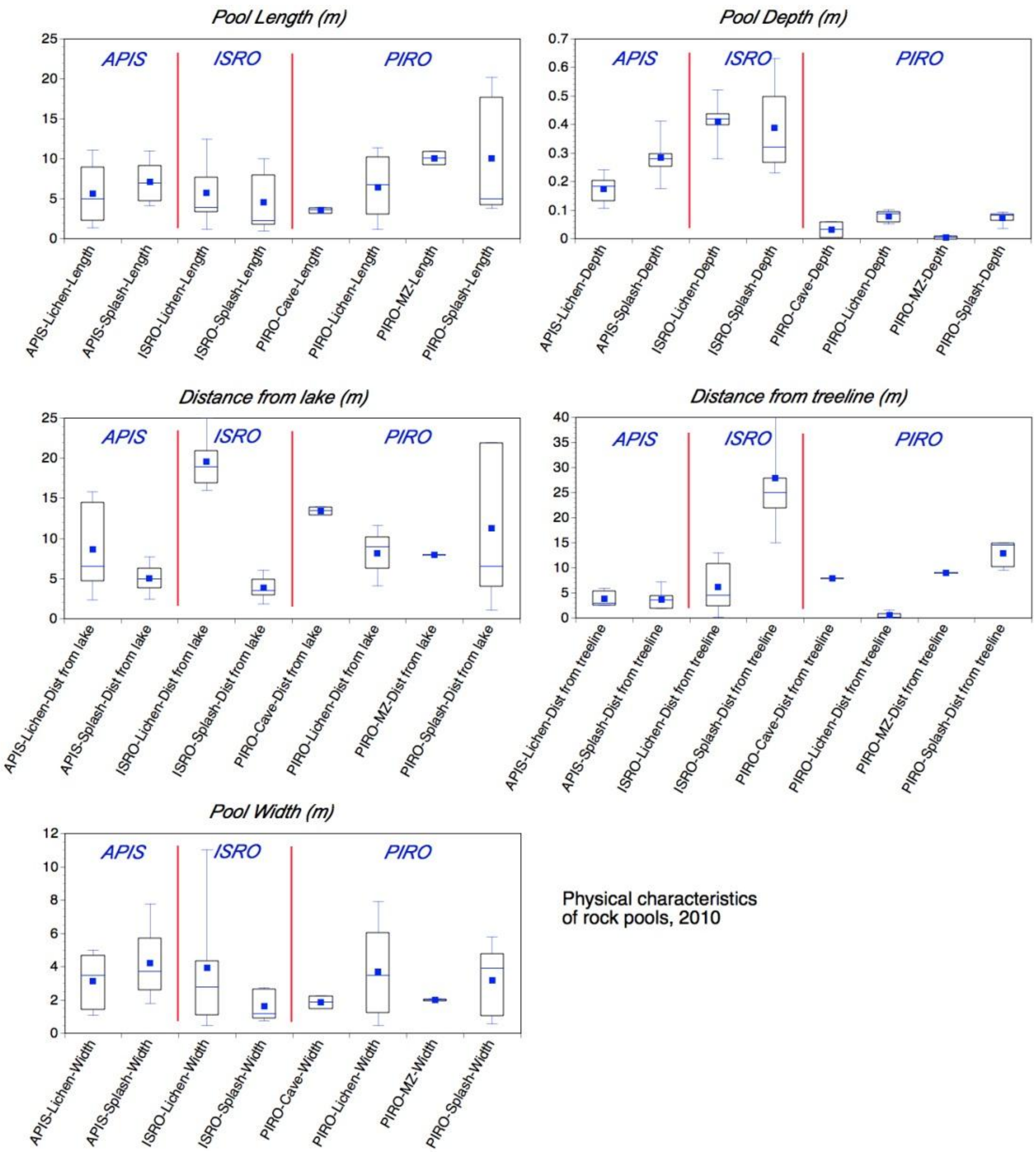


Figure 33. Physical characteristics of rock pool zones or types (lichen, splash, cave, medicolous zone) at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. Plots include pool length (m), width (m), and depth (m), and position on shoreline relative to Lake Superior (m) and treeline (m). See Figure 28 for explanation of boxplots.

<u>ISRO</u>

The length of Isle Royale rock pools ranged from <1 m to 10 m. There was no difference in length between splash and lichen pools. Pools at ISRO were the deepest among the parks, averaging approximately 40 cm deep. The shoreline at ISRO provides what we consider “classic” rock pool habitat, with a sloped bedrock shoreline extending upward and away from the lake and a distinct differentiation between splash and lichen pool zones. As such, lichen pools are much closer to treeline and much farther from the shoreline than splash pools (Figure 33).

APIS

Pools on Bear and Devils islands at APIS were similar in length to ISRO pools but were shallower (Figure 33). The shoreline geology at APIS is primarily sandstone and in most sites pools were located on large flat expanses of bedrock located 1-3 m above the lake level. The delineation of rock and splash pool zones was much less clear than at ISRO due to this shoreline configuration. Lichen pools were farther from the lake, but the distance to treeline varied little between the APIS lichen and splash pools we studied (Figure 33).

PIRO

Pools were highly variable in length and width, ranging from the rather small cave pool at Mosquito Harbor to very large splash pools (Figure 33). Pools at PIRO were the shallowest among the parks; notably the groundwater-fed cave and medicolous zone pools were less than 10 mm deep. Whereas splash pools were located more distant from treeline at the PIRO sites, distance from the lake was highly variable among pools.

Field Measures of Pools

Measures of dissolved oxygen, pH, specific conductivity, and water temperature were taken during each water quality sampling event on permanent pools and for Lake Superior (Figure 34).

Dissolved Oxygen

All pools were well-saturated with oxygen during the 2010 sampling. Dissolved oxygen levels were >80% saturation and rarely less than 8 mg/L (Figure 34). The lowest average oxygen levels were in the medicolous pool at PIRO. Sampling of pool water generally occurred at least several hours after sunrise in full daylight. No diurnal sampling efforts were made to determine potential for oxygen stress at night.

pH

The pH of most rock pools and Lake Superior waters was between 7.5 and 8.5 (Figure 34). The pH of lichen pools at APIS was lowest and rather variable, with some pools measuring below pH 6. Splash pools at ISRO were consistently between pH 8.0 and 9.0. The pH of the cave pool at PIRO was close to 8.5, likely a reflection of its groundwater source.

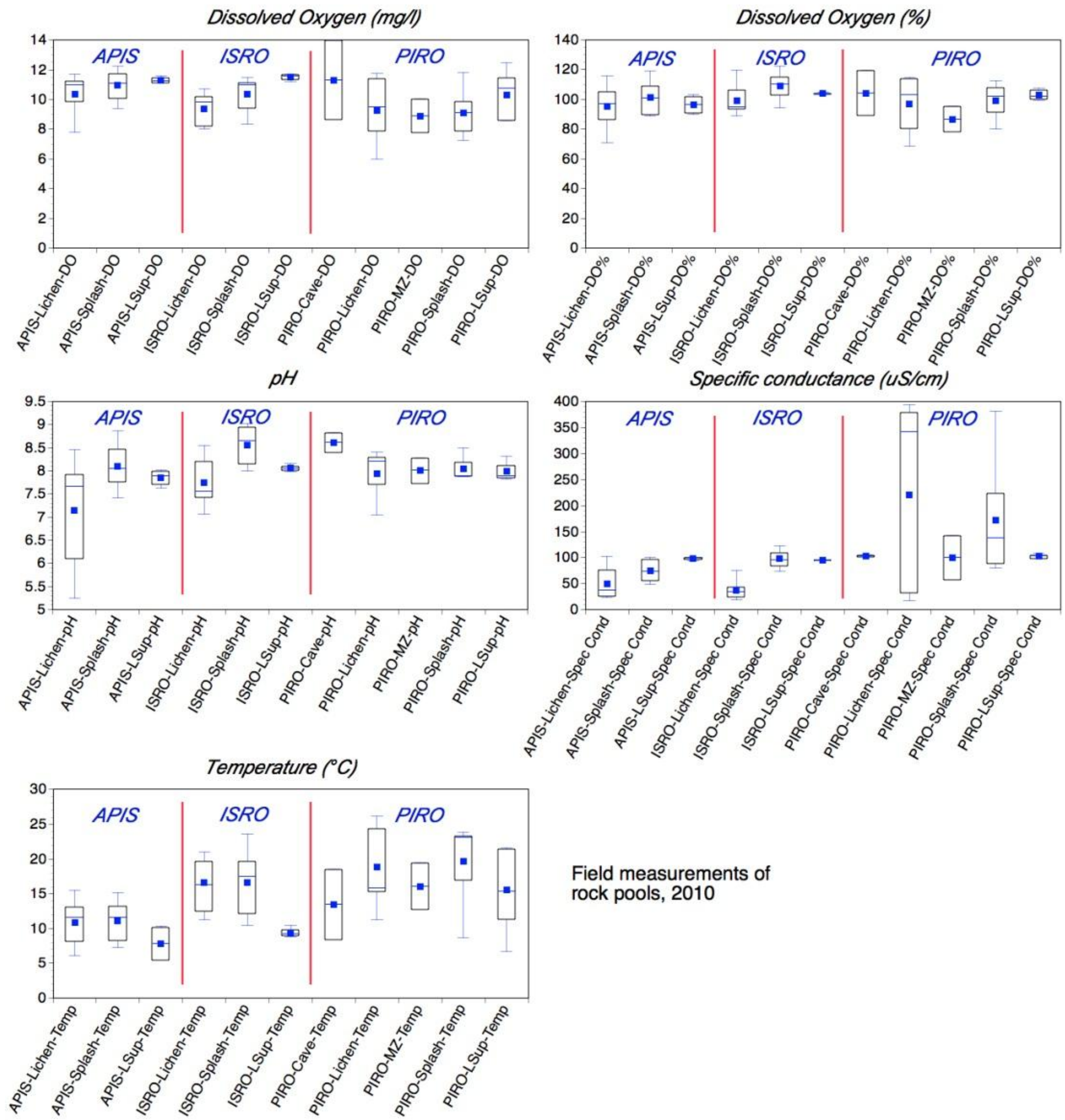


Figure 34. Dissolved oxygen (mg/L and % saturation), pH, specific conductivity (µS/cm), and water temperature (°C) for permanent pools (lichen, splash, other) and Lake Superior (LSup) at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. Data are presented by park unit and separated by pool zone (lichen, splash, other). See Figure 28 for explanation of boxplots.

Specific Conductivity

The specific conductivity of Lake Superior at all three park units was 100 µS/cm, despite the time of year (Figure 34). Specific conductivity of splash pools at ISRO was nearly identical to that of Lake Superior, whereas splash pools at APIS had lower conductivity than that of Lake Superior. At both ISRO and APIS, lichen pools had markedly lower conductivity in comparison to splash pools and

Lake Superior waters. Conductivity values were highly variable at PIRO; the conductivity of lichen pool waters ranged from <50 to over 350 µS/cm at Miners Beach, whereas splash pool waters were >100 µS/cm, and the cave and mediculous pools were approximately 100 µS/cm.

<u>Temperature</u>

During the regular sampling of pools in 2010, water temperatures typically reflected seasonal air temperatures (Figure 34). Pool temperatures were usually warmer than Lake Superior temperatures. Rock pools at APIS were cooler than the other parks; APIS was the first park sampled (5–6 May 2010), which produced cooler temperature data. Pools at PIRO were the most variable among the parks. The coolest temperatures were recorded at the cave pool, which reflected its groundwater source. The narrow range of Lake Superior water temperature at ISRO is a result of reporting only the October 2010 data.

Temperature data taken at the time of regular sampling did little to capture the thermal behavior of pools on shorter and more ecologically relevant time scales. To determine seasonal and diurnal changes in rock pool temperature, HOBO® temperature loggers (Onset Computer Corporation) were deployed at select ISRO rock pools in 2010 and 2012 near each pool's point of maximum depth. Loggers were set to record temperature each hour. In 2010 temperature loggers were deployed at a single lichen and a single splash pool at each of four sites: Blueberry Cove, Passage Island, Datolite Mine, and Raspberry Island. In 2012, loggers were placed in a single lichen and single splash pool at Blueberry Cove, Passage Island, and Raspberry Island. We report here on the thermal behavior of a splash and lichen pool at one site (Blueberry Cove) from 2012 (Figure 35). Data from the other pools and other sample years are recorded in the project database.

Between 6 May and 26 August 2012, temperatures ranged from 7.8 °C to 32.8 °C in the lichen pool and from 5.0 °C to 29.8 °C in the splash pool (Figure 35). The overall pattern of temperature fluctuation was similar between the pools, with coolest seasonal temperature in early May and warmest temperatures in July. Splash pool temperatures dropped to near 5 °C several times between early May and mid-June. Some of these low temperatures appear correlated to low air temperatures; however, others may be indicators of wave scouring as lichen pools do not record simultaneous temperature minima. A strong diurnal difference exists between the pool zones (Figure 35). Lichen pools had greater diurnal temperature ranges compared to splash pools. In July 2012, lichen pools regularly warmed and cooled daily across a range of 10 °C to 15 °C. Splash pools had a much narrower range of daily temperature extremes, typically 5 °C to 8 °C.

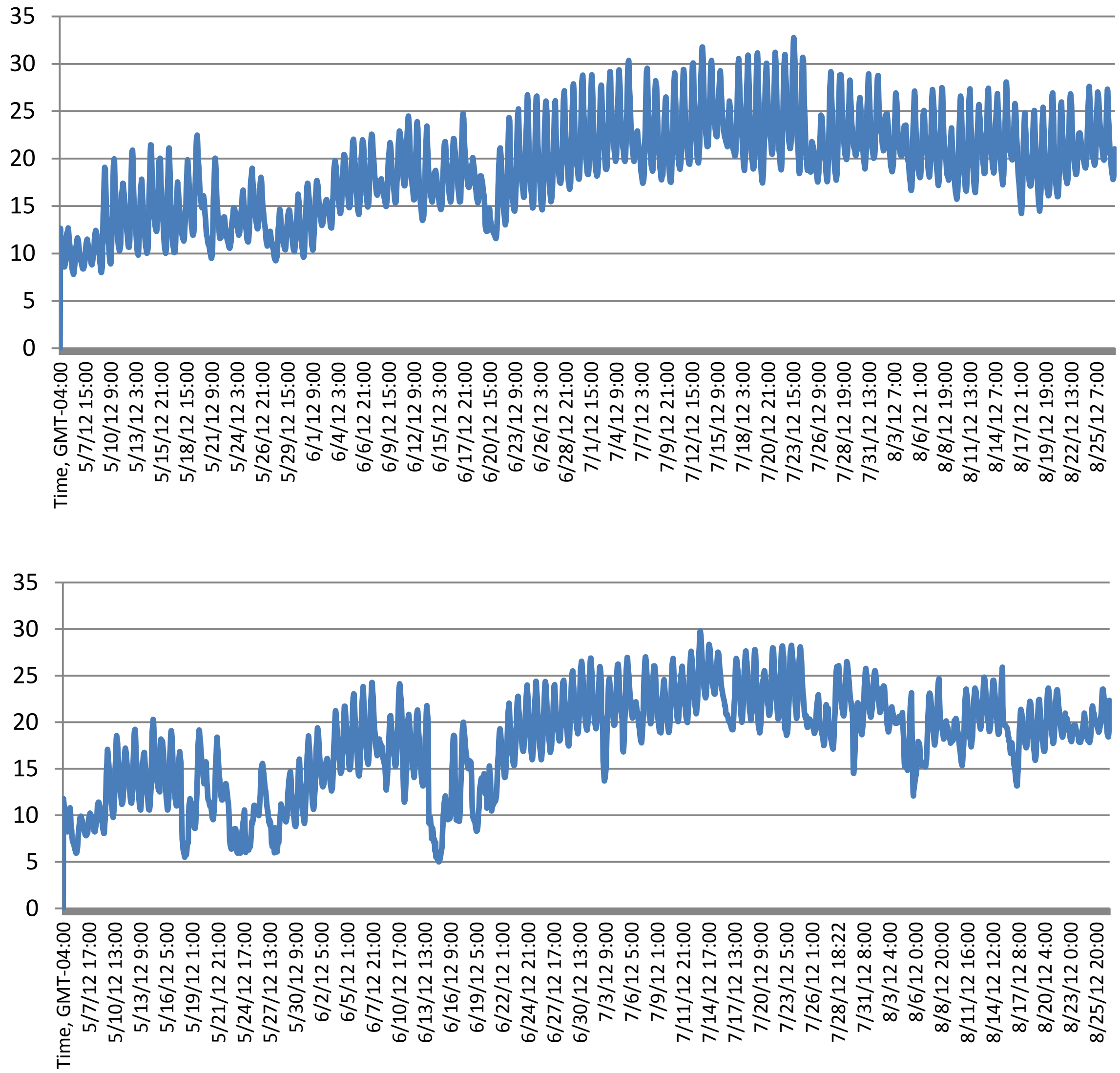


Figure 35. Hourly temperature (°C) readings from one permanent lichen pool (top panel) and one permanent splash pool (bottom panel) at the Blueberry Cove site, Isle Royale National Park, 6 May–26 August 2012.

Chemical Characteristics of Pools

Nutrients and Chlorophyll

Boxplots of nutrient and chlorophyll values from Lake Superior and lichen and splash pools show clear differences between lichen and splash pools and strong connections in water quality between splash pools and Lake Superior waters (Figures 36–38). Based on nutrient and chlorophyll values, lichen pools would mostly be considered mesotrophic systems, with total phosphorus levels usually >10 µg/L, SRP levels >2 µg/L, and chlorophyll levels of >1 µg/L. A few pools were highly productive systems, with chlorophyll values of over 100 µg/L recorded at some PIRO lichen pools in August 2010 (Figure 38). In contrast, splash pools were less productive and generally oligotrophic systems, with chlorophyll values >1 µg/L and TP of <10 µg/L. Splash pools were notably similar to Lake Superior waters with regard to TP, TN, NH_4-N, SRP, and chlorophyll.

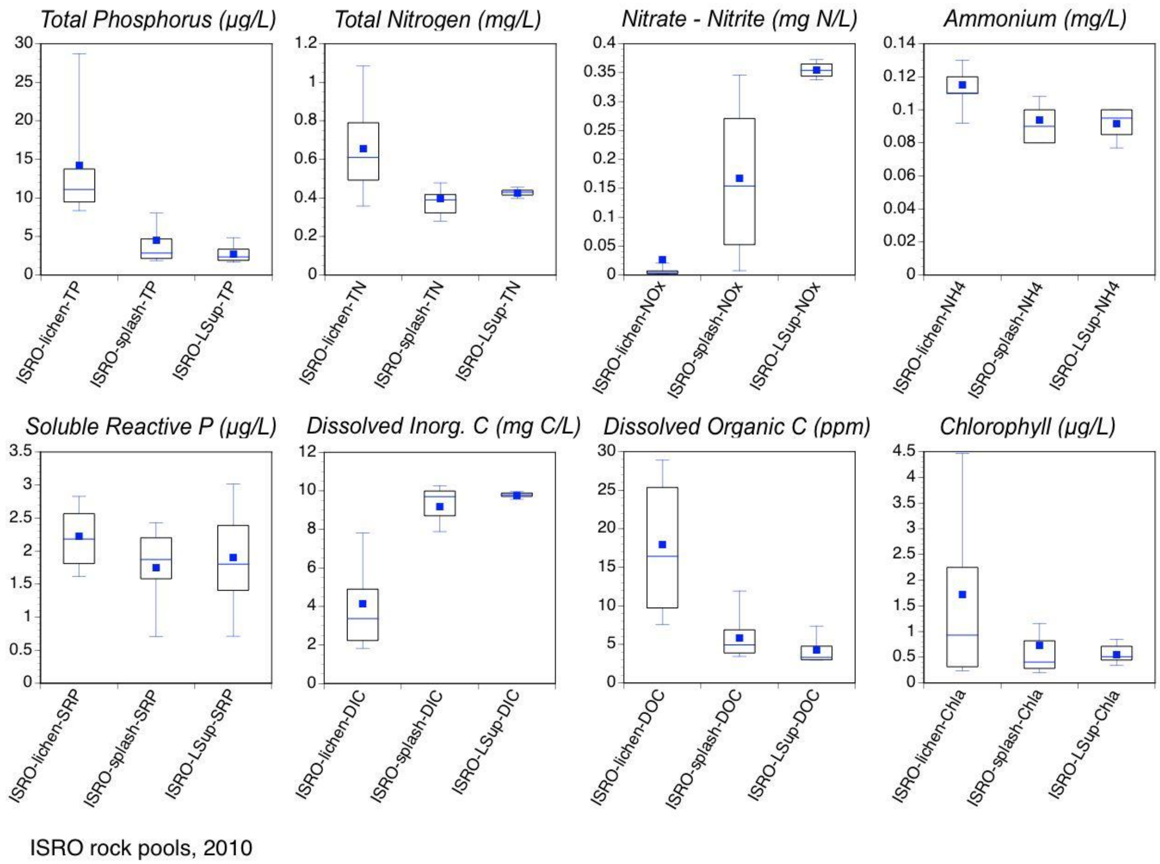


Figure 36. Boxplots of select water quality variables measured for lichen pools, splash pools, and Lake Superior (LSup) at Isle Royale National Park, 2010. See Figure 28 for explanation of boxplots.

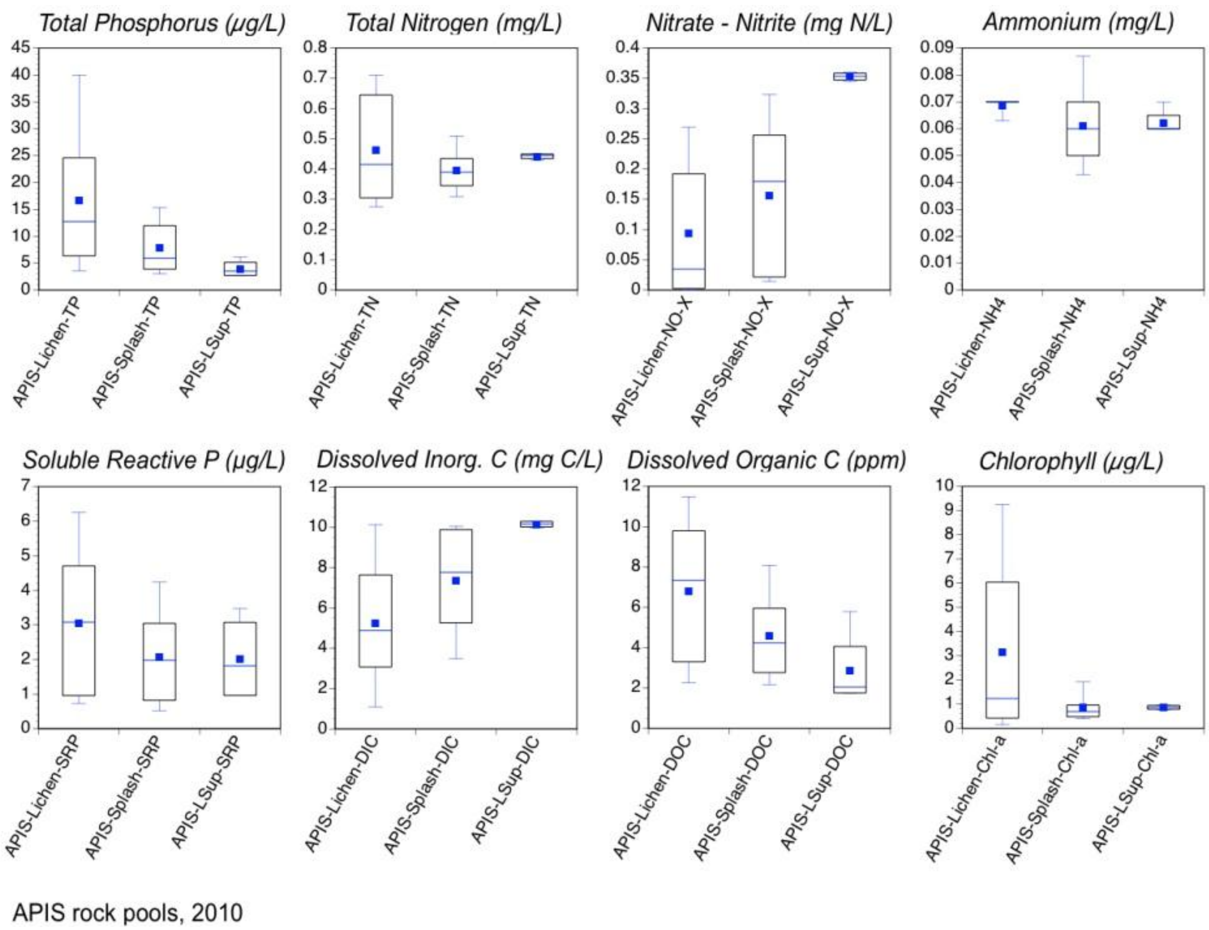


Figure 37. Boxplots of select water quality variables measured for lichen pools, splash pools, and Lake Superior (LSup) at Apostle Islands National Lakeshore, 2010. See Figure 28 for explanation of boxplots.

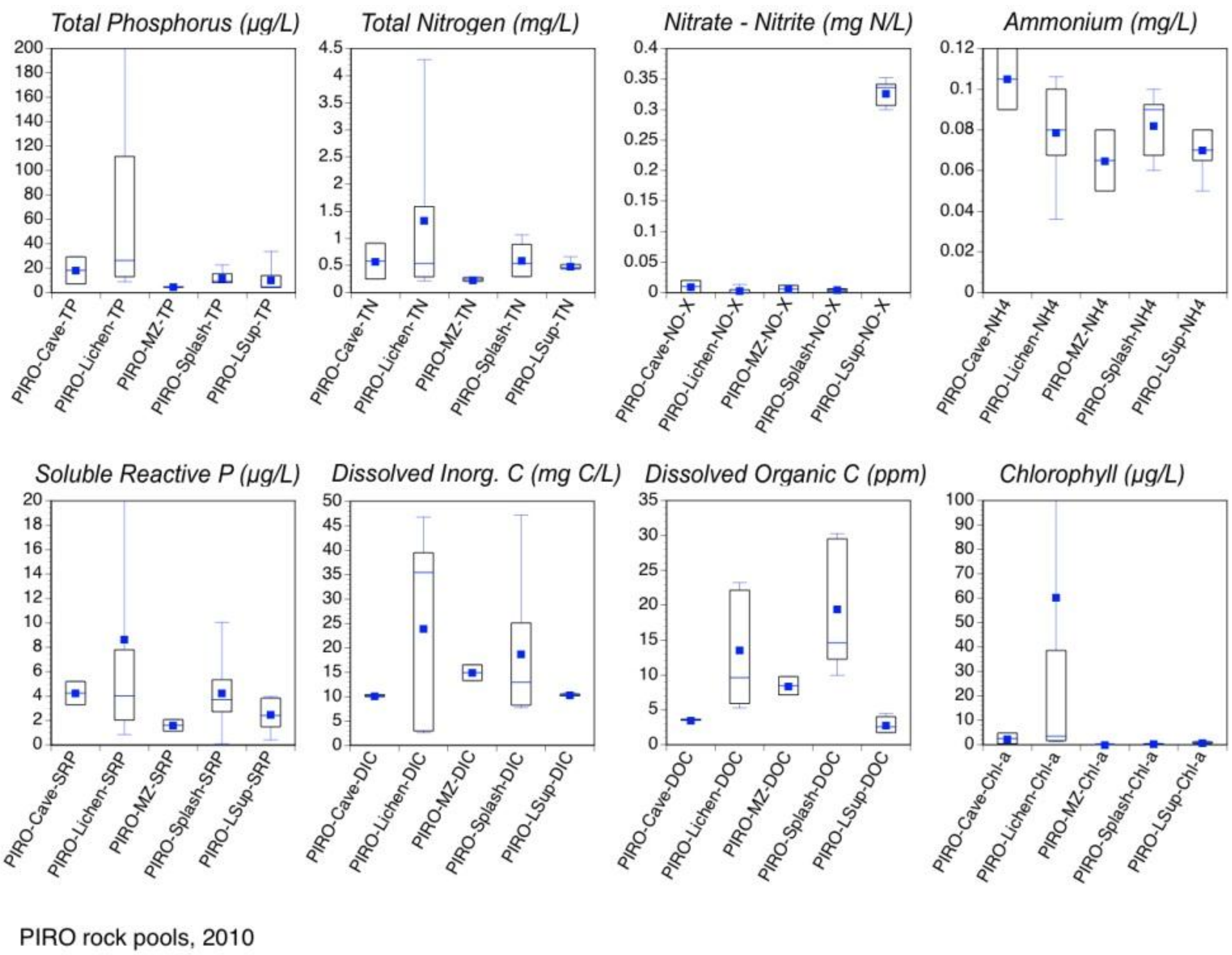


Figure 38. Boxplots of select water quality variables measured for lichen pools, splash pools, cave pool, mediculous zone (MZ), and Lake Superior (LSup) at Pictured Rocks National Lakeshore, 2010. See Figure 28 for explanation of boxplots.

Nutrient ratios are one way to measure nutrient limitation in the rock pools. The TN:TP and the dissolved inorganic nitrogen to total phosphorus (DIN:TP) ratios were calculated for each pool and for Lake Superior water (Figure 39). TN:TP ratios greater than 30 are indicative of phosphorous-limitation; ratios less than 30 indicate systems not limited by phosphorous (Bergström 2010). DIN:TP ratios of >3.4 indicate phosphorous-limitation, ratios <1.5 represent nitrogen-limitation, and ratios between 1.5 and 3.4 suggest N-P co-limitation (Bergström 2010). Lake Superior waters, all splash pools, and the groundwater-fed cave and mediculous pools (PIRO) were decidedly P-limited regardless of which ratio was used. Among the lichen pools, all of ISRO's and most of the APIS's pools showed P-limitation, whereas lichen pools at PIRO were N-P co-limited or N-limited systems (Figure 39).

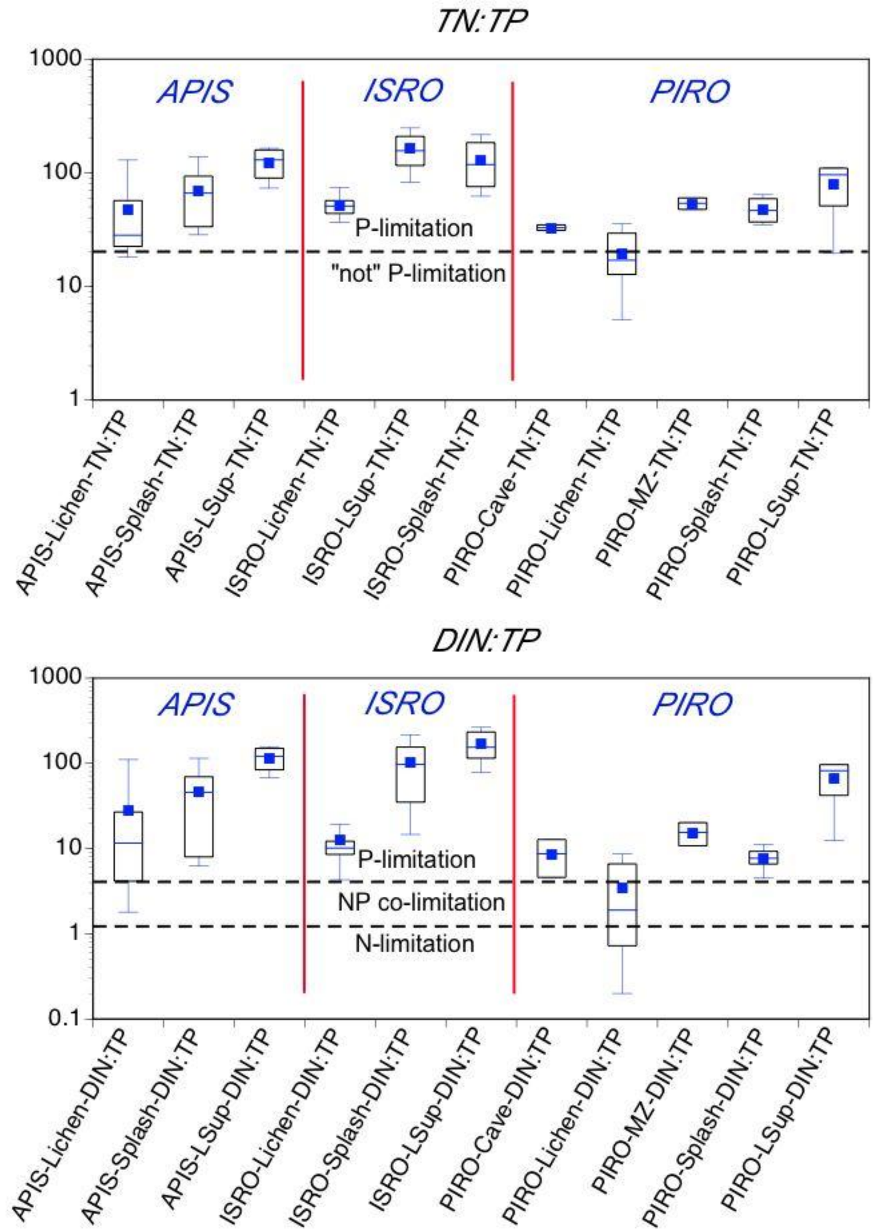


Figure 39. Nutrient ratios as indices of nutrient limitation in rock pools (lichen, splash, other) at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore. TN:TP ratios greater than 30 are indicative of P-limitation; ratios less than 30 indicate systems not limited by P (Bergström 2010). DIN:TP ratios >3.4 indicate P-limitation, ratios <1.5 represent N-limitation, and ratios between 1.5 and 3.4 suggest N-P co-limitation (Bergström 2010).

<u>DIC/DOC</u>

Dissolved inorganic carbon (DIC) represents the combined amount of CO_2, carbonic acid, HCO_3- and CO_3^{2-} that is dissolved in water. DIC is closely related to pH of the water (greater DIC at higher pH), microbial mineralization, and water sources including lake and groundwater inputs. Lake Superior waters at all parks showed 10 mg/L DIC, and splash pools at ISRO were identical (see Figures 32–34). Splash pools at APIS had lower DIC values, and splash pools at PIRO had higher and variable DIC. Lichen pools at both ISRO and APIS had very low DIC levels of 2–8 mg/L. In constrast, lichen pools at PIRO showed a large range of DIC (3 to >40 mg/L), with the lichen pools at Miners Bay showing the highest values. The cave and medicolous pools at PIRO had DIC values of 10–15 mg/L (see Figure 38).

Dissolved organic carbon (DOC) represents the amount of dissolved organic matter in water and is most easily seen as tea or coffee-stained water. DOC is typically a product of the breakdown of plant matter or the leaching or direct contact of water with organic soils. The clear waters of Lake Superior had very low levels of DOC, generally less than 5 ppm (see Figures 36–38). Splash pools at ISRO were similar to Lake Superior waters, while splash pools at APIS were slightly higher in DOC, perhaps because of their closer proximity to treeline. Splash pools at PIRO had much higher levels of DOC compared to other parks' splash pools (see Figures 36–38). Lichen pools had the highest DOC among pool types at ISRO and APIS, with ISRO lichen pools having similar levels to PIRO lichen pools, but two-fold greater levels than APIS. Cave pools at PIRO had DOC values only slightly greater than Lake Superior waters, whereas the medicolous pool had slightly higher DOC of approximately 7-10 ppm (see Figure 38).

Relationships Among Field and Water Quality Measures

Principal components analysis (PCA) was used to explore relationships among field and water quality variables. Water quality variables that were measured in all pool and Lake Superior water samples (including TP:TN and DIN:TP) were run as a pooled analysis of all parks, but with ordinations separated by park unit for clarity (Figures 40–42). Water quality variables that were strongly and positively loaded on PCA axis 1 included TP, SRP, TN, DOC and chlorophyll-*a*; NO_3-NO_2, TN:TP, and DIN:TP were highly correlated and negatively loaded on PCA Axis 1. Specific conductivity and DIC were most strongly loaded on PCA axis 2 (Figures 40–42). At ISRO, pools were separated along axis 1 with lichen pools positively loaded and splash and Lake Superior waters negatively loaded on axis 1 (Figure 40), differences that reflect the higher productivity, higher nutrients (except nitrates), and higher DOC of ISRO's lichen pools. APIS pools are similarly disposed on the PCA ordination with lichen pools trending positive on axis 1, and Lake Superior and splash pools positioned more negatively along axis 1. At APIS, however, the pools show some overlap in distribution on the ordinations, likely a consequence of the poor distinction between lichen and splash zones at APIS (Figure 41). Lake Superior and pool samples from PIRO are most strongly distributed along PCA axis 2. Lichen pools from PIRO separated into two groups—one with the higher nutrient and chlorophyll pools of AuSable Point, and the other including the higher conductivity lichen pools at Miners Bay; the latter loaded negatively on axis 2. Lake Superior waters at PIRO are tightly grouped near the PCA origin, whereas splash pools are distributed along axis 2 and interspersed within the lichen pools, reflecting the highly variable water chemistry encountered at PIRO. The cave and medicolous pools of PIRO were centrally grouped in the ordination and slightly negative on axis 2 (Figure 42).

Anions, Cations, Trace Metals

In addition to the physical, field, and water quality parameters presented above, the full suite of anions, cations, and trace metals were analyzed for each water sample. These data are presented in tabular form in Appendix C to provide baseline measures for Lake Superior rock pools.

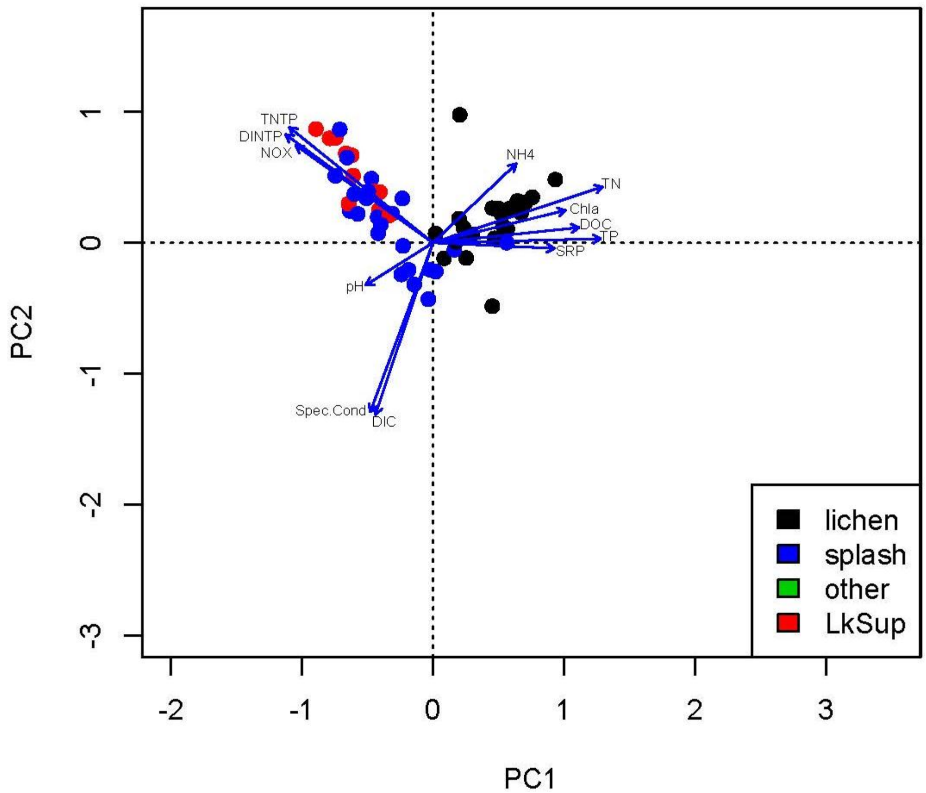


Figure 40. Principal components analysis of water samples from lichen pools (black), splash pools (blue), other pool types (green), and Lake Superior (red) from Isle Royale National Park, 2010. Blue arrows represent environmental vectors.

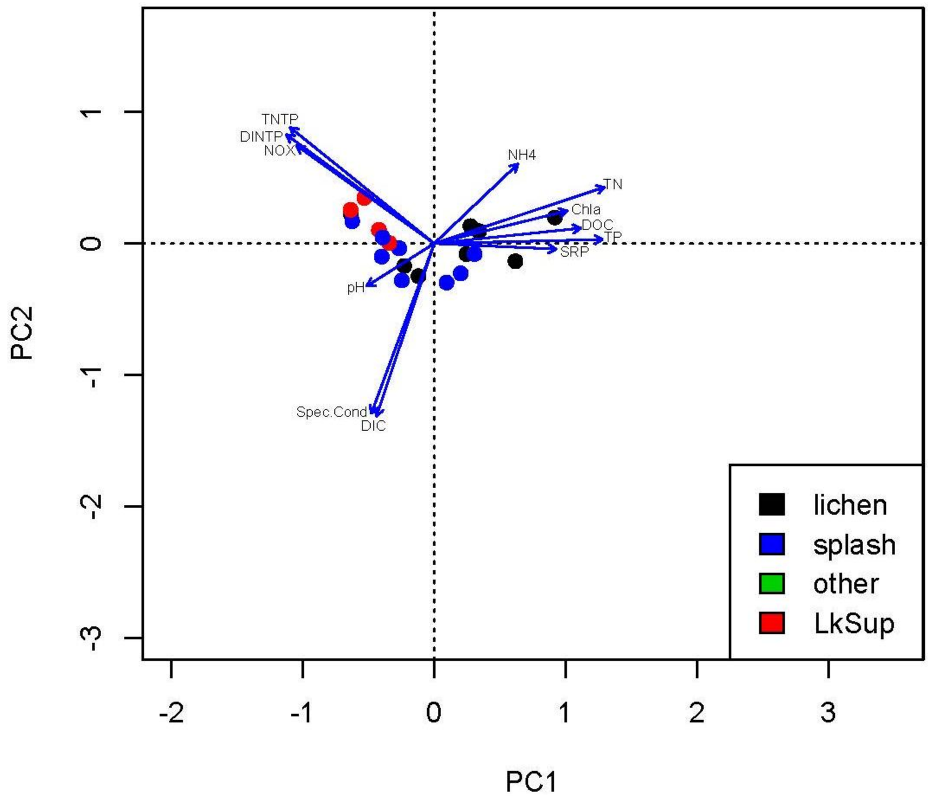


Figure 41. Principal components analysis of water samples from lichen pools (black), splash pools (blue), other pool types (green), and Lake Superior (red) from Apostle Islands National Lakeshore, 2010. Blue arrows represent environmental vectors.

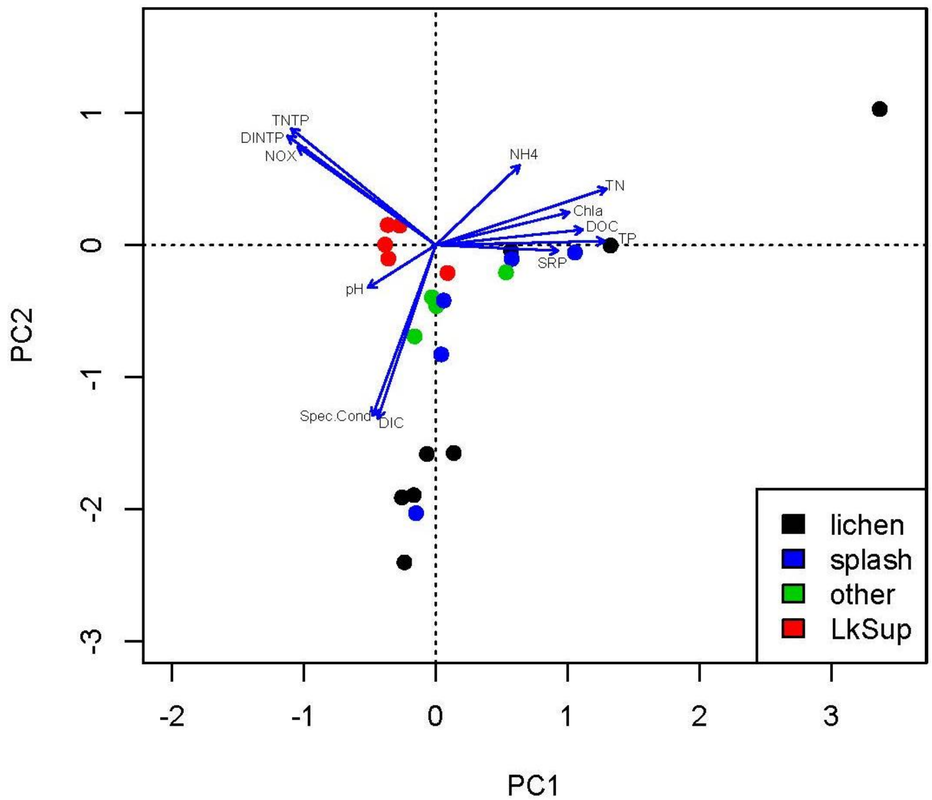


Figure 42. Principal components analysis of water samples from lichen pools (black), splash pools (blue), other pool types (green), and Lake Superior (red) from Pictured Rocks National Lakeshore, 2010. Blue arrows represent environmental vectors.

Coastal Habitat Mapping

As described in Appendix B, mapping at Isle Royale in 2011 and 2012 established a unique geodatabase which can be used by managers in responding to coastal spills, or as a baseline dataset for future research. Forty-eight kilometers (30 miles) of shore was comprehensively mapped, yielding 71,931 pools. Almost 63% (45,164) of the pools occurred on Passage Island (Figure 43). From Blake's Point to West Caribou Island, an area comprising mostly barrier islands (see Figure 2), 15,870 pools were mapped, and 10,897 pools were between West Caribou Island and the Datolite Mine area on the south shore of Isle Royale. In addition to describing the pools, mapping provided an opportunity to document amphibian occupancy of the rock pools (Table 22). Isle Royale was expected to have the most abundant habitat and was therefore the only park mapped.

Pools along the south shore of Isle Royale are typically in the splash zone, and most pools are ephemeral (see Figure 43). The forest transition zone has relatively few pools, and seep recharge is much less common, yet almost 5,000 pools fit this description. Numbers in Figure 43C do not add to 71,931 because some pools were removed when outliers lacked credibility (e.g., a pool depth reading of 40 m— a mistake of data entry, or 0 m—a forgotten entry).

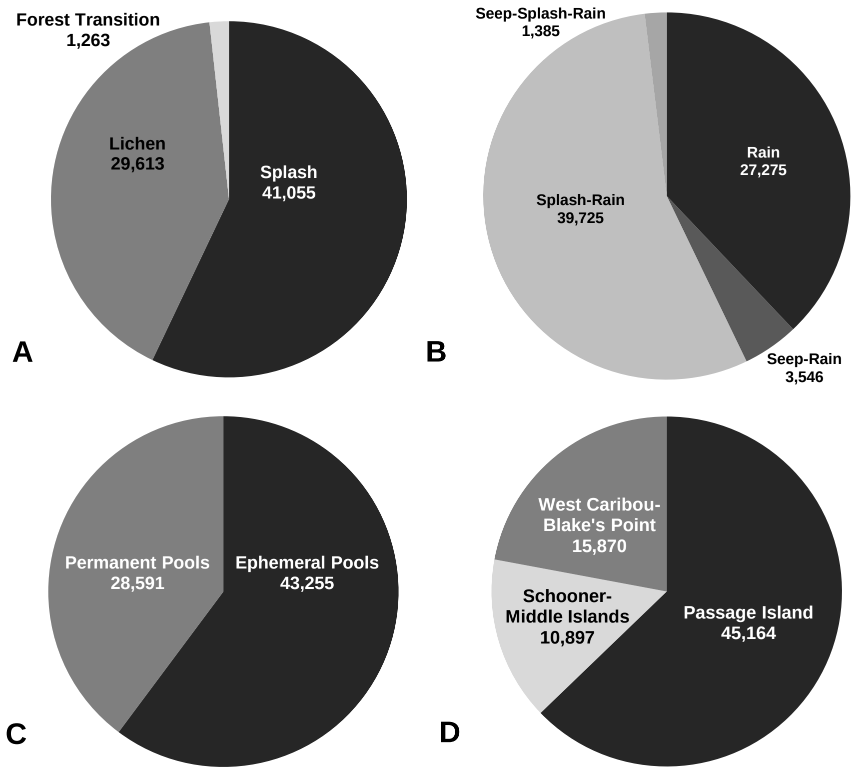


Figure 43. Characterizations and numbers of pool types, including all pools mapped between the Datolite Mine area (including Schooner Island) and Passage Island, Isle Royale National Park, 2011–2012. Pools were characterized by zone occupancy (A), recharge sources (B), permanence (C), and density along selected areas (D).

Table 22. Amphibian occupancy of south-shore coastal rock pools (from Datolite Mine area to Passage Island) at Isle Royale National Park, 2011–2012.

Species	Pools with larvae or eggs	Pools with adults	Total	Proportion of pools occupied
Chorus frog (*Pseudacris triseriata*)	2,098	16	2,114	3%
Blue-spotted salamander (*Ambystoma laterale*)	945	0	945	1%
Spring peeper (*Pseudacris crucifer*)	74	1	75	<1%
Green frog (*Rana clamitans*)	2	22	24	<1%
Wood frog (*Rana sylvatica*)	1	12	13	<1%
Eastern newt (*Notophthalmus viridescens)*	6	7	13	<1%
American toad (*Bufo americanus*)	2	6	8	<1%

Discussion

Chironomidae

Fifty-nine genera of Chironomidae, the most numerous and diverse macroinvertebrate group encountered in rock pools, were collected from all three parks. Several of the genera contain species that are known to occur in rock pools elsewhere, or in similar habitats such as splash zones, madicolous habitats, small springs, seeps or soils saturated by groundwaters in close proximity to the ground surface. In addition, larvae of species in several genera are known to have specialized adaptations for resisting desiccation as ephemeral aquatic habitats become dry. Consequently, these taxa may be considered part of a core of species forming the chironomid community in rock pools along the shores of Lake Superior. Overviews and autecologies follow for selected abundant and ecologically noteworthy taxa. Our discussion will focus on ISRO chironomids followed by highlights from APIS and PIRO.

Selected Chironomidae Genera

***Orthocladius* (*Eudactylocladius*):** The most recent taxonomic revision of the genus was by Sæther (2005), who stated that "the subgenus *Eudactylocladius* Thienemann of the genus *Orthocladius* v. d. Wulp often dominates the fauna of thin water films in temperate regions. In arctic regions the larvae occur in inundated or damp soil and lake margins."

***Psectrocladius* (*Psectrocladius*):** This taxon was second-most abundant in rock pools. Sæther and Langton (2011) published a partial revision of this genus and stated that "*Psectrocladius* larvae occur in shallow, standing or slow-flowing water bodies (including shallow ditches) and some species appear to be especially common in habitats where no or few fish predators are present". Larvae can often be concentrated in growths of filamentous algae in small pools or, less commonly, can graze on periphyton films in very shallow water (Ferrington, personal observations).

***Corynoneura*:** Larvae of this genus are among the smallest and fastest growing species of Chironomidae. They are widespread and recorded from a broad array of aquatic habitats, including three species known from springs, splash zones and small pools (Ferrington et al. 1995).

***Paratanytarsus*:** This genus was most common at ISRO, but less abundant at the other two parks. Larvae are known to be able to resist dehydration by producing cocoons as small pools and temporary habitats become dry (Grodhaus 1980).

***Orthocladius* (*Orthocladius*):** This subgenus of *Orthocladius* is very species-rich, with at least 31 species known from North America, and larvae are recorded from a wide variety of aquatic habitats. Chou et al. (1999) recorded three species from an intermittent stream in Kansas and considered them to be potentially desiccation-resistant in the egg or larval stages. Williams and Hynes (1976) considered one species of the genus to be capable of over-summering as resistant eggs.

***Limnophyes*:** The larvae of most *Limnophyes* species live in moss, wet earth, or wet leaves of hygropetric zones, springs, streams, seeps or road cuttings. A few species, however, appear to be truly aquatic, living in the littoral zone of lakes or in small streams (Sæther 1990). More recently, Przhiboro and Sæther (2007) record three species in shallow wave-swept littoral zones of lakes, where larvae graze in 1–5-cm layers of detritus with abundant macroscopic spherical colonies of cyanobacteria.

***Thienemanniella*:** Similar to *Corynoneura* species, larvae of this genus are among the smallest and fastest growing Chironomidae. *Thienemanniella* are widespread and recorded from a broad array of aquatic habitats, including two species known from springs, splash zones, and small pools (Ferrington et al. 1995).

***Eukiefferiella*:** Larvae of *Eukiefferiella* tend to be most species-rich and abundant in small cooler streams with good-to-excellent water quality. Larvae of *Eukiefferiella claripennis* (Lundbeck) are known to construct larval cocoons (Madder et al. 1977).

***Micropsectra*:** Larvae of *Micropsectra* can be species-rich and abundant in small cooler streams with good-to-excellent water quality, including trout streams (Anderson and Ferrington 2012). Larvae are known to have desiccation-resistant eggs (Williams and Hynes 1977) and larvae that can survive in dried streambed sediments (Williams and Hynes 1976).

***Polypedilum*:** Although species of this genus are common in lakes, ponds, streams, and rivers, at least one species is known to have adaptations to resist the effects of desiccation. One African species can survive extreme dehydration as larvae and persist in a cryptobiotic state for more than 15 years (Hinton 1951).

***Heterotrissocladius*:** Species of the genus *Heterotrissocladius* tend to be most species-rich and abundant in oligotrophic to moderately mesotrophic lakes, especially larger and deeper lakes. However, *Heterotrissocladius boltoni* (Sæther) is known from temporary streams and vernal pools in Ohio (Sæther 1992).

***Diamesa*:** Larvae of species in this genus are commonly encountered in mountain streams or river systems in higher latitudes in North America. At lower latitudes, larvae develop during winter, and can be most common in smaller streams with good-to-excellent water quality. *Diamesa mendotae* (Muttkowski) is common during winter in springs and trout streams of the Driftless Region of southeastern Minnesota (and possibly Wisconsin).

Chironomidae – Isle Royale
New genus detections continued across all months (April to October) in 2009, while new genus detections were limited to spring and late summer samples in 2010 (lower solid line in Figure 6). The estimated and detected genera trends level off at around 174 and 202 samples, respectively; suggesting that sampling at that level should detect the majority of genera present. Based on comparisons between 2009 and 2010, in which the numbers of sample sites were 15 and 4, respectively, the number of sampling sites may be less important than employing sampling strategies that effectively target taxa of interest. Many genera were only detected in one of the two years, so long-term sampling that spans many years would be most effective for detecting rare species.

Caution must be taken when comparing 2009 and 2010 data for several reasons: collections occurred at slightly different intervals, each year had different climatic conditions (2009 with a cold spring, varying patterns of rainfall between years), and methods became more refined in 2010. Comparison between 2009 and 2010 based on zone differences should be legitimate because zone-based sampling did not substantially differ between years.

Chironomid emergence events were often patchy, with emergence observed on some pools but not on others nearby that appeared morphologically similar. While some rock pools were deep enough to retain water during dry periods, even shallow ephemeral pools (<10 cm) had chironomid hatches. Deeper pools maintained insect activity from spring snow melt until regular frosts in the fall.

Most samples were completely enumerated due to low densities; 76% had ≤20 exuviae, the cutoff to determine if subsampling was required. Using a minimum number, such as a 100-count subsample, could also be applied, and has been shown to detect a majority (68%) of richness in lotic systems (Bouchard and Ferrington 2011). Coffman (1973) reported that many of his samples from a stream in Pennsylvania had ≥10,000 exuviae, while few ISRO samples had over 100 exuviae. Only 35 samples (12% of total samples; 19 from 2009 and 16 from 2010) required subsampling. The species accumulation curve suggested 54 genera are present; 46 genera were detected, an 85% detection rate (see Figure 6). With our 2-phase subsampling technique, subsampled individuals were not only randomly collected, but a scan for morphologically unique taxa likely included most generic diversity present in the sample. This should allow us to determine real community differences (Vinson and Hawkins 1996).

Of the 46 genera and subgenera detected, 15 (33%) were detected in only one year (see Table 5). Most of those 15 genera were rare, with only a handful of individuals detected, although larger numbers of *Pagastia* and *Polypedilum* were likely the result of collections occurring shortly after or during a synchronized emergence. Emergences often occurred during approximately the same months between the two years (see Table 5 – gray fill), with low abundances typically occurring outside these months. Many of the common genera emerged continuously throughout the season, a phenomenon which has been found in other studies (Coffman 1973), yet there were often conspicuous peak emergence times. Depending on the goals and target taxa, collection timing is important to avoid missing major emergences or rarities because exuviae probably sink within 7–10 days on coastal pools, depending mostly on microbial breakdown, temperature, and mechanical disturbance (Kavanaugh 1988). Less sclerotized genera will break down faster, so there is likely a bias against these groups (Coffman 1973).

The asymptote shape of the genus accumulation curve shows that we achieved sufficient sampling effort to reveal community diversity, even though new taxon detections are expected with additional effort (Southwood and Henderson 2000). The shape of the species accumulation curve is dependent upon community structure (in this case, emergence of chironomids) during sample collections, so the curve is expected to rise more steeply during peak emergence months (Southwood and Henderson 2000). Abiotic conditions such as inclement weather can also influence the shape of the species accumulation curve; this influence was minimized in EstimateS by randomizing the sample order 50 times without replacement (Colwell 2009). Once the majority of more common taxa were detected in 2009, additions in 2010 were few and focused in spring and mid-summer.

In species-rich communities, many species may go undetected, and true richness may be higher than expected (Tuomisto 2010). Chironomidae generic richness for pools at Isle Royale was higher than we initially expected and equivalent to studies using similar techniques in lakes and streams. Rufer (2007) detected 46 genera in metropolitan lakes in Minnesota, while a large river study in southern Finland, with similar collection techniques and timeframes to ours, detected 51 genera from 3,622 specimens (Raunio and Muotka 2005), which is comparable to our results. Conversely, Bouchard (2007) detected 96 genera/subgenera from biweekly collections over 12 months in lotic woodland systems of southeastern Minnesota. Diversity for rock pools is not substantially different from many studies in lotic and lentic systems in North America and Europe, as summarized in Coffman (1973). With limited substrate, nutrients, and apparently limited niche availability, along with important disturbance from waves and drying, it is surprising that Chironomidae communities are so diverse in coastal rock pools. Rare taxa, though difficult to detect and measure, are valuable to a community if they are impacted (positively or negatively) by stressors, have some specialization, or play a key role in a system (Courtemanch 1996).

The rarefaction curve can help inform managers regarding effort needed to accomplish specific goals of diversity detection. With an estimated genera richness of 54, a goal of detecting, for example, 75% of genera during annual surveys would require approximately 94 samples to accomplish. However, sampling, processing, and identifying specimens is time-consuming, and decisions must be made regarding satisfactory effort to achieve goals. Clearly, not all taxa will be detected, even during a long-term monitoring survey. If ecological conditions change, either naturally or from human-mediated processes like climate change, community components would be expected to change. Species adapted to the new regime may immigrate to the area, while previous species that are intolerant to the new regime may decline or become extirpated.

While Jaccard's analysis does not show a significant difference in communities between zones when 100% occupancy is considered, there is clearly an ecological division where only 12 of 46 genera are shared between the two zones. The analysis becomes significant if species with 90% occupancy for a zone are considered; if 75% occupancy is considered, then a very distinct zonal separation in chironomid communities becomes apparent, with only two genera not appearing to prefer a zone. For these two genera, 68% of *Orthocladius* (*Eudactylocladius*) and 73% of *Corynoneura* individuals occupy the splash zone. Rarities—those with only one or two individuals detected in both years—account for 12 (26%) of the total genera. But the rarities are not driving results strongly in terms of significance to zonal stratification.

Some caution is warranted in interpreting splash zone occupancy. While the chemistry of splash pools and Lake Superior are very similar, some exuviae from genera that prefer oligotrophic conditions could have washed into pools prior to collections, resulting in false positives. Even if several genera show no sign of using splash pools for development, zonal preferences appear strong for nearly all genera. *Corynoneura* and *Heterotrissocladius*—expected from deep, oligotrophic lakes—were detected as larvae in pools from both zones and can therefore be safely included as members of the rock pool community. However, *Protanypus* and *Sergentia*, expected from the same habitat, are only known from small numbers of exuviae in splash zone pools and may not be pool inhabitants.

Pool type was not shown to be important ecologically, with communities in different pool types being statistically similar. Many genera occupied both pool types, which was surprising since ephemeral pools are likely to be dry after 2–3 weeks without rainfall or wave splash. No common genera stratified exclusively into one pool type, although some did appear to have a preferrence: *Cricotopus*, *Corynocera*, *Dicrotendipes*, and *Paratanytarsus* preferred permanent pools, and *Orthocladius* (*Eudactylocladius*) preferred ephemeral pools.

Richness and diversity differences between sites must be viewed with caution as abiotic conditions varied between the two years. The only site with samples from both years— ISRO's Raspberry Island (designated RA in 2009 and RS in 2010)—varied strongly in genus richness, with 15 genera detected in 2009 and 27 detected in 2010. Yet Simpson's index suggests that diversity was similar at Raspberry Island between years. Blueberry Cove and Raspberry Island had the highest genus richness, but in terms of diversity they either did not stand apart or were somewhat lower than several other sites.

The low richness and diversity in the West Caribou to Outer Hill Island area, and on Bat Island, may be a result of limited habitat. Yet Davidson and Shaw islands also had limited habitat with a higher diversity, and the area from Scoville Point to South Government and Third islands have abundant habitat with only low-to-moderate richness and diversity. When comparing the number of individuals collected and the proportion of total genera detected at each site, the most productive sites for sampling included Blueberry Cove, Smithwick Island, Raspberry Island, Bat Island, South Government Island, Blake's Point, and Passage Island.

Time frames for future collecting will depend on personnel and resources available. April and June are most productive, yet June and July have the highest proportion of taxa present. Ideally, samples would be collected monthly in order to detect a large majority of taxa present. Realistically, sampling may only be possible 2–3 times a year, and results indicate April, June, and July–August as good time frames if only three sampling rounds are possible. If only two are possible, April–May and June–July should be considered. July may be an efficient collection month, with a large proportion of genera detected, though annual variation may not follow this pattern consistently. If particular taxa are targeted, their emergence times and conditions, along with habitat type, should be taken into account.

Determining which taxa within Chironomidae might make reliable ecological indicators is challenging in part because there are no prior studies on coastal freshwater pools in the Great Lakes. In addition, identifying appropriate indicators prior to impacts (in this case, oil spills, climate, or direct human manipulation by park visitors) may be challenging, and a single taxon is not likely to be a good indicator of all impacts (Murray et al. 2002). In response to these challenges, many taxa should be chosen to represent the community, or a majority of taxa can be used collectively (Murray et al. 2002). This strategy may be useful with chironomids because some groups should respond in the opposite direction to others, such as *Chironomus* increasing with degrading conditions and most other groups decreasing at the same time. Tracking all taxa is not recommended because there will be increased time, cost, and expertise needed to work with so much variety (Murray et al. 2002). Choosing common chironomid taxa is important because collections of uncommon or rare taxa can be highly variable depending on overlap between sample date and synchronized emergence date.

Many genera were represented by fewer than ten individuals. The two most abundant genera, *Orthocladius* (*Eudactylocladius*) and *Psectrocladius* (*Psectrocladius*), were nearly as abundant as all others combined. The four most abundant and widespread genera are good candidates for long-term monitoring because they should be detected in useful numbers annually (Raunio et al. 2007). Less common but still widespread genera may be used if emergence times can be adequately determined. For example, *Dicrotendipes* was predominantly detected in June (see Table 5), almost exclusively in permanent pools in the lichen zone (see Tables 6 and 7), and were generally found emerging at Datolite Mine and Raspberry Island (see Table 8), making sampling a relatively clear process.

Based on ecological conditions in coastal rock pools at ISRO, the difference between 46 detected Chironomidae genera and the 54 estimated genera (by the accumulation curve) could be represented by several genera of chironomids that are known to occur in springs or other types of semi-aquatic, intermittent, or ephemeral aquatic habitats, and could include one or more of the following genera: *Lapposmittia*, *Hydrosmittia*, *Oliveridia*, *Reomyia*, *Acamptocladius*, *Lappokiefferiella*, *Acricotopus*, *Boreosmittia*, *Chaetocladius*, *Arctopelopia*, and *Thienemannia*.

Macroinvertebrates – Isle Royale

The broader non-chironomid macroinvertebrate community was determined from ad-hoc collections during sampling, a sampling strategy that does not address abundances. The most common groups noted during collection, which probably reflect the general community utilizing rock pool habitats at ISRO, include Collembola, Aeshnidae, Corixidae, Gerridae, Dytiscidae, Apataniidae, Hydropsychidae, Limnephilidae, Culicidae, Tipulidae, and Phoridae. Less common but regular groups included Notonectidae and Gyrinidae. Cryptic taxa were probably under-represented due to sampling techniques that targeted Chironomidae. This wider community appears, as with nearly all target groups in this study, to be much more diverse than initially expected.

Chironomidae – Apostle Islands

Taxonomic richness of Chironomidae was lowest in the pools at APIS, where no taxa were unique in our study. The fauna detected showed the strongest similarity to the fauna of PIRO, where 19 of 21 taxa co-occurred. It appears that the fauna were most strongly influenced by dispersal and oviposition of adults from other rock pools more typical of the lichen zone pools at PIRO. However, based on this subjective interpretation, there is no strong indication or expectation that taxa more reflective of trout streams along the southern shores of Lake Superior will actually occur in the pools. Consequently, it seems reasonable to conclude that more intensive sampling of the homogeneous pool habitats on Bear Island and Devils Island would not yield substantial numbers of additional taxa.

These predictions contrast with the results of Chao's model. Output of the analysis predicts that the species/sample curve has not reached an asymptote for the sampling effort, and that several additional taxa could occur in the pools. Additional sampling effort and more routine monitoring of the pools should resolve the conflicting predictions for taxonomic richness of Chironomidae in rock pools at APIS.

Chironomidae – Pictured Rocks

Thirty-four of the 41 taxa detected at PIRO were collected in at least one of the permanent lichen pools at PIRO. Permanent lichen pools therefore serve as the primary habitat for diversity of Chironomidae in rock pools within the park. Nineteen of the taxa consist of genera or subgenera in which one or more species is known to occur in pools or have specialized physiological adaptations as larvae that would be beneficial for persisting in pools that are susceptible to shrinking or drying over long time frames.

Eleven of the taxa collected at PIRO were not found in pools at ISRO or APIS. These taxa are commonly encountered in small-to- medium-sized trout streams in Minnesota, and their occurrence in pools at PIRO is probably influenced by the streams that flow into the park and act as a reserve of adults, some of which occasionally oviposit in the pools. In most cases, these 11 taxa were restricted to, or were more abundant in samples from lichen zone pools.

Most Chironomidae are not considered to be highly selective of water chemistry conditions for oviposition, but some species show slight preferences based on water temperature. Once eggs are deposited, however, embryogenesis and larval growth are strongly governed by water temperature. Consequently, factors considered to most strongly influence composition of Chironomidae include microhabit and water temperature/thermal regimes (Bouchard 2007), with water chemistry playing a smaller but potentially supporting role. Therefore, if the groundwater seepages into the lichen zone pools at PIRO modulate the thermal regime so that it more closely matches stream temperatures, the resulting oxygen equilibrium saturation concentrations may allow several of the 11 taxa to complete their development in these pools compared with pools that have less groundwater influence, warmer summer temperatures and lower equilibrium concentrations of oxygen.

The presence of these 11 taxa more commonly encountered in trout streams suggests that additional sampling effort could result in substantially increased cumulative richness across pools at PIRO. This conclusion, based on our experience in a wide variety of streams in Minnesota, runs counter to Chao's species richness model predictions based on species detected with only one or two specimens. Although the predicted richness seems to reach an asymptote, it is likely that additional stream-dwelling taxa could be shown to inhabit lichen zone pools if more pools are sampled in the future and/or sampling was repeated in a more routine manner.

Zooplankton

Zooplankton populations in coastal rock pool systems of Lake Superior are astounding. These systems are easy to overlook, but highly diverse, dynamic, and teeming with life. In this study, 177 zooplankton taxa were identified from rock pools at ISRO, APIS, and PIRO. Rock pools supported not only great zooplankton diversity but also abundance, with densities up to 78,000 individuals per cubic meter of water. Rotifer diversity was the greatest, with 96 identifiable species. There were 26 species of cladoceran plankton, compared to seven from a much smaller study of pools on ISRO's North Government Island (Van Buskirk and Smith 1991). In contrast, only four species of cladocera were found in a study of ISRO's inland lakes (Larson et al. 2000). Many of the dominant rock pool species are rare or littoral species considered incidental in Lake Superior (Stemberger 1979, Balcer et al. 1984).

Zooplankton communities in rock pools were highly variable over regional and even local scales, where pools near each other had very different species compositions. There were, however, regional patterns in zooplankton distribution that qualitatively relate to geomorphology. ISRO and APIS pool systems were on exposed bedrock, while PIRO pools were in sandstone between open seeps. Only one site at PIRO had pool structure like APIS and ISRO with several pools arranged in zones. PIRO was subject to more disturbances during the study period, with storms moving enough sand to completely cover an entire splash pool zone. At another PIRO site, a storm during September sampling drove waves eight meters over a cliff to pound what was presumed to be a lichen zone pool. The complete story at PIRO is difficult to assess with our data alone because the patterns of diversity we observed may be due to these disturbances. The nearshore and coastal environment at PIRO certainly warrants further study.

Whatever the cause, PIRO rock pools in this survey were significantly less diverse with fewer zooplankton than the other regions. Species present were more cosmopolitan, with only 13 taxa unique to PIRO and few shared with just one other park; the majority (67%) of species at PIRO were found in both of the other parks. Rock pool systems at APIS and ISRO supported 40 and 32 unique taxa, respectively, and shared more with each other (29 species) than they did with PIRO (8 and 10 common species, respectively). While it is certain that the exact numbers and species found will vary from year to year, the patterns are indicative of the ecological relationship among the parks.

APIS showed the highest overall zooplankton density and diversity. Factors that could influence this include the nearshore environment, with at least two diverse zones of plankton communities in Lake Superior near the islands (Kerfoot 1997). The Lake Superior zooplankton samples taken for this study were intended to capture what was immediately available to splash pool zones. Our samples are not nearly comprehensive enough to characterize the nearshore zooplankton community, so the link between rock pool zooplankton and nearshore pelagic plankton at APIS remains a hypothesis. Other regional factors could be responsible, including local weather or density of amphibians or other planktivores.

The most surprising result was the relationship between zooplankton and different pool zones at the regional scale. Splash pools appeared to contain taxa that arrived via wave action and were consequently trapped. These pools are likely to experience severe disturbance during storms, and it seems reasonable that splash zones would be a hostile environment for such poor swimmers as cladocera and copepods. Conversely, some splash zone pools may possibly serve as a sort of refugia or even an area conducive to increased growth that is mostly devoid of major predators. Some organisms could wash into splash pools, thrive there for a short or moderate length of time, then be washed back into Lake Superior to finish out their lifecycle. Lichen zone pools, on the other hand, appeared highly productive and stable. Testing the entire data set at once, however, showed no difference in zooplankton abundance, diversity, or relative abundance of various major groups between the two zones. This is, in part, due to the low power of the statistical tests— an effect directly related to the high variability in the data as power is proportional to σ (sigma). Increasing sample size in this case also increases the patchiness and variability, so traditional ways of increasing statistical power do not hold.

This statistical problem can be solved by limiting the data analyzed either to taxonomic groups or specific sites. Taking a closer look at taxonomic groups showed some patterns but not the expected ones. It makes a great deal of sense, for example, that cladocerans are significantly more abundant in the lichen zone. What is surprising is that *Daphnia* species were found in only ephemeral lichen pools. It is possible that this is due to predation by amphibians in the permanent lichen pools and hostile conditions in splash pools, or that it is simply part of the unexpected variation in these systems.

A close look at other taxa unique to each zone also reveals interesting patterns. One hypothesis was that the splash zone would structure communities with continual disturbance, and for zooplankton it seems reasonable to think that a protist/rotifer-dominated community would do best in these conditions. There were, in fact, quite a few rotifer species unique to splash zones, but even more copepods were unique to this zone. Testate protists and rotifers, on the other hand, make up a large proportion of the unique taxa of the lichen zone. Both splash zone and lichen zone pools have strongly established zooplankton community types (sharing 75 taxa between them, with 42 and 40 unique taxa, respectively). Nearshore copepod communities appear to get into the splash zone (and likely out of it again during storms) and thrive. Some rotifer species follow this pattern too and are likely subject to the same physical forces, but other rotifers are unique to the lichen zone and comprise much of its diversity.

Zooplankton communities appear much more sensitive to pool permanence than splash-lichen zone placement. Including testate protists and rotifers unique to permanent pools, 66 unique taxa occurred in permanent pools compared to 15 taxa in ephemeral pools. However, quite a bit of overlap existed, with 70 taxa found in both pool types. These results suggest that ephemeral pools are full of incidental species with few taxa specifically exploiting this habitat. The most striking exception, as mentioned above, is that all species of *Daphnia* found in this study were in ephemeral lichen pools.

Again, these regional-level differences did not produce significant results with traditional tests. The second way to handle the high variability was to focus the scale of analysis to a specific site. Our detailed analysis of Passage Island illustrates the mechanisms that are likely structuring the rock pool zooplankton communities in other locations. Lake Superior had the highest abundance, but this was driven principally by testate protists. The lichen zone supported significantly higher zooplankton diversity and higher cladoceran abundance and diversity. Seasonal phenology is also easier to see at this scale. Zooplankton of Passage Island were significantly more diverse and abundant in July and August due to cladocera, copepods, and ostracods. Rotifers were significantly less abundant in May but equally abundant in other seasons.

The complexity of rock pool ecology will require a more thorough examination of the data presented here. Despite this complexity, zooplankton communities show a surprisingly robust rock pool ecology with extremely high regional and local taxonomic diversity and abundance. Differences existed among parks at the regional level. APIS supported greater abundance, while PIRO had more shared and fewer unique taxa. We suspect that PIRO is subject to different wave disturbance patterns, terrestrial input from groundwater seeps that dominate the shorelines, or other factors that restrict zooplankton to more cosmopolitan species. Locally, zooplankton communities are structured by permanence and zone (splash versus lichen) but these differences are swamped by regional variation when tested at larger scales.

Diatom Communities

Within the Lake Superior park units, rock pool zonation is strongest along the basaltic shorelines of ISRO. This delineation of pool zones produces significant differences in both pool chemistry and pool biology. We have clearly shown that diatom communities stratify by pool zonation similar to differences noted among the invertebrate (this study) and vertebrate (Smith 1983) inhabitants of rock pools. The diatom communities of rock pools are also unique among the diatom communities that have been studied in the inland lakes of the three park units (Edlund et al. 2011). Conspicuously absent from the rock pool diatom communities are soil diatoms or diatoms that are capable of forming resting structures in response to desiccation. Some species of diatoms including *Luticola* species, *Hantzschia amphioxys*, and *Pinnularia borealis* varieties are commonly found in soil collections and adapted to harsh habitats with only ephemeral moisture. Other diatom species form internal resting structures called spores or internal valves that allow them to perennate through harsh conditions including drying. Some of these species (e.g., *Meridion circulare*, *Aulacoseira italica*, and *Hantzschia amphioxys*) are common in ephemeral habitats such as prairie potholes. During planning of this project, it was anticipated that these groups of diatom species would be common in rock pools given the probability for pools to dry up, but in no pools were soil diatoms or spore formers encountered.

The diatoms of rock pools can be grouped into several categories including generalist taxa that are found in all pool zones and types, taxa that are limited or strongly selected by pool zone, and some species that are characteristic of Lake Superior but may periodically establish viable populations in rock pools. We will discuss first these groups of taxa from the ISRO rock pools and then broaden the discussion to pools at APIS and PIRO.

Generalist taxa that are abundant in both lichen and splash pools at ISRO include *Achnanthidium*, *Gomphonema*, *Navicula*, and large *Tabellaria* species. Of special note in this group are *Achnanthidium* species, which are among the most common diatom species reported not only in splash zone pools but also in river samples and lake periphyton. *Achnanthidium* species are r-selected and pioneer species capable of rapid colonization of habitats and as a group have broad ecological tolerances making them well adapted to living in rock pools. They are also a common component of the nearshore Lake Superior epilithic community. Other generalists, such as *Gomphonema* and *Navicula* represent very diverse genera. Their inclusion as generalists across pool types may be a reflection of poor taxonomic resolution due to limiting identification to the genus level in this initial survey. For some groups of diatoms, identification to the species-level may be necessary to fully appreciate the potential selectivity for rock pools. The large *Tabellaria* species are good examples. *Tabellaria fenestrata* and *T. flocculosa* var. *linearis* are more common in Lake Superior and splash pools, whereas *T. flocculosa* IIIp and *T. quadriseptata* are more likely encountered in softer water and lower pH lichen zone pools.

In addition to the chrysophyte cysts, many diatom genera can be classified as lichen pool specialists at ISRO including *Nitzschia*, *Encyonema*, *Brachysira*, *Eunotia*, *Pinnularia*, small *Tabellaria* species, *Stauroforma*, and *Rossithidium* species. From a broad perspective, this group of taxa is most commonly found in low conductivity and lower pH systems (Camburn and Charles 2000). Again, the chemical differentiation of lichen pools (with lower pH and conductivity, and higher nutrients, chlorophyll, and DOC) from splash pools at ISRO creates strong abiotic and ecological gradients that support this unique group of diatom taxa. Among these taxa are strong acidophiles including *Stauroforma exiguiformis*, *Brachysira microcephala* and *B. rossii*, many *Pinnularia* and *Eunotia* species, and the small *Tabellaria* species such as *T. flocculosa* strains III and IV (Camburn and Charles 2000).

Similarly, ISRO's splash pools also house a specialized diatom community that includes taxa such as *Synedra*, *Encyonopsis*, *Denticula*, *Cyclotella*, *Delicata*, *Cymbella*, *Discostella*, *Eucocconeis*, and *Ulnaria* species. Among this group are planktonic forms that may have originated in Lake Superior, but seem to be very capable of establishing viable and abundant populations in splash zone pools. *Cyclotella* species including *C. comensis* and *C. delicatula*, *Discotella stelligera* and *D. pseudostelligera*, and the larger *Ulnaria* species (*U. ulna* var. *danica*, *U. ulna* var. *chaseana*, and *U. delicatissima*) make up this group of plankters. Other splash pool specialists are also found in nearshore periphyton in Lake Superior, but seem to do very well in splash pools perhaps as a result of warmer temperature, less physical stress, or release from herbivory. Regardless, *Encyonopsis* species (*E. cesatii*, *E. microcephala*), *Denticula tenuis*, *Delicata delicatula*, and *Eucocconeis* species (*E. flexella*, *E. laevis*) are strongly selected for splash pools along ISRO's shoreline. We have also identified one group of taxa called "*Synedra*" which is specialized for ISRO's splash pools. This group of taxa lives as unicells or in rosette colonies attached to substrata and has a contentious nomenclatural history with many of the species being reported as *Synedra*, *Fragilaria*, or *Ulnaria* species. Again, these taxa are often common in nearshore periphyton of Lake Superior, but appear perfectly capable of establishing and thriving in the splash pool environment. Large community shifts were also noted in some of the late season samplings from splash pools that likely corresponded to recent inundation by wave wash. Increased numbers of *Aulacoseira* species, especially *A. islandica*, and *Hannaea superiorensis* (a Lake Superior endemic) signal these late season inundations.

The weaker physical and chemical zonation of pools at APIS was further manifested in weaker separation of diatom communities between pool zones. Similar to ISRO, *Achnanthidium* remained the most abundant genus at APIS, regardless of pool type. Only two taxa, chrysophyte cysts and *Denticula* species, significantly differed in mean abundance between pool types; in the case of APIS they were both more abundant in lichen zone pools. Similar to ISRO, *Delicata* and *Synedra* species tended to be more abundant in splash pools at APIS, and *Eunotia* was more prevalent in lichen pools. Strangely, many of the species that characterized splash or lichen pools at ISRO had opposite preferences for pool type at APIS. For example, *Denticula* and *Cyclotella* were more abundant in lichen than splash pools at APIS, and in contrast, *Brachysira* was more abundant in splash pools. We do note that fewer sampling events and sample sites at APIS restricted our ability to more clearly distinguish diatom communities among pool types.

All pool types at PIRO were strongly dominated by *Achnanthidium* species, similar to ISRO and APIS. *Nitzschia* species and *Navicula schmassmannii* were the only two taxa that differed significantly in mean abundance between lichen and splash zone pools; both were more abundant in PIRO lichen pools. Few additional abundant taxa (*Gomphonema* and *Psammothidium*) showed strong preference for lichen pools. Supporting the patterns we established at ISRO, PIRO splash pools had higher abundance of *Synedra*, *Encyonopsis*, and *Delicata*. But the splash pools also had more *Encyonema*, *Cymbella*, and *Brachysira* than lichen pools. The additional pool types at PIRO's Mosquito Harbor—the cave pool and medicolous zone—were also dominated by *Achnanthidium* species, but contained unique sets of taxa among pools. The cave pool shared some characteristics with splash pools (high abundance of *Delicata* and *Encyonopsis* species), but was further characterized by populations of *Eolimna* and *Kobayasiella* species, diatoms that are indicators of low alkalinity. The medicolous zone pool had high levels of *Cymbella*, *Gomphonema*, *Synedra*, and *Brachysira*—an odd combination of taxa—that likely indicates some flow is present in the pool and that water chemistry is low alkalinity.

Multivariate analysis of diatom communities provides the final support for our contention that weaker physical and chemical zonation of pools at APIS and PIRO compared to ISRO is similarly manifested in the separation of diatom communities among pool zones. Our DCA clearly separates ISRO's lichen and splash zone pools along axis 1; community differences between pool types at ISRO are unambiguous. In contrast, patterns of diatom communities are more muddied at APIS and PIRO. Some APIS lichen pools are separated, for example, along vectors of higher cyst and *Psammothidium* abundance, but the remaining APIS pools are not clearly separated by pool zone. Similarly, the PIRO pools are tightly grouped near the origin of the DCA and show no clear separation by pool zone or type.

Diatom Sampling and Analysis

Sampling of diatoms took place on three dates at ISRO (May, July, and October) and only two dates at APIS and PIRO (May and August/September). In comparing our measured genus richness to the Chao estimator and Cole rarefaction curves, it appears that our sampling does come close to capturing the potential genus richness of the sites. At ISRO, where we sampled the most sites and more frequently, Chao 1 estimates of 61 genera compare favorably to the 59 genera we encountered. At APIS and PIRO, poorer sampling of potential richness was evident, suggesting additional sampling events should be considered to capture the genus richness of rock pools. We also note that with so few sampling events it is difficult to draw any strong conclusions on seasonal changes in diatom communities.

For diatom sampling and analysis, our counting and sampling techniques were qualitative not quantitative, in contrast to the chironomid and zooplankton sampling that occurred simultaneously. Although techniques are available for quantitative sampling and analysis of diatom communities, in this preliminary study of rock pool habitats, we chose not to attempt them. To minimize variability due to microhabitat and spatial features of pools, we chose to sample a single microhabitat (the epipelon at max depth) to keep samples among pools most comparable. We took composite samples, rather than defining a sample size (e.g., a 1.0 cm^2 rock scrub), to further capture spatial variability within pools. Field duplicates were taken for 10% of samples and analyzed to determine our sampling variability. In the laboratory, samples were processed to create evenly distributed microscope slides, but no effort was made to prepare slides quantitatively. As such, diatoms counts represent 500 specimens and final data are presented at percent abundance by taxon. Although many diatom studies use percent abundance or qualitative measures of community composition, differences in pool productivity (as measured by chlorophyll) will not be captured with this sampling and analytical strategy. Lastly, two samples had such low abundance of diatoms in the final microscope slide preparations—Datolite Mine splash pool 1, sampled in both May and October—as to call into question our analytical techniques. These two samples had very few diatoms such that only 144 and 29 diatoms could be counted, respectively. It is likely that this pool was easily and frequently wave washed shortly before our sampling events.

For most ecological studies, the identification of diatoms takes place at the species level. In this study, we chose to analyze diatoms to the genus-level (with a few extra subdivisions) to make diversity measures comparable among our target organismal groups. Several recent studies (Hill et al. 2001, Potter et al. 2006) have considered or used diatom genus-level idenfications for community analysis. The critical results in our rock pool study were that we were able to characterize pool zones using genus-level identifications and that diatom communities described at the genus level strongly supported physical and chemical gradients among parks and pools. It is anticipated that additional analyses will be completed to formally compare community descriptors and analysis based on species- vs genus-level identifications.

Physical and Water Quality Characteristics of Pools

The geologic setting of the Lake Superior national parks creates three very different rock pool settings. At ISRO, the basalt shorelines slope up from the lake and produce within the cracks and depressions a zone of nearshore splash zone pools clearly delineated from upgradient lichen pools. Although there is no difference in size or depth of pools between lichen and splash zones, they strongly differ from a geographic standpoint; ISRO lichen pools are significantly closer to treeline and farther from shoreline than splash pools. This simple arrangement is what we define as classic Lake Superior rock pool habitat, and which has been observed to be virtually identical to numerous sites on the Minnesota shore (Egan, unpublished data). However, due to bedrock differences this is not how the shorelines are arranged at APIS or PIRO. Smith (1983) and Van Buskirk and Smith (1991) did not differentiate permanent and ephemeral pool types or lichen vs splash pools in their studies on ISRO rock pools. They censused all pools along their study sites at Edwards and North Government islands and monitored relationships among pool persistence (accounting for both desiccation and wave wash), location, and volume as it related to amphibian breeding and predation.

They similarly found no relationship between pool size (volume) and location relative to shore and treeline.

At APIS, sandstone bedrock at Bear and Devils islands creates a much narrower and table-like rock pool zone that has very little slope away from the lake. There, again, is no clear differentiation in size and depth between lichen and splash zone pools (although APIS pools are shallower than ISRO pools), but the delineation of lichen from splash pools is much less clear compared to ISRO. Lichen pools are somewhat farther from the shoreline than splash pools at APIS, but much closer to the lake than ISRO lichen pools. In contrast, both pool zones are similarly close to treeline at APIS, a testament to the much narrower rock pool zone on these sandstone islands.

The shoreline habitat at PIRO is also less conducive to rock pool formation compared to ISRO. Underlain by sandstones of various ages, the sites sampled in 2010 were the only areas in the park with rock pools. Pools are similar in surface area to ISRO and APIS, but they are much shallower. Furthermore, ground water seeps create additional shallow shoreline pool types at PIRO, what we termed the cave pool and medicolous zone at Mosquito Harbor. Last, the splash pools at PIRO appear to be more temporary features; by the August sampling, splash pools at Miners Bay and AuSable Point had been inundated with sand deposits.

Hydrology of rock pools is most strongly controlled by wave inundation, precipitation, runoff, and groundwater sources. Pool zone topography and geology further influence the hydrology and ultimately the water chemistry of the pools. At ISRO, where lichen and splash zone pools are clearly delineated relative to lakeshore and treeline, the water chemistry further supports the separation of these pool zones. Splash zone hydrology appears to most strongly reflect wave inundation, as the water chemistry of splash pools is identical to Lake Superior waters with regard to conductivity, TP, TN, SRP, ammonium, DIC, and DOC. Splash pools, like Lake Superior waters, also have low levels of chlorophyll and so exhibit low productivity. In contrast, lichen pool hydrology at ISRO—with significantly lower conductivity, pH, NO_x and DIC than splash pools and Lake Superior, but higher TP, TN, DOC, ammonium, and overall productivity—is more strongly controlled by direct precipitation and runoff, by greater pool permanence (allowing enhanced productivity), and by an increased influence of terrestrial and authigenic carbon sources. Our observations on rock pool hydrology are confirmed by Smith (1983) and Van Buskirk and Smith (1991). They monitored and estimated pool persistence (in terms of biological stability, not hydroperiod) and found that, except for very small pools (<10 L), pools nearer to the shoreline had shorter permanence most often interrupted from wave wash, regardless of pool volume. Pools with >10 L volume that were located more than half-way from shore to treeline were more persistent if they were larger, reflecting hydrology more strongly controlled by precipitation and runoff.

The weak physical zonation of APIS pools is further manifested in pool hydrology and water chemistry. Whereas APIS lichen pools have lower conductivity, pH, NO_x and DIC than splash pools and Lake Superior, the splash pools are not nearly as similar in water chemistry to Lake Superior as on ISRO. Rather, APIS splash pools represent a transitional condition between Lake Superior and the lichen pools, the latter of which have higher TP, TN, SRP, DOC, and chlorophyll. Although we only sampled APIS pools in May and September, it appears that the hydrology of splash pools is not as strongly controlled by wave wash, but instead may be controlled by runoff and autochthonous inputs (higher DOC and TP, lower DIC and NO_x).

Weak pool zonation and multiple pool types create additional hydrologic options on PIRO's sandstone shorelines. High variability in lichen and splash pools for DIC, DOC, conductivity, TN, and SRP support the poor zonation and hint at hydrologic relationships that combine wave wash, precipitation, runoff, and groundwater seeps as potential water sources for pools. PIRO splash pool chemistry bears little resemblance to Lake Superior waters compared to ISRO splash pools, and lichen pools are highly variable in TP, SRP, and chlorophyll at PIRO, suggesting that pool persistence may be a strong factor to consider among PIRO pools. The cave and medicolous pools at PIRO have many indicators of hydrology controlled by groundwater including cooler temperature and lower DOC, DIC, SRP, and TP compared to their neighboring lichen pools.

Nutrient limitation has not been considered previously in any Great Lakes rock pool settings. Lichen pools at PIRO were the only pools that consistently showed N-limitation or N-P co-limitation among the three parks and among all pool types. Some APIS lichen pools also showed N-P co-limitation during our sampling. Lake Superior and all splash pools were always strongly P-limited suggesting that lichen pool hydrology, permanence, chemistry and ultimately pool productivity can shift nutrient ratios toward N-limitation (even ISRO lichen pools were much less P-limited than their splash pool counterparts). We discuss below the impact of these nutrient resource trends on diatom communities and their ecology.

The thermal behavior of Great Lakes rock pools has also not been considered previously. We assumed that all rock pools we investigated freeze solid during winter, although we note this has never been confirmed. Localized climate moderation along the shoreline due to the influence of Lake Superior might allow pools to be only ice-covered in the winter; this would of course strongly influence pool physical structure and open water season biology. During our scheduled sampling of rock pools in 2010, splash and lichen pools were typically warmer than Lake Superior by 5–10 °C, with maximum measurements of 27.1 °C at PIRO (a lichen pool), 23.8 °C at ISRO (splash pool), and 16.1 °C at APIS (lichen pool). We saw no indication of thermal stress during regular sampling among our target organisms, and these temperatures seem to bring dissolved oxygen levels only occasionally below 80% saturation. More telling was the continuous thermistor data that we collected at ISRO's Blueberry Cove pools in 2012. During the early sampling season, splash zone pools were slightly cooler than lichen pools, and they showed periods of rapid cooling that were likely instances of wave wash. During the summer months, splash and lichen pools appear to have similar daily average temperatures, but their diurnal swings in temperature were very different. During late June through mid-July, lichen pools warmed as much as 15 °C daily and reached temperatures over 30 °C many times. In contrast, splash pools warmed less than 10 °C daily and only regularly reached 28 °C. Nighttime cooling during mid-summer was less in splash pools as well, with nighttime temperatures staying a couple of degrees warmer than lichen pools. The accentuated warming of lichen pools is likely a consequence of (a) their greater absorbance due to DOC staining, and (b) the moderating effect of Lake Superior (cooler right near the lake during the day and comparatively warmer by the lake at night). Lacking in our interpretation of these data are any links to diurnal stresses that temperature and oxygen levels may have on pool organisms. With climate predicted to warm and indications that pools provide critical habitat for amphibian breeding, rare and threatened plants, and disjunct organisms, it becomes paramount that we understand diurnal and seasonal characteristics, monitor interannual pool condition, and develop a better understanding of pool ecology.

Coastal Habitat Mapping

As with nearly all aspects of the rock pool study, mapping was more complicated and interesting than we expected at the outset of the project. To get a stronger sense of what amount and type of habitat was broadly available for rock pool aquatic communities, we decided to map all pools along the south shore of Isle Royale between Passage Island and Schooner Island. The south shore was most likely to contain good quality habitat due to bedrock type and slope, with the north side of Isle Royale generally steep and cliff-like and the west end of the island composed of conglomerate bedrock that is not conducive to pool formation. With the international shipping lane between Passage Island and Blake's Point, this area of the park is also most susceptible to shipping spills.

The 71,931 pools were far more than expected. The fact that Passage Island contained 45,164 of these pools (almost 63%) was astounding. Further, the dominance of the chorus frog among the amphibians inhabiting Passage Island, clearly makes this one of the most critical areas of shoreline to protect from pollutants. No credible observations of chorus frog occupation and breeding have been reported from inland habitats at ISRO, despite many years of nighttime amphibian surveys by park staff. Although chorus frogs and blue-spotted salamanders exhibited the greatest abundance and widest range of amphibians in pools, they are present in only about 3% and 1% of pools, respectively, which means that negative impacts to occupied pools could have important implications for these populations. Coastal pools do not appear important for any other amphibian species at ISRO.

Conclusions and Recommendations

Study Design

Because abundant chironomid genera were similar between years, we suggest sampling for macroinvertebrates at fewer sites more intensively, and with replication at each site (Murray et al. 2002). If spaced over several years, this amount of sampling should be easily accomplished with a moderate personnel and time commitment. For general diversity monitoring, collections should occur about one month apart from ice-out until mid-autumn. If collections must be limited, a three-sample design is suggested, with collections occurring in May, June, and July–August. The emergence patterns of target groups should be determined before a sampling program is initiated so sample times coincide with likely emergence times. A single, well-timed sample can be very effective and may even detect a relatively large proportion of the community, but a solid understanding of emergence times and conditions is required to do this (Raunio et al. 2007).

For Chironomidae, the pupal exuviae collection method appears to collect a large majority of genera present in coastal rock pools. Because this method is relatively inexpensive, taxa and habitats can be targeted, the community is not impacted by removal of larvae or adults, and exuviae are identifiable to genus and species, it should continue to be used for rock pool studies. Contracting tasks such as slide mounting and, more importantly, identifications to a laboratory with expertise in these groups is suggested. Enumerating entire samples is a realistic goal due to typically low sample abundances, and the two-phase subsampling process will give the best results for detecting diversity present in samples. Identifications to genus will be more cost effective and faster, although this lower resolution may gloss over some important information, particularly for species-rich groups like *Tanytarsus* and *Orthocladius* (Raunio et al. 2007).

The study design worked very well for zooplankton community analysis, representing a balance between regional coverage and site-specific detail. Sample pool distribution did not appear significant at the regional scale, but this is a sign of the diverse and dynamic zooplankton population regionally. Local site-specific analysis shows that the zonal study design does have biological significance for zooplankton.

Design of sampling nets produced strong results, with no sample pool absent of organisms. The 30 µm mesh size was a good compromise and captured far more rotifers than previously found in rock pools or in Lake Superior itself. Testate protists were adequately sampled by this mesh size. Serious study of the protists, however, would require far different sampling techniques. Not only a smaller (or no) mesh would be required but live samples are preferred since the ethanol preservative destroys non-testate protists. Our work underestimated protist diversity.

For diatom sampling, a common community among rock pools (detritus or epipelon) was identified to make sampling efforts among parks, sites, and rock pool types as comparable as possible. Had other microhabitats in the pools been sampled, we surely would have increased our estimates of richness and diversity. No efforts were made to sample diatoms quantitatively, and it is not clear how this could be easily accomplished given that there is little diatom plankton development in the pools and likely high spatial variability in diatom abundance within pools. Only two pool samples did not permit full analysis. These were from a single splash pool at Datolite Mine on different sampling dates and were likely reflecting low diatom abundance due to recent wave wash. Analytical procedures for diatoms coupled with consistent sampling technique produced strong support for one of our major hypotheses—that diatom communities differed between lichen and splash zone pools. These differences were most marked at ISRO, where physical, hydrological, and chemical differences in pools between zones supported strong differences in biological communities.

Our methods of field and water quality sampling worked very well during the intensive 2010 sampling even with having multiple types of sampling gear and multiple groups of researchers taking and processing the samples. Key to this level of success was pre-season planning for fieldwork; provision of prelabeled collection and sample vessels; careful preparation of SOPs and equipment for sampling, processing, and handling of samples; and minimal numbers of highly qualified analytical laboratories. The resulting data are of high quality and met QA/QC guidelines used in each lab. These sampling and processing techniques are highly recommended for any type of wilderness sampling where full lab facilities are not available. As for logistics, it appears that sampling and processing of samples from a single site is a reasonable daily goal. Additional analysis of our water quality data (in particular trace metals, anions, and cations) is necessary to prioritize what additional parameters are best suited for rock pool monitoring.

Mapping was very revealing regarding the densities of pools along coastal areas of ISRO and distribution of amphibian populations. Limited analysis is included here, but funding for further study of this unique dataset is being pursued. Expected analyses include determining areas of shoreline that require additional protection in the event of an oil spill or other pollution, geographic distribution differences from main island to more distant island sites, and the pattern of habitat use by amphibians.

Study Sites

Murray et al. (2002) suggested focusing study efforts on habitats and locations that have high value or are sensitive to stressors, either from a human or ecological perspective. Based on chironomid abundances and diversity, the most appropriate sites for long-term monitoring at ISRO are Blueberry Cove on the south shore, Smithwick/Raspberry Islands, South Government Island, and Blake’s Point. Passage Island is an ideal candidate for long-term study due to its remote location, extremely abundant habitat, presence of at least one species of management interest (boreal chorus frog), proximity to shipping lanes and potential pollutants, and a cold, lake-influenced climate. For zooplankton, Blueberry Cove and Passage Island are good sites for further ISRO work. Diatom communities have the highest richness and diversity, and strongly differentiated splash and lichen pool communities at Blueberry Cove and Passage Island, supporting the choice of these sites for further work. There was strong physical and chemical separation of rock pools between splash and lichen zones at all of the 2010 ISRO sites; however, the Passage Island and Blueberry Cove sites had strong separation of pool zones based on chemistry and field measures.

At APIS and PIRO, the sites we sampled are likely the best sites for future studies, although a long-term monitoring project should also assess pool densities in other locations. The Stockton Island site at APIS should also be strongly considered for further coastal rock pool work. It is important to note that PIRO is a unique system with few rock pool sites, different rock pool types, less permanence in splash pools, and greater influence by groundwater. These discrepancies require a modified study design (i.e., we could not identify a single site with two classic lichen zone pools and two seasonally permanent splash zone pools), and PIRO should be treated independently in the future.

Communities and Implications for Rock Pool Ecology

Chironomidae

Chironomidae abundances were low compared to year-long, temporally-intensive studies in lotic systems (Bouchard 2007, Coffman 1973). Nonetheless, diversity was higher than expected based on limited nutrients and low apparent niche availability in these habitats. Nineteen genera form the core of the chironomid community across the three parks, and several of these are known to survive in desiccation-prone habitats. Diversity at ISRO is similar to many studies in other habitats, which is interesting given the small volume of collective habitat available in pools compared to river and lake systems. It is likely that proximity to other habitats increases richness.

There is a clear ecological importance to zonal differences for Chironomidae at ISRO and PIRO, both for the entire community and for nearly all individual genera. Shipping pollutants are most likely to impact the splash zone, which has unique taxonomic components not present in the adjacent lichen zone pools. As a result, recolonization following impacts would require longer-distance dispersal events from potentially distant non-impacted sites.

We found no differences between permanent and ephemeral pools within each zone in any of the parks, although some rare genera did stratify based on this difference. Most results show significant similarity between these pools, suggesting that recolonization to habitats impacted by pollution should be possible from nearby pools of any type within the same zone. Depending on the genera targeted for long-term monitoring and the sampling limitations, effort may be saved by lumping samples by zone at a site instead of distinguishing pool types within a zone.

Zooplankton

Compared to other rock pool systems (e.g., Dodson 1987) the rock pools of ISRO, APIS, and PIRO show a staggeringly high diversity and abundance. The splash-lichen structure also appears unique although there may be analogues in studies of tidal pools. The unique zooplankton community structure found in this study has important implications for the advancement of ecological science. Managers of biological resources and park visitors are likely to be surprised at the diversity and complexity of rock pool systems. Managers can track zooplankton communities to monitor various environmental changes (discussed below) and the impacts these changes have on higher trophic levels.

The surprising zooplankton dynamics in rock pools revealed by this study show that these systems are far different than previously thought. Rock pools have been characterized as good systems for "natural experiment" because of their simplicity (Brendonck et al. 2010). The results from Lake Superior rock pool systems suggest otherwise. First, rock pool communities are not ecologically simple. Second, they are not small reflections of larger lake systems. The results discussed above show that rock pool zooplankton form unique communities in these systems. These rock pools do offer an opportunity to study fundamental ecological processes if care is taken to account for their unique community structure.

The functional role of zooplankton communities in rock pools, and the connection between these coastal habitats and Lake Superior, inland lakes, and terrestrial environments remain to be examined. Future work with the data from this study will help, while a combination of future monitoring and detailed study will sort out many of the questions raised here. The high diversity and abundance of zooplankton in rock pools provides a solid baseline for monitoring future changes or impacts.

Diatoms

Strong differentiation of diatom communities supported the physical, hydrologic, and chemical differences between ISRO's lichen and splash zone pools. Although the genus *Achnanthidium* was usually the most abundant among all parks, sites, and pool zones, other genera have strong specificity for pool zones and together produced communities that characterized each pool zone. Lichen pool diatom communities could be characterized as indicators of low conductivity, acidophilous, moderate productivity, and stained DOC conditions. This community included taxa such as chrysophyte cysts, *Encyonema*, *Brachysira*, *Nitzschia*, *Eunotia*, *Pinnularia*, *Tabellaria*, and *Stauroforma* species. Many of these taxa were absent from splash pools. Splash pool communities reflected sources that could be connected to Lake Superior. *Delicata*, *Synedra*, *Encyonopsis*, *Denticula*, and *Cymbella* species, although commonly found attached to rocks in the nearshore zone of Lake Superior, appear to be capable of thriving in nearshore splash pools. Splash pools also house planktonic forms that are common in nearshore Lake Superior waters including *Cyclotella*, *Tabellaria*, and *Ulnaria* species. Missing from the rock pool communities were soil and resting spore forming species that might have been expected in pools that are subjected to periodic drying.

The weak zonation and greater diversity of pool types at APIS and PIRO creates poorer physical and chemical differentiation of pool zones at these parks. As such, diatom communities are also less distinct among pool zones and types compared to ISRO. Groundwater influence at Mosquito Harbor (PIRO) was also evident in the other pool types (cave and medicolous pools), as indicated by the presence of taxa associated with flowing and cooler water (*Meridion*, *Cymbella*, *Gomphonema*, *Diatoma mesodon*). Fewer sites and sampling events at PIRO and APIS may have further minimized our ability to fully characterize and differentiate diatom communities among pools and pool zones.

Water Chemistry

The geological and topographic setting at ISRO produces many shoreline reaches that support rock pool habitat and creates well delineated lichen and splash pool zonation. As such, differences in rock pool zones are most evident at ISRO where lichen pool chemistry reflects hydrology controlled by precipitation and runoff. Water quality that includes higher nutrient (TP, TN), DOC, and productivity levels and lower conductivity, DIC, and NO_x further differentiates lichen pools from splash pools. In contrast, splash pool hydrology is most directly influenced by wave inundation as splash pool chemistry shares much in common with Lake Superior waters.

At APIS and PIRO, rock pool habitat is less abundant, typically occurs across a narrower zone, and the physical separation of rock pool zones is less clear. Differences between types are also less evident at APIS and PIRO where the weak zonation creates more of a gradient in pool chemistry as one moves from lakeshore to treeline. In addition, the rock pools at PIRO show evidence of hydrology controlled by groundwater inputs and other types of pool habitat (cave and medicolous pool types).

Target Taxa and Parameters for Monitoring and Future Study

Focal species or genera should be: 1) dominant within a zone or other stratification of interest, 2) ecological keystones that other community members rely on (such as prey species), 3) common and abundant at many sites, 4) relatively well known for life history, and 5) practical to monitor (Long and Mitchell 2010). Murray et al. (2002) also suggest using indicator species that would either be highly susceptible or highly resistant to anticipated effects of disturbances such as oil spills or warming local climates in the current study. Additional considerations for choosing focal groups include range limitations (e.g., on the southern edge of the known range), disjunct taxa that are not otherwise known to the region, limitations of habitat or niche (e.g., cold-adapted or drought-prone), and general rarity of the genus throughout its range. Many of the additional considerations relate directly to potential population declines in response to regional climate warming.

Because of their regular use as biological indicators of aquatic systems, a scale was used to rank Chironomidae genera collected from Lake Superior parks (Table 23). Based on considerations listed above, genera that fit over 50% of the considerations were listed as potential groups for long-term monitoring (Ferrington et al. 2008, Wiederholm 1986). These groups should be targeted based on known emergence sites and times. Chironomidae can be challenging to identify but they are ubiquitous, diverse, and individual species have highly variable responses to ecological conditions. These variable responses can help managers understand ecological changes occurring in coastal habitats, although species-level identification may be necessary for success. This process, along with more in-depth analyses, should also be accomplished with other macroinvertebrates that are commonly encountered in pools, such as Corixidae, Culicidae, Dytiscidae, and Limnephilidae.

No individual zooplankton species stands out as an indicator of rock pool ecosystem health (other than the inverse need to monitor for invasive species like *Bythotrephes* and their impacts). Communities of cold water stenotherms, particularly rotifers, may be useful but their distribution is very patchy. It would be more productive to use future monitoring of communities (composition, diversity, and abundance) to track changes in rock pool systems. The first step is to more comprehensively analyze the current data for other environmental gradients that help explain community dynamics. Second, longer term monitoring will more accurately describe community variation and identify species unique to different zones. Community ecology linking rock pool community elements would greatly improve understanding of how zooplankton distribution supports rare, threatened, and endangered species. Conversely, invasive zooplankton were found in pools at APIS in a non-study system (Stockton Island). Rock pools may offer a unique natural laboratory for examining the immediate impacts of invasive species in small systems. Finally, the extraordinary diversity of zooplankton in the rock pools warrants a closer look at the interaction between coastal and nearshore aquatic environments.

There are reasons to particularly track *Daphnia* species in the pools. If *Daphnia* consumption mediates chytrid infections of amphibians (Buck et al. 2011), the presence of *Daphnia* in rock pools could be important because these systems are likely a major breeding and development area for amphibians at ISRO, APIS, and PIRO. Ironically, many amphibians, and newts in particular, eat *Daphnia*. The absence of this genus from permanent lichen pools could in fact be due to their nocturnal feeding to avoid predators (a simple test would be to visit pools at night). The importance of this relationship and applicability to these rock pool systems needs to be addressed. Critical failures of amphibian populations in recent years warrant a quick investigation.

Climate change will also certainly impact zooplankton communities. The relationship between zooplankton and permanence of the rock pools shows that as a whole, this community will be a strong indicator of changes relating to increased temperatures and evaporation rates. Again, whole community assessment is key, and our results highlight how spatial scale needs to be considered in order to properly detect changes.

Zooplankton distribution is highly variable and it does not seem possible or appropriate to direct emergency response directly at this group. Zooplankton are rapid colonizers and will serve very well as a means of assessing recovery of coastal rock pools from localized disturbance (e.g., an oil spill). First, the structure of zooplankton communities shown in our results indicates the different groups of organisms expected in permanent or ephemeral pools. This information can be used to track recovery. Zooplankton community structure as a whole is an important measure of recovery, particularly because of their critical role in the food web for higher organisms. Our results also show which areas are most alike for purposes of comparing recovery based on community composition and response to local variables.

Table 23. Target Chironomidae genera/subgenera for coastal rock pool monitoring at Isle Royale National Park, based on a ranked scale of traits. All other genera collected did not meet ranking guidelines for consideration.

Genus/subgenus	Common	Ecological importance	Dominant in pool subsets	Known life history	Practical to monitor	Range limitations	Disjunct	Limited habitat or niche
Ablabesmyia	Yes	Predatory larvae	Lichen permanent	Yes	Yes	Widespread	Not for genus	No
Corynoneura	Very	Tolerant of low DO	No	Yes	Yes	Widespread	Not for genus	No
Cricotopus	Very	Often tolerant of stressors and cold adapted	Splash, permanent	Yes	Yes	Widespread	Not for genus	No
O. Eudactylocladius	Very	Occupies many lentic systems	No	Yes	Yes	Widespread	Not for genus	Often in small pools or water films
O. Orthocladius	Somewhat	Some are very cold adapted	Splash, ephemeral	Yes	Yes	Widespread	Not for genus	No
Parakiefferiella	No	Often needs good water quality	Splash	Yes	Maybe, limited ISRO range	Not well documented in Midwest	Uncertain	No
P. Psectrocladius	Very	Some are warm adapted and tolerant of low DO	Lichen	Yes	Yes	Widespread	Not for genus	Standing water
Chironomus	Yes	Often warm adapted and tolerant of pollution	Lichen	Yes	Yes	Widespread	Not for genus	No
Neozavrelia	Somewhat	Cold-adapted	Splash permanent	Yes	Yes	Not known in Midwest?	Yes	Muddy sediments in cold lentic
Dicrotendipes	Somewhat	Often warm adapted and tolerant of pollution	Lichen permanent	Yes	Yes	Widespread	Not for genus	Lentic
Glyptotendipes	Somewhat	Often warm adapted and tolerant of pollution	Lichen	Yes	Maybe, low abundances at sites	Widespread	Not for genus	No

Diatom richness and diversity in our rock pool sites were not as high as those from some other Great Lakes sites. For example, richness can exceed 150 taxa (species-level identification) in deepwater benthic diatom collections from Lake Michigan (Kingston et al. 1983). However, diversity of rock pools is similar to what was found in surface sediments collected from inland lakes at ISRO, APIS, and PIRO (Edlund et al. 2011). Most importantly, rock pool diatom communities represent unique communities that can be characterized among pool zones and types. It is this characteristic that should inform future monitoring approaches; breakdown of the distinction between pool zones (biologically and chemically) would indicate gross ecological change to the nearshore region. It is difficult with the data in hand to target specific taxa for monitoring. Some of the most interesting communities, such as lichen pools with abundant *Stauroforma* are more representative of individual pools. For example pool L2 at Passage Island had 21–33% abundance of *Stauroforma* during 2010, while its L1 counterpart had only 0–1.8% abundance of that genus.

Limiting our ability to designate sites and species for diatom monitoring is the lack of data to determine interannual and even within-year variation within and among pools. Dominance by r-selected taxa, probability of full winter freeze, and notable differences even within pool types at one site begs for sampling in multiple years and in slightly more annual detail on some pools to fully understand seasonal and interannual dynamics. Questions remaining include: do pools have regular diatom assemblages each year? How quickly does a wave washed splash pool generate a typical splash community? And what determines the diatom community structure each year in lichen pools? The minimal sampling that we did at PIRO and APIS only scratches the surface of rock pool ecology at those sites. With greater sampling frequency, would we find that pool communities are more clearly segregated, as seen at ISRO? Are groundwater-controlled pools an additional type that needs to be considered in Lake Superior parks? We only sampled these sites at PIRO's Mosquito Harbor, but there were also shoreline seeps present at APIS, other sites at PIRO (Sand Point), and up to 4,931 seep-influenced pools identified along the south shoreline during rock pool mapping at ISRO.

Our genus-level analysis of diatom communities was able to readily characterize differences among pool zones. However, in most studies, species-level identifications are used and may provide greater detail on real diversity and better characterize pool communities. More importantly, many of the diatoms that were abundant in pools show cryptic patterns of diversity and need to be studied in greater detail. For example, some of the *Achnanthidium*, *Delicata*, *Synedra*, *Gomphonema*, small *Cyclotella*, and *Brachysira* species encountered could not be readily assigned to known species and likely represent species new to science. New species of Great Lakes diatoms continue to be described, especially when previously understudied habitats are sampled (Bixby et al. 2005).

Although diatoms are a common primary producer in rock pools, other algal groups are also notably abundant in pools. Green algae form visible growths in some pools and may be rapid indicators of nutrient addition. Cyanobacteria are also common in pools, especially ephemeral pools, and likely contribute ample primary production and nitrogen to pools through nitrogen fixation. The significant abundance of chrysophyte cysts in lichen pools at ISRO and APIS warrants some survey efforts to determine their species-level diversity. Similar arguments could be made regarding cryptophytes in splash pools as they can be a significant component of Great Lakes plankton.

Our field measures and water quality sampling provided the first characterization of the chemical and many physical aspects of rock pools. Field techniques and sampling procedures were carefully developed to easily and rapidly characterize rock pools; these methods provide a template for monitoring of rock pool conditions. Stark differences between chemistry of ISRO lichen and splash zone pools were readily apparent with only three samples from one year and suggest that conductivity, DO, pH, nutrient levels, productivity, DIC, and DOC are important parameters for monitoring, characterizing, and inevitably structuring the ecology of rock pools. Other environmental parameters that we measured can also be considered for future monitoring of pools (e.g., chlorophyll and thermal characteristics).

Similar to diatom sampling, our understanding of interannual, within-year, and even diurnal extremes in water chemistry is limited with our current data set. Continuous thermistor data from select ISRO pools show how thermal behavior varies within and between lichen and splash pools on a daily basis, but does not begin to consider the other chemical parameters or ecological impacts on oxygen content and thermal stress to organisms. Deploying thermistors year-round would also help determine whether pools freeze solid in the winter. We also have no understanding of pool persistence in our sites. Smith (1983) showed how pool persistence varied by pool volume and location relative to shore and treeline. These areas should be studied further, given current climate trends and future scenarios of predicted warming and loss of ice cover near ISRO (Allan et al. 2012).

Patterns of nitrogen deposition have changed in recent decades in the Great Lakes region (Stottlemyer et al. 1998, Toczydlowski and Stottlemyer 2009). Measures of nutrient limitation indicated that lichen pools in some parks were N-limited or N-P co-limited. Monitoring should include assessment of potential nutrient limitation.

The apparent hydrologic differences between lichen and splash zone pools, as well as the potential for terrestrial inputs of carbon from allochthonous sources in lichen pools, most likely create a gradient in food web dynamics that could be incorporated into a monitoring scheme. Carbon and nitrogen isotope studies of pool biota may inform our understanding of food web dynamics across pool zones and types.

Finally, a broader synthesis of these data sets is required to fully appreciate patterns of diversity, food web dynamics, and relationships between physical, chemical, and biological patterns in rock pools. Understanding the complexity of these so-called simple ecosystems is crucial to understanding how they will respond to threats in a future certain to bring change.

Summary

- Field procedures and sampling techniques were developed to rapidly characterize the physical, biological, and chemical structure of coastal rock pools.

- Bedrock geology and shoreline topography differs among park units. The sloping basalts of ISRO produce many reaches of shoreline with abundant rock pool habitat well differentiated into lichen and splash pool zones. Less rock pool habitat, narrower rock pool zones, and weaker zonation of rock pool types are characteristics of shorelines at APIS and PIRO. These characteristics appear to translate into biological and chemical differences between zones at ISRO, with less differentiation at APIS and PIRO.

- Rock pools are subject to some of the same stresses as larger nearby systems, such as the presence of invasive species found in Lake Superior. However, rock pool systems are also subject to stresses and ecological processes not impacting nearby inland lake systems, such as wave scouring in storms, periodic desiccation and re-wetting, and potential impacts such as oil or chemical spills that could influence entire collections of pools on individual islands. Rock pools are likely to be more sensitive to climate change due to their small size.

- Chironomidae diversity at ISRO was similar to that found in multi-lake or large river studies; this diversity is surprising given the limited substrate, nutrients, and likely limited niche availability. Chironomid communities at ISRO are dominated by two subgenera, *Orthocladius* (*Eudactylocladius*) and *Psectrocladius* (*Psectrocladius*), which were nearly as abundant as all other genera combined.

- Chironomidae richness at APIS was low and the fauna was similar to PIRO. Communities at PIRO appeared to be influenced by stream and groundwater inputs.

- For Chironomidae there was no support for community differences between permanent and ephemeral pools, and overall these strata were statistically similar in all three parks. Nearly all chironomids stratified completely or partially based on differences between lichen and splash zones.

- Zooplankton communities in the rock pools were unique and varied. Zooplankton species diversity was an order of magnitude higher than that found in inland lakes at ISRO, and dominant species were not always those most expected from Lake Superior or nearby inland lakes.

- Permanent splash zone pools supported unique zooplankton assemblages primarily driven by copepod diversity. Ephemeral splash pools were the least diverse and supported the fewest organisms. Lichen zone pools, particularly permanent ones, supported unique zooplankton assemblages of cladocerans and rotifers.

- At local scales (particular rock pool systems) there was higher zooplankton abundance and diversity in lichen zones and in permanent pools. Communities changed over time in composition and abundance with a peak in diversity and abundance in late summer driven by increases in copepod and cladoceran species.

- Zooplankton taxa included desirable food sources for larger invertebrates and vertebrates.

- Zooplankton taxa included cold-water stenotherms and other potential environmental indicators, but community composition as a whole is the best way to track environmental change.

- Diatom communities in rock pools were not as diverse as some Great Lakes samples, but were similar in diversity to inland lake sediment samples previously analyzed from ISRO, PIRO, and APIS. However, the rock pool communities represent unique assemblages that had not been previously sampled or analyzed for diatoms.

- Several generalist diatom taxa were common in all parks and across all pool zones and types (e.g., *Achnanthidium* and *Gomphonema* species). Lichen pool diatom communities were further characterized by a greater abundance of species indicative of low pH, low conductivity, and higher productivity waters; these included chrysophyte cysts, *Nitzschia*, *Encyonema*, *Brachysira*, *Eunotia*, *Pinnularia*, *Tabellaria*, and *Stauroforma* species.

- Splash zone pool communities at ISRO were characterized by epilithic diatoms that are also commonly found in the nearshore zone of Lake Superior including *Denticula*, *Synedra*, *Delicata*, *Cymbella*, and *Eucocconeis* species. Splash pools also contained plankton species found in Lake Superior including *Cyclotella*, *Discostella*, and *Ulnaria* species.

- Diatom communities at APIS and ISRO were not as clearly differentiated among pool zones compared to ISRO.

- Water chemistry and hydrology differed between lichen and splash zone pools at ISRO. Lichen pools were characterized by higher levels of nutrients (TP, TN, SRP), DOC, and chlorophyll, and lower levels of DIC, NO_x, and conductivity compared to splash pools. At APIS, there were weaker differences in chemistry of pool types. At PIRO, water quality measures did not clearly characterize pool zones, and pools were further influenced by groundwater inputs.

- Thermistors deployed in several ISRO pools in 2010 and 2012 collected continuous thermal data that showed lichen zone pools commonly experienced diurnal temperature swings of 15 °C and reached temperatures over 30 °C on many days. In contrast, splash pools did not warm or cool as much as lichen pools on a diurnal basis due to the moderating effect of Lake Superior.

- Mapping at ISRO determined habitat densities that were much greater than expected, particularly at Passage Island. Chorus frogs were the most commonly encountered species, though generally limited in range to Passage Island and barrier islands at the east end of the park. Blue-spotted salamanders were widely distributed along the south shoreline of ISRO.

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Appendix A: Study Site Maps

Study Site Access and Delineation of Sites

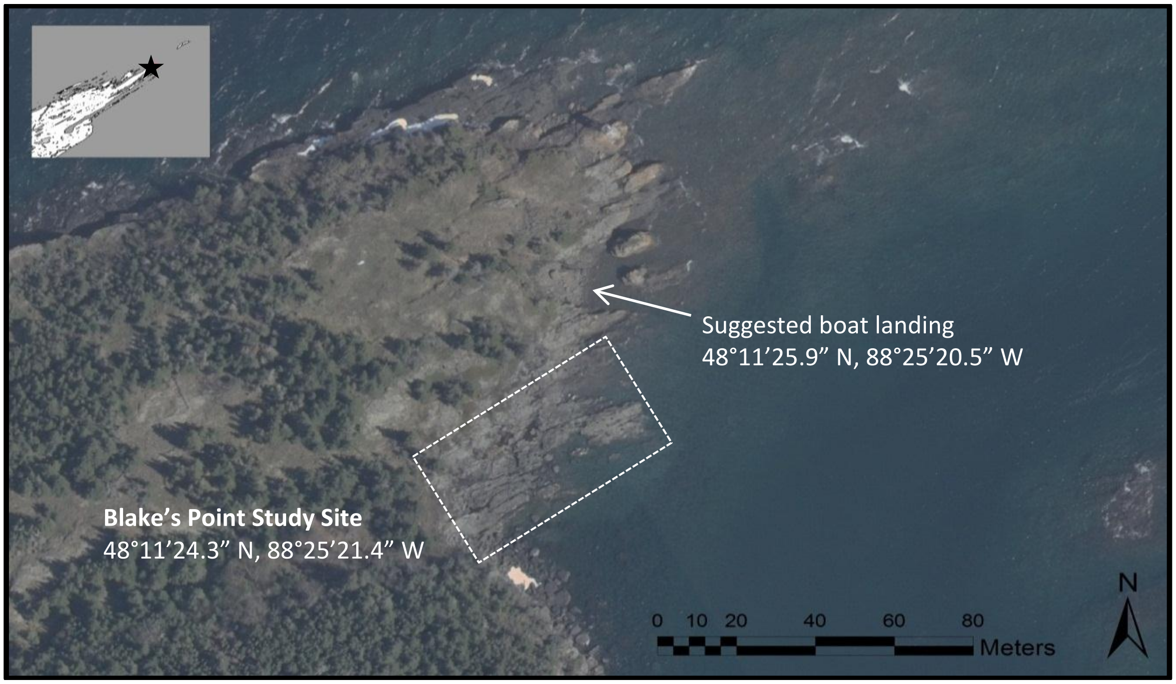


Figure A1. Blake’s Point (BP) sample site overview, Isle Royale. Land in the small cove only in very calm weather. May need to tilt engine and paddle boat into cove, or paddle a canoe from Merritt dock.

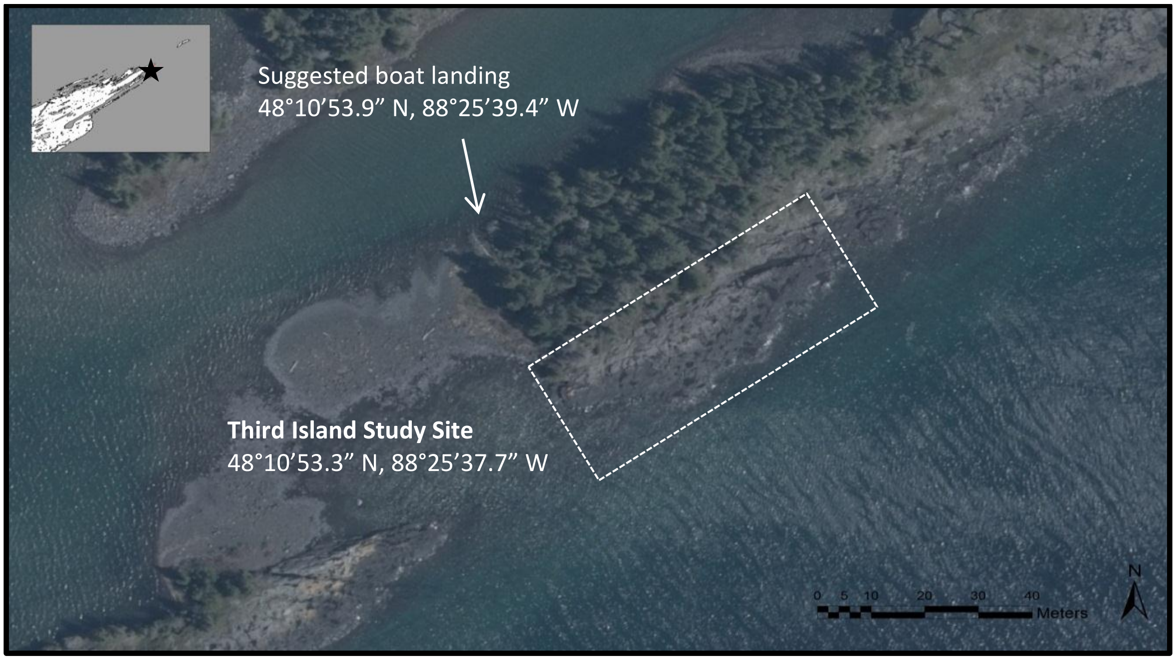


Figure A2. Third Island (TH) sample site overview, Isle Royale. Land on the north side of the island. (Note: the channel north of Third Island is a no-wake-zone).

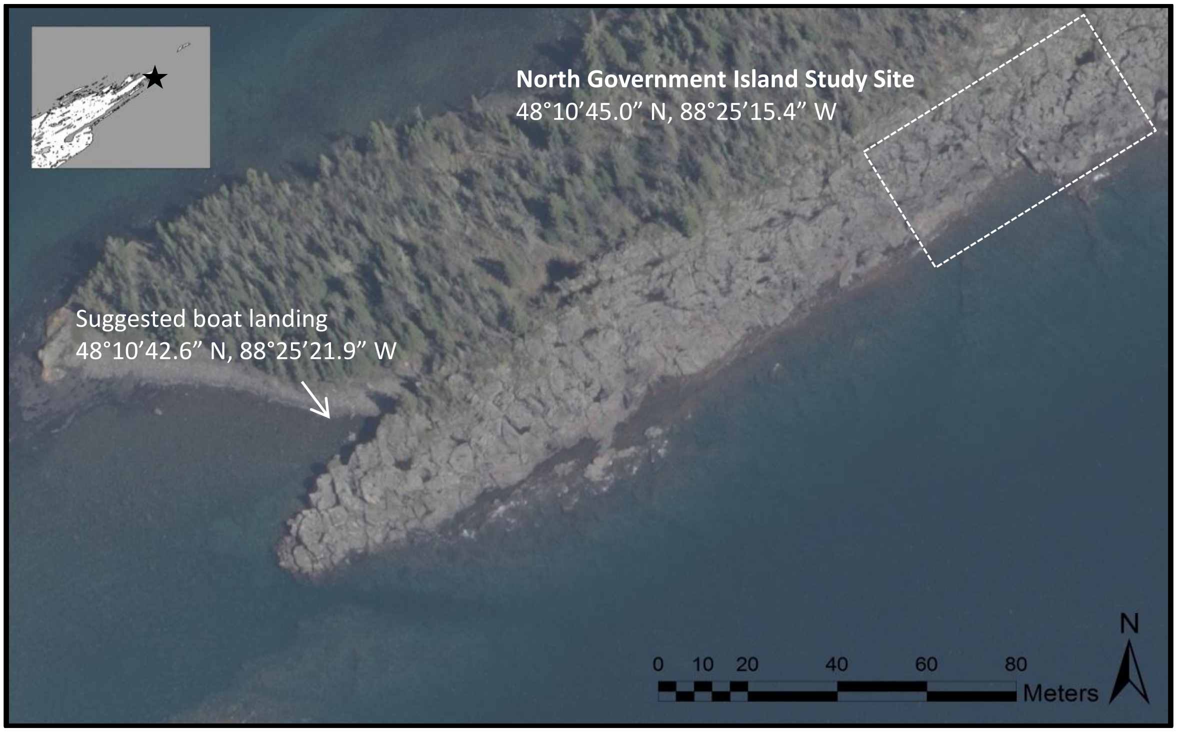


Figure A3. North Government Island (NG) sample site overview, Isle Royale. Land in the shallow slot at the west end of the island. May need to tilt engine and paddle to shore if seiche is out.

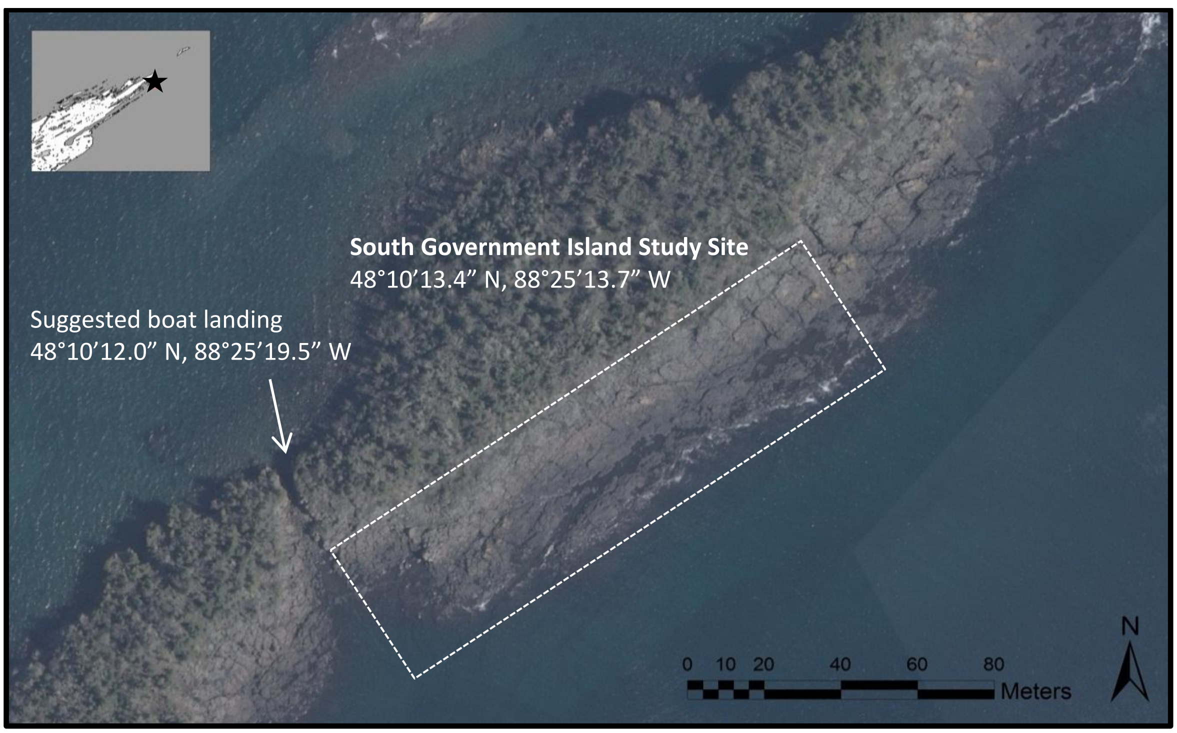


Figure A4. South Government Island (SG) sample site overview, Isle Royale. Land on the north side at the large cleft in the island.

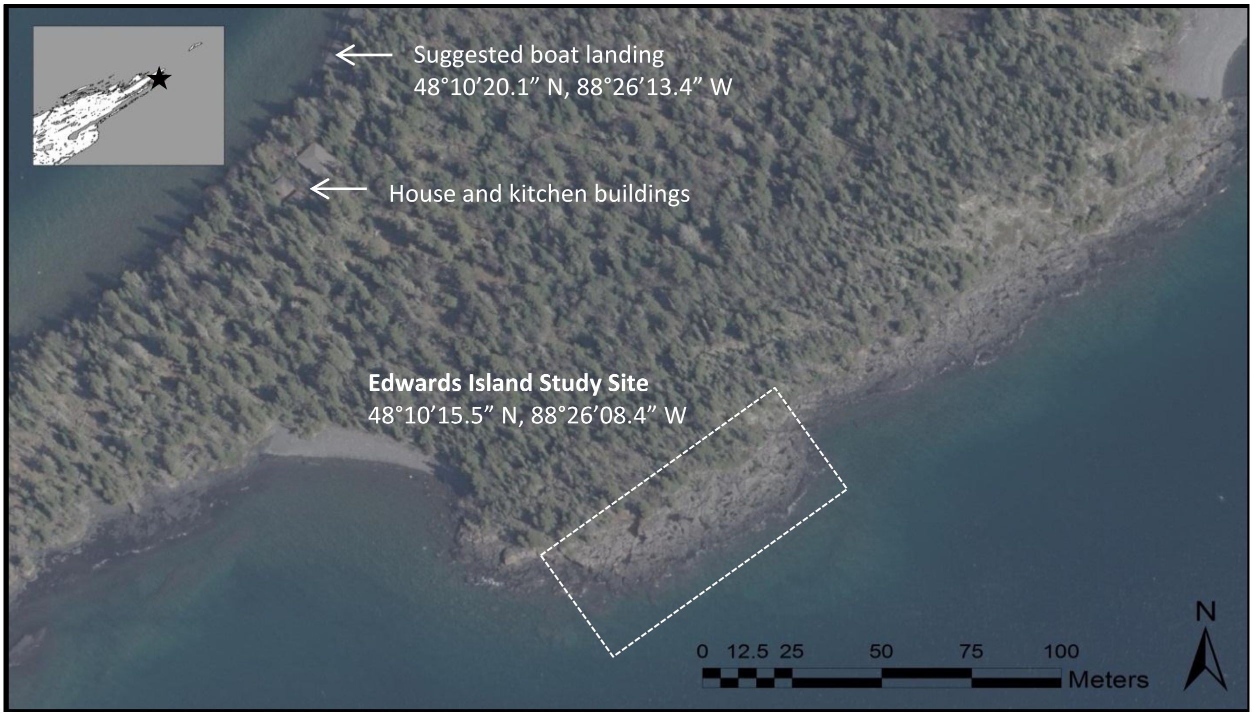


Figure A5. Edwards Island (ED) sample site overview, Isle Royale. Land on the beach near cabin on north side of island and follow trail behind kitchen building to the beach on south side.

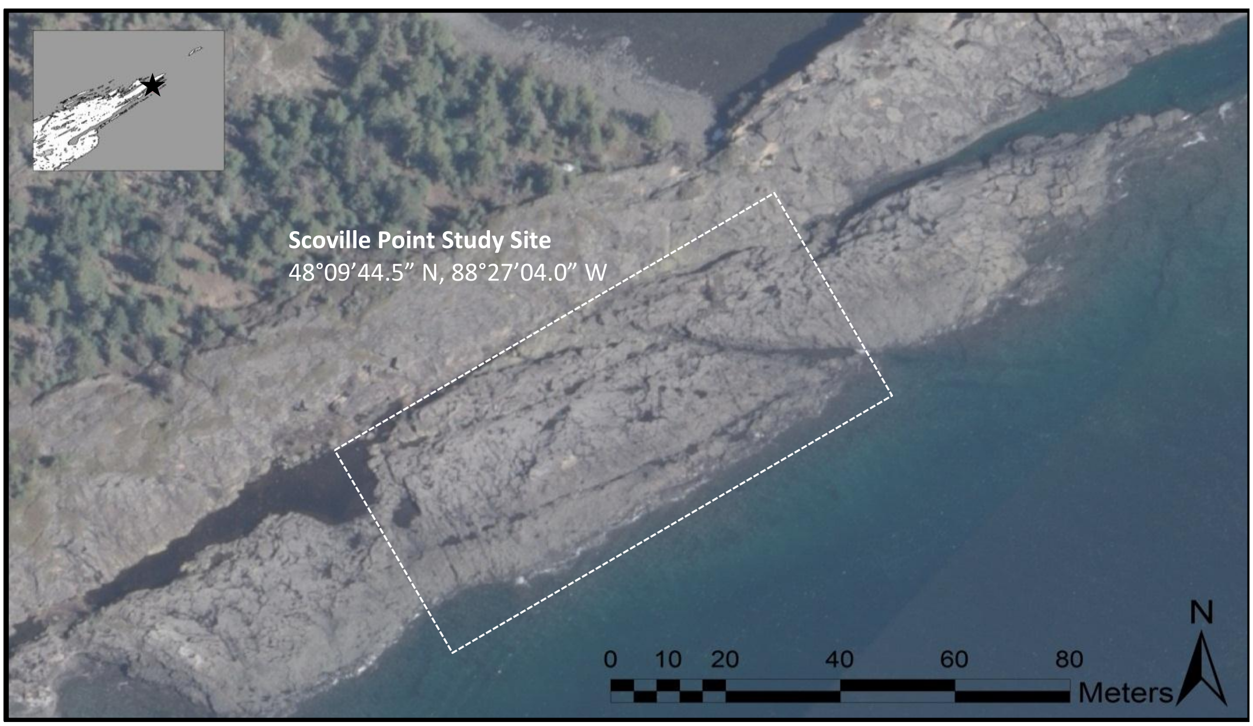


Figure A6. Scoville Point (SP) sample site overview, Isle Royale. Land at the first dock in Tobin Harbor and follow trail to the point.

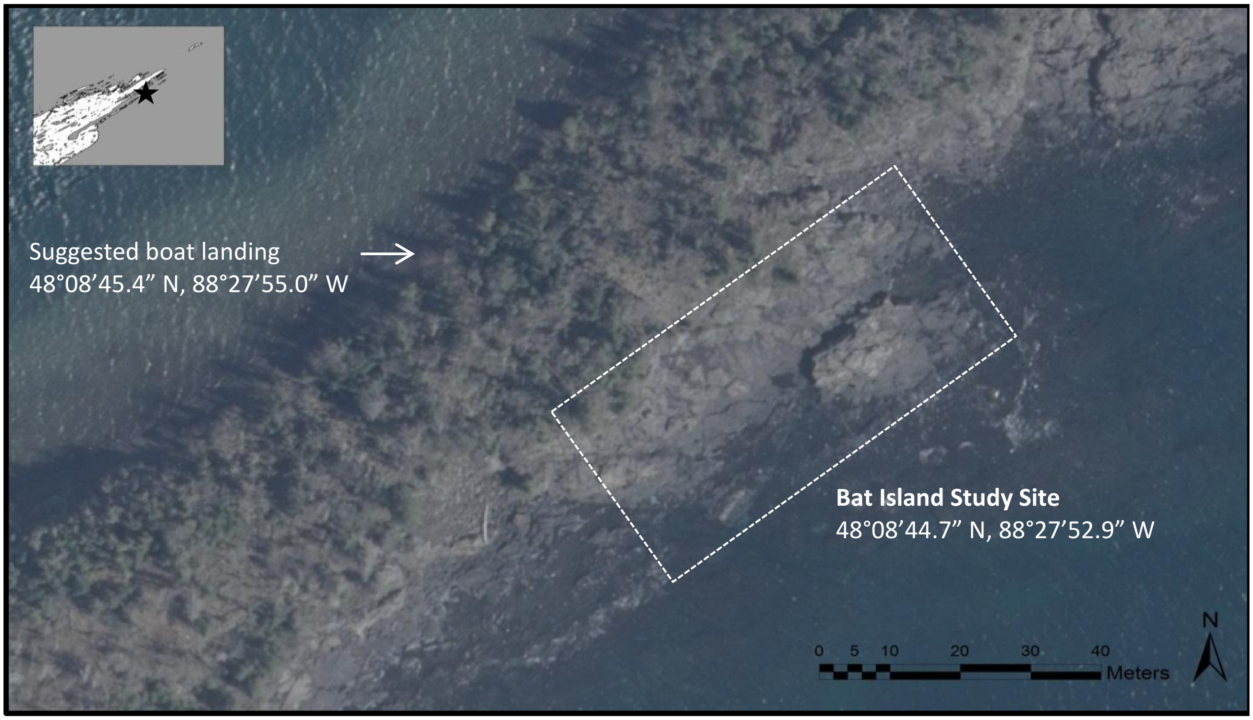


Figure A7. Bat Island (SP) sample site overview, Isle Royale. Land on the north side of island, southwest of the small islet, and hike across to the site.

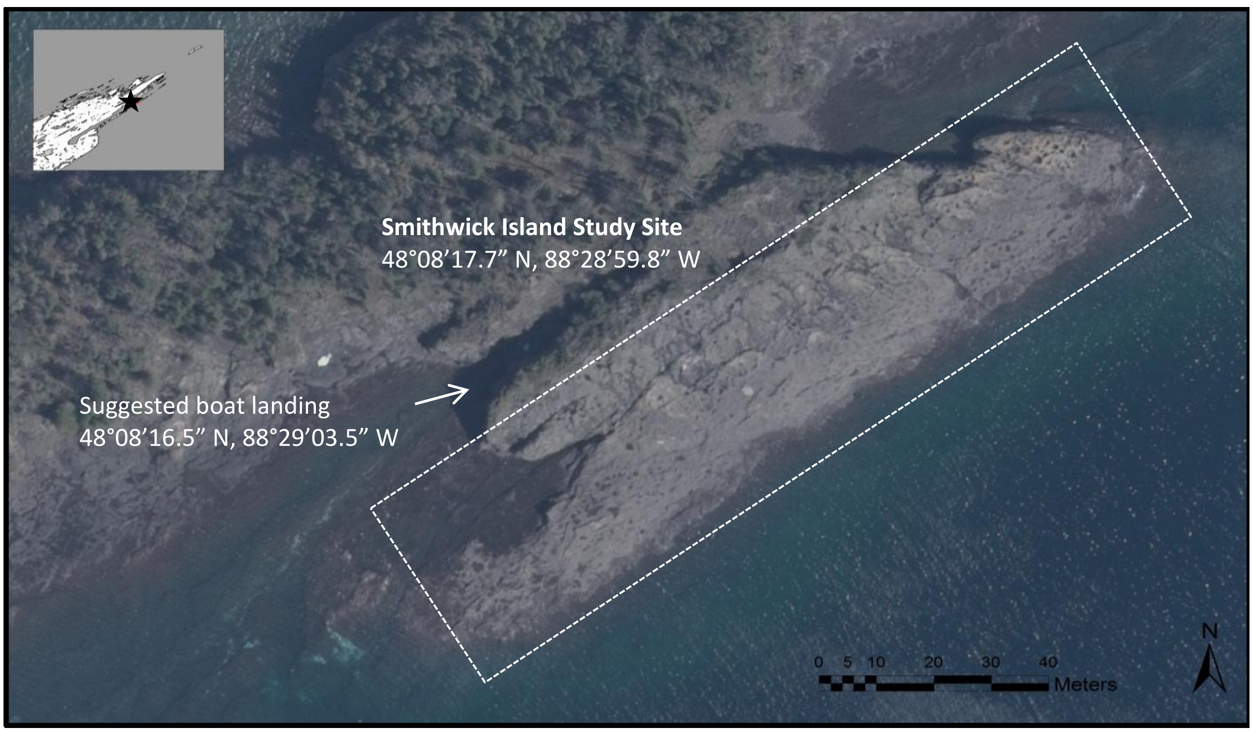


Figure A8. Smithwick Island (SM) sample site overview, Isle Royale. Land in the slot southwest of Smithwick Gap, or on north side and hike over. May need to tilt engine and paddle if seiche is out.

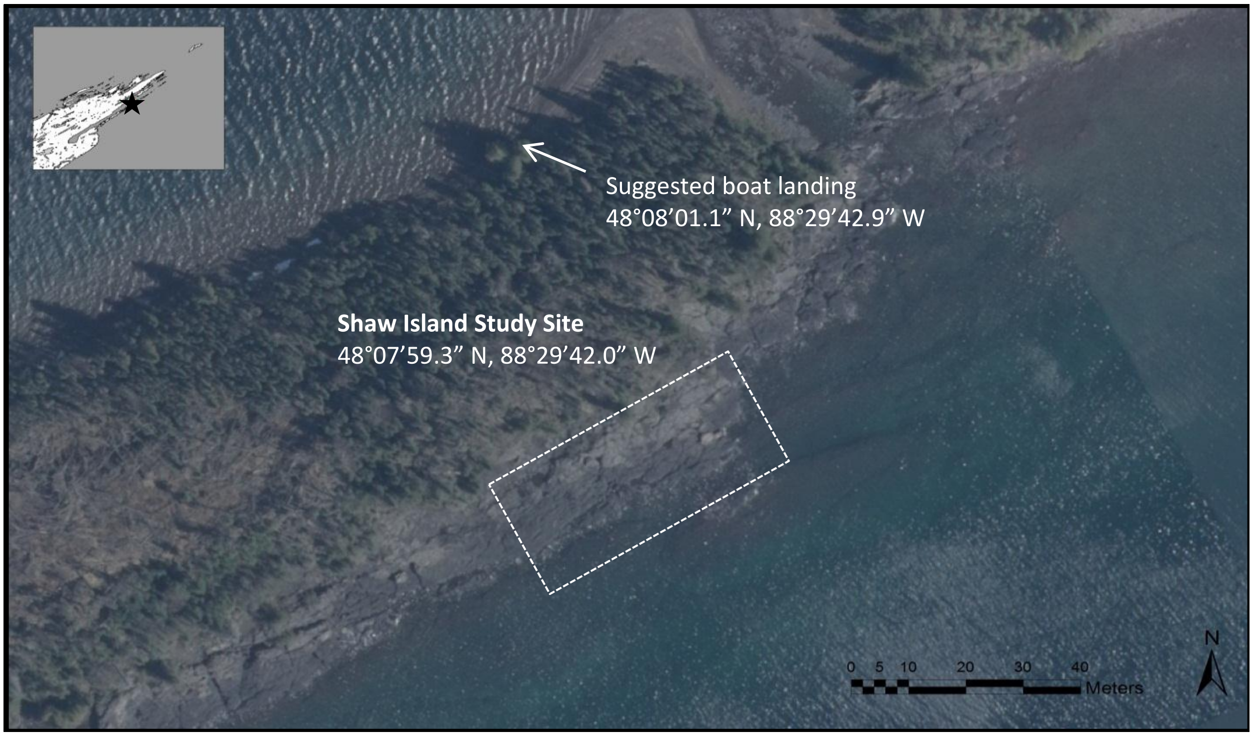


Figure A9. Shaw Island (SH) sample site overview, Isle Royale. Land on north side of island near Smithwick-Shaw gap and hike around on shoreline.

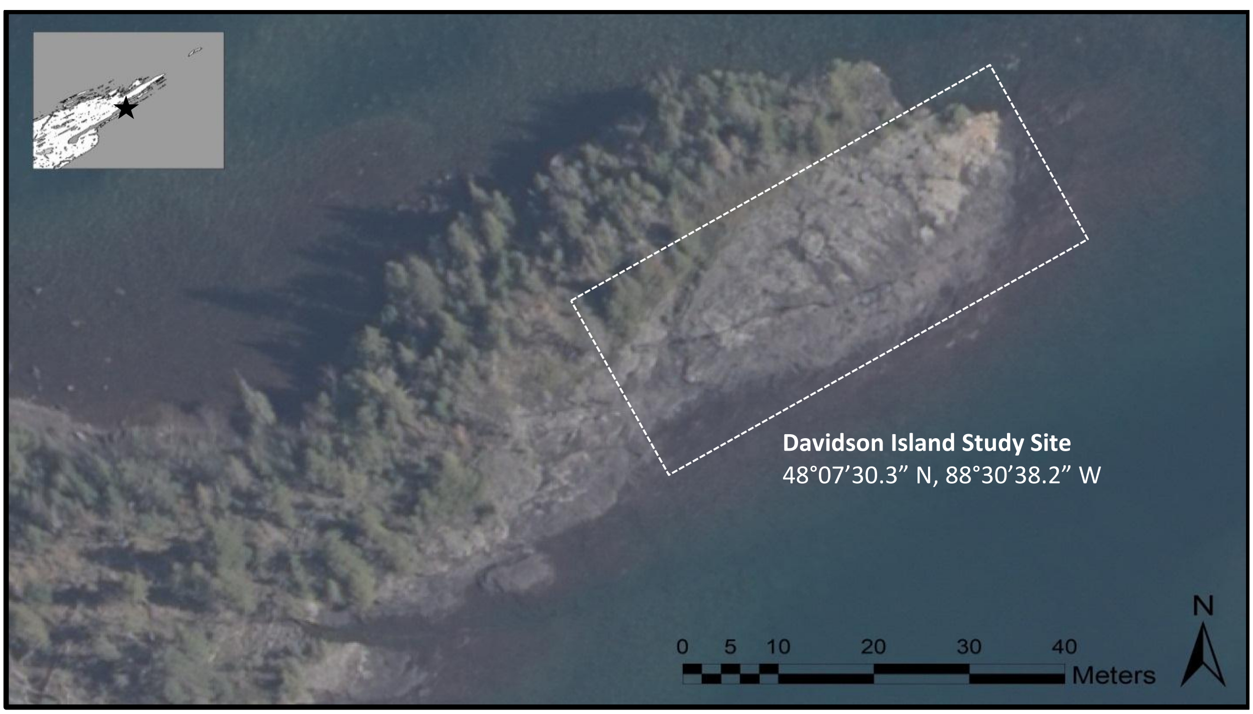


Figure A10. Davidson Island (DA) sample site overview, Isle Royale. Follow trail from Boreal Research Station to the east end of island.

Figure A11. Outer Hill Island (OH) sample site overview, Isle Royale. Land in Lorelei Lane in small cove. (Note: Lorelei Lane is a no-wake-zone; also, avoid disturbing loons that may be nesting nearby.)

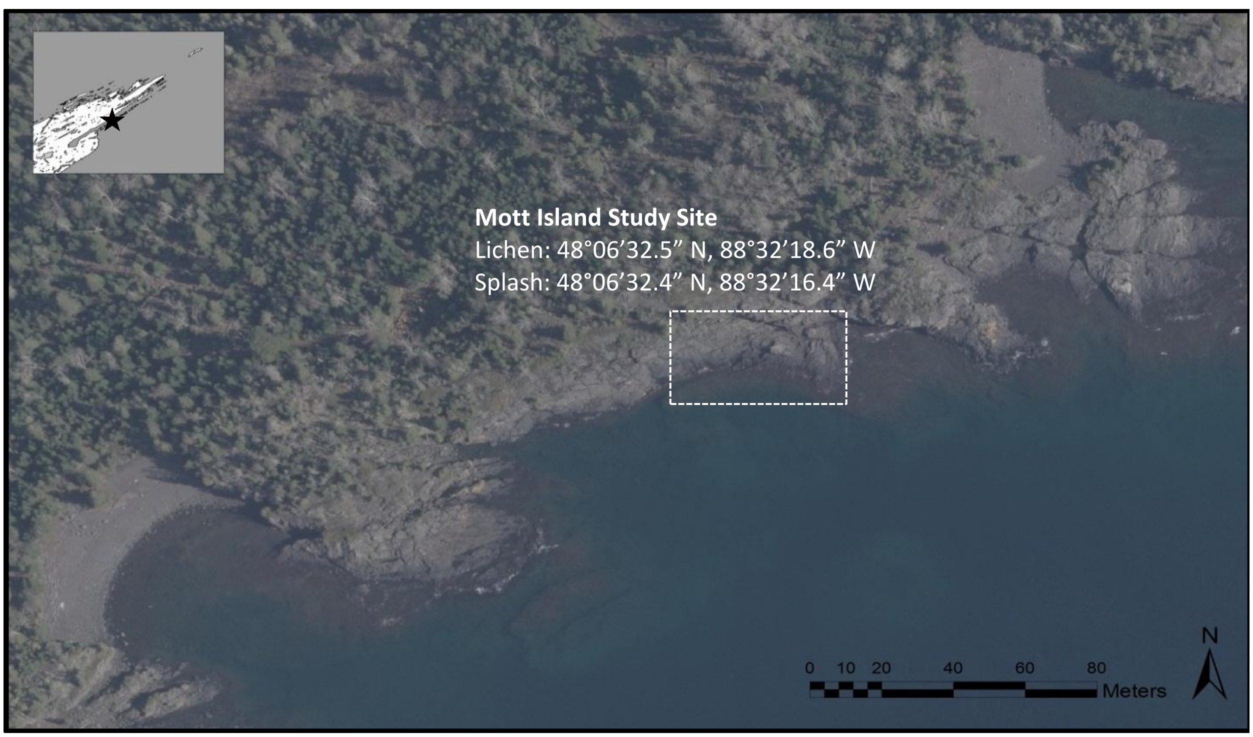


Figure A12. Mott Island (MO) sample site overview, Isle Royale. Follow lake-side trail from developed area. After large crescent-shaped beach continue to follow trail until it briefly exits onto bedrock.

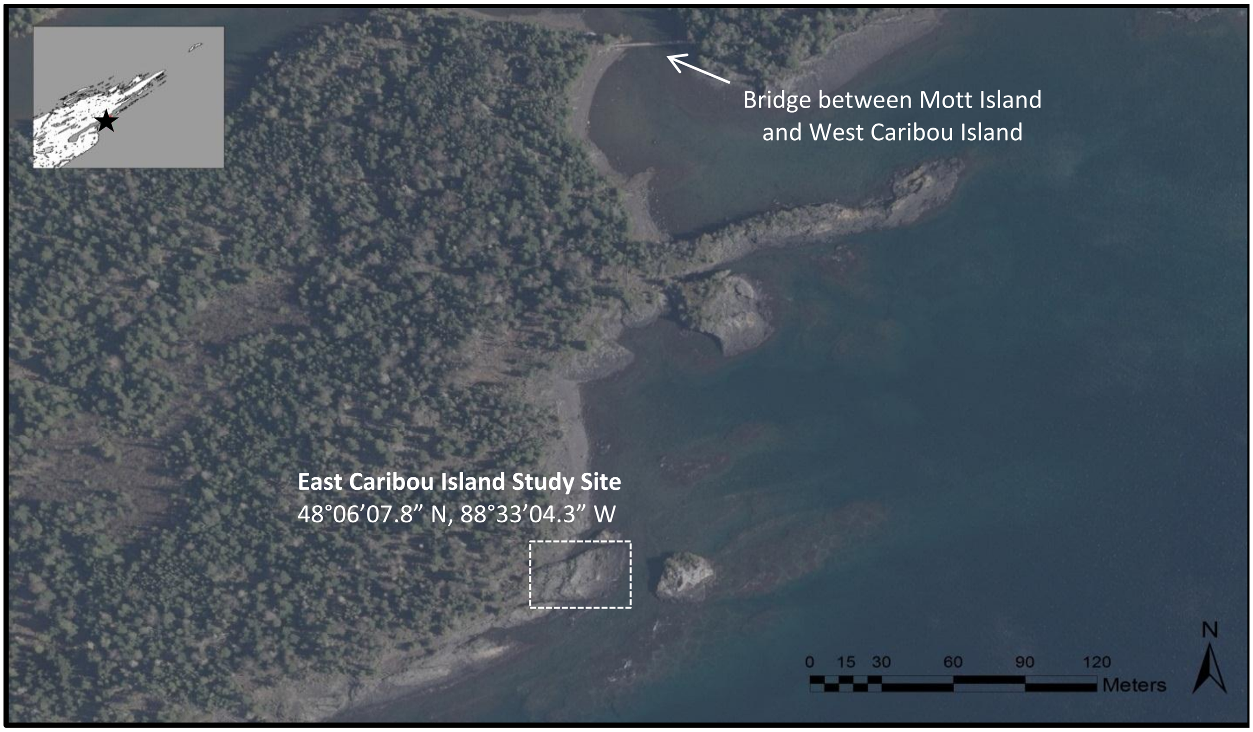


Figure A13. East Caribou Island (EC) sample site overview, Isle Royale. Follow trail across bridge between Mott and East Caribou Islands, then hike beaches south to the site.

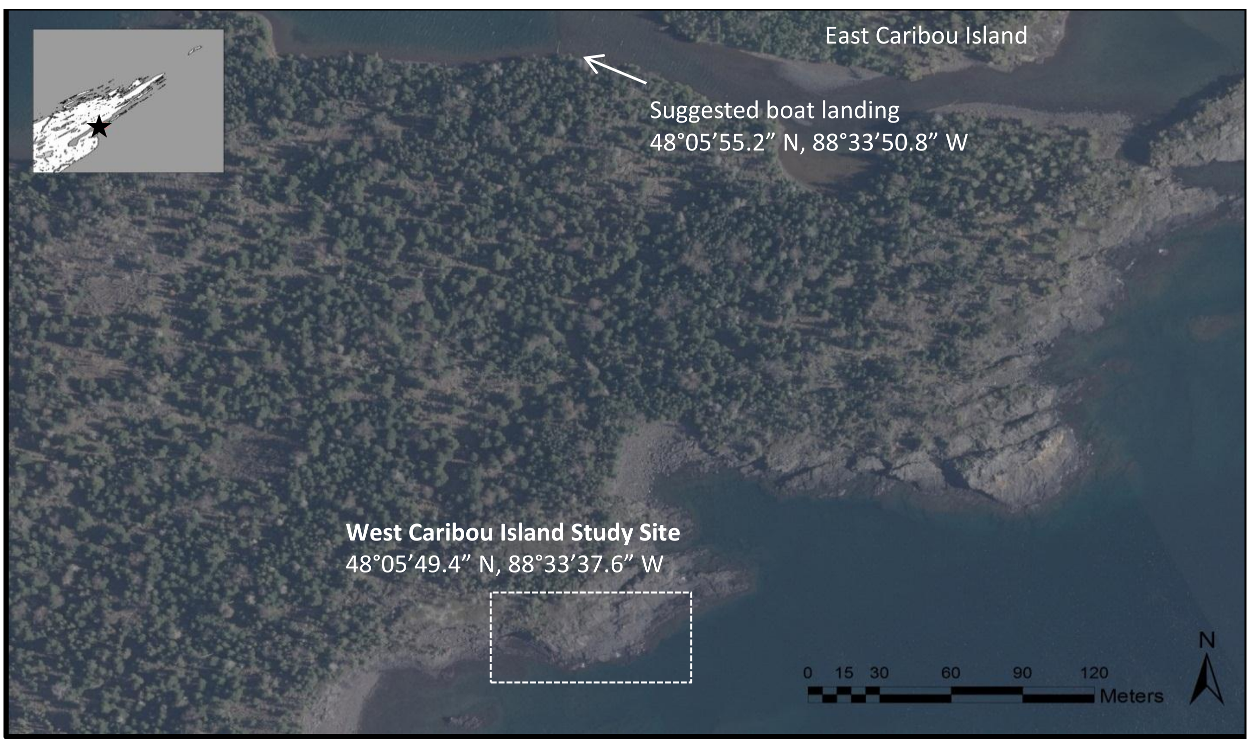


Figure A14. West Caribou Island (WC) sample site overview, Isle Royale. Land between East Caribou, West Caribou, and Rabbit Islands, then follow compass south to site. Large arch marks west end of site.

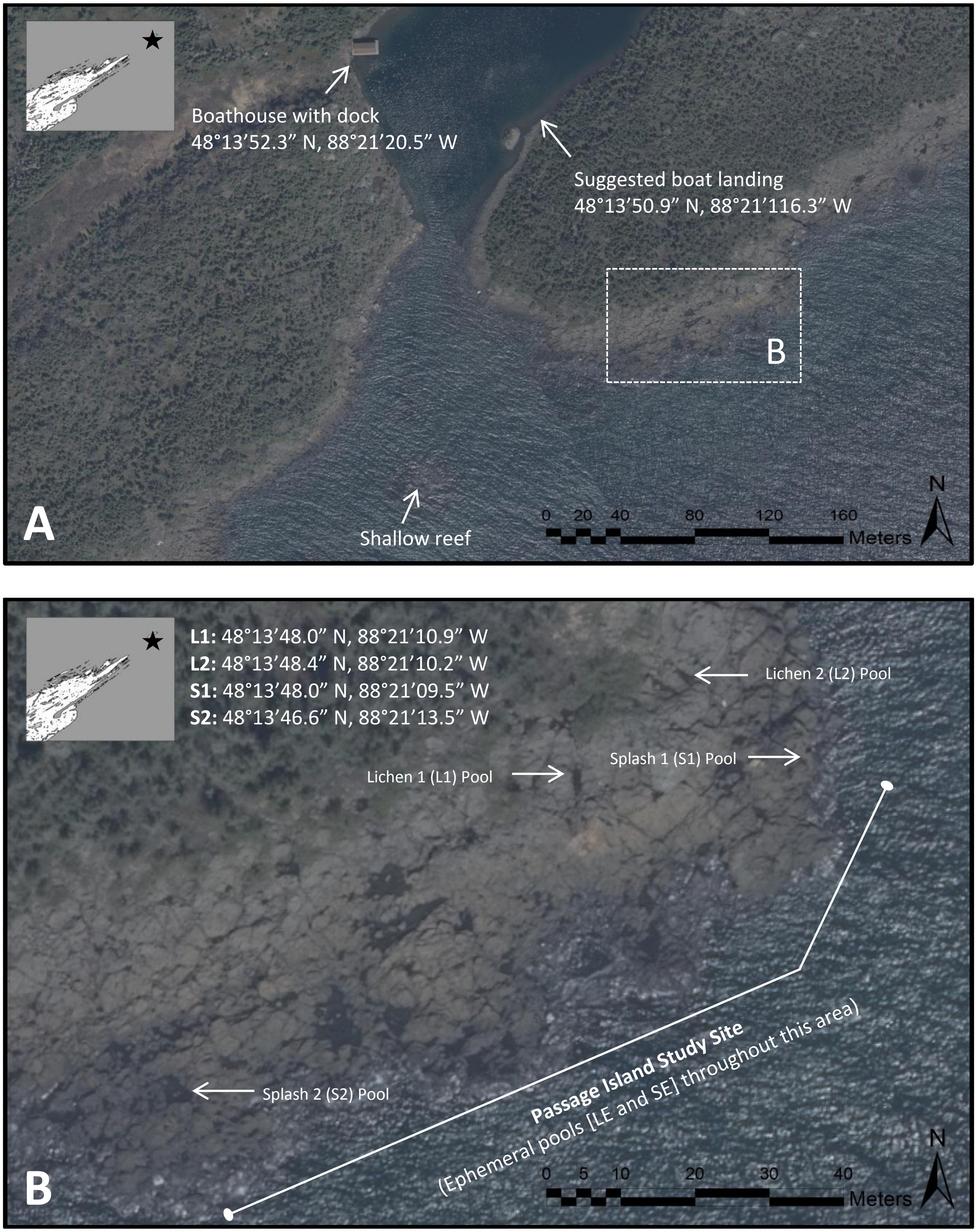
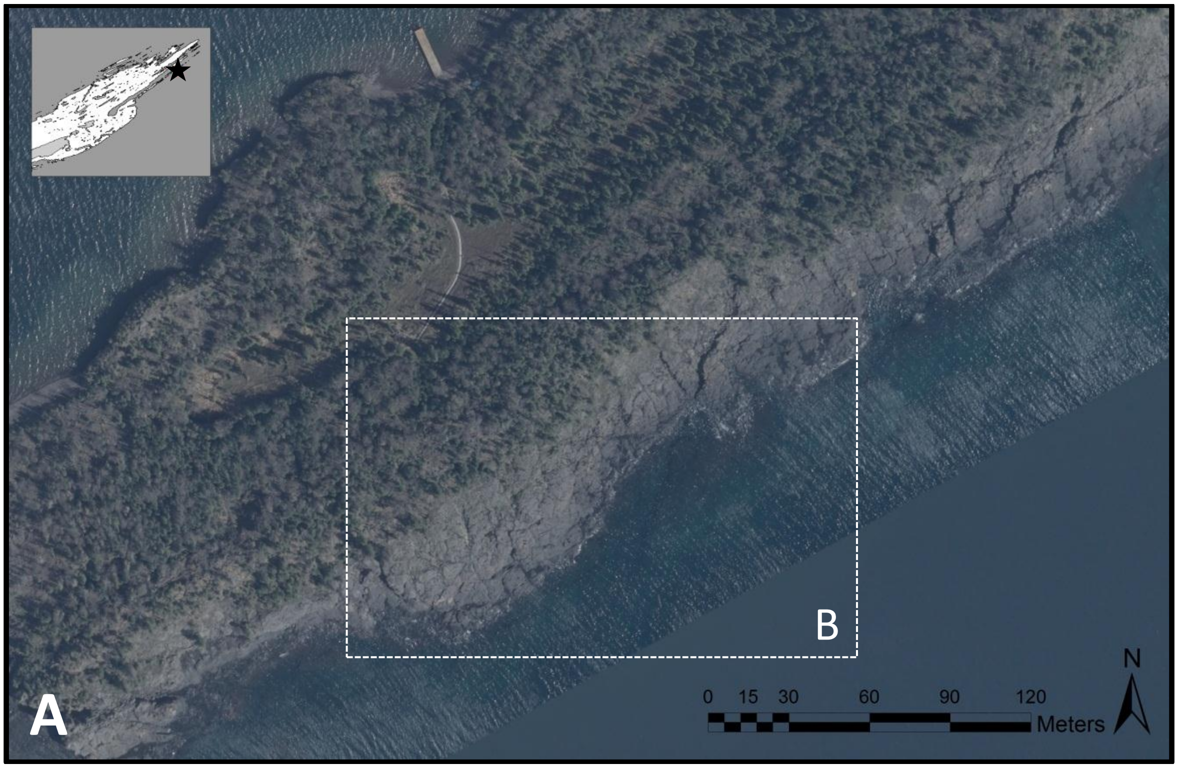


Figure A15. Passage Island (PA) sample site overview (A) and detail (B), Isle Royale. Land behind the small islet in the cove and hike to the site, or land at the boathouse and paddle across cove. When entering the cove, slow down and watch for the very shallow reef.



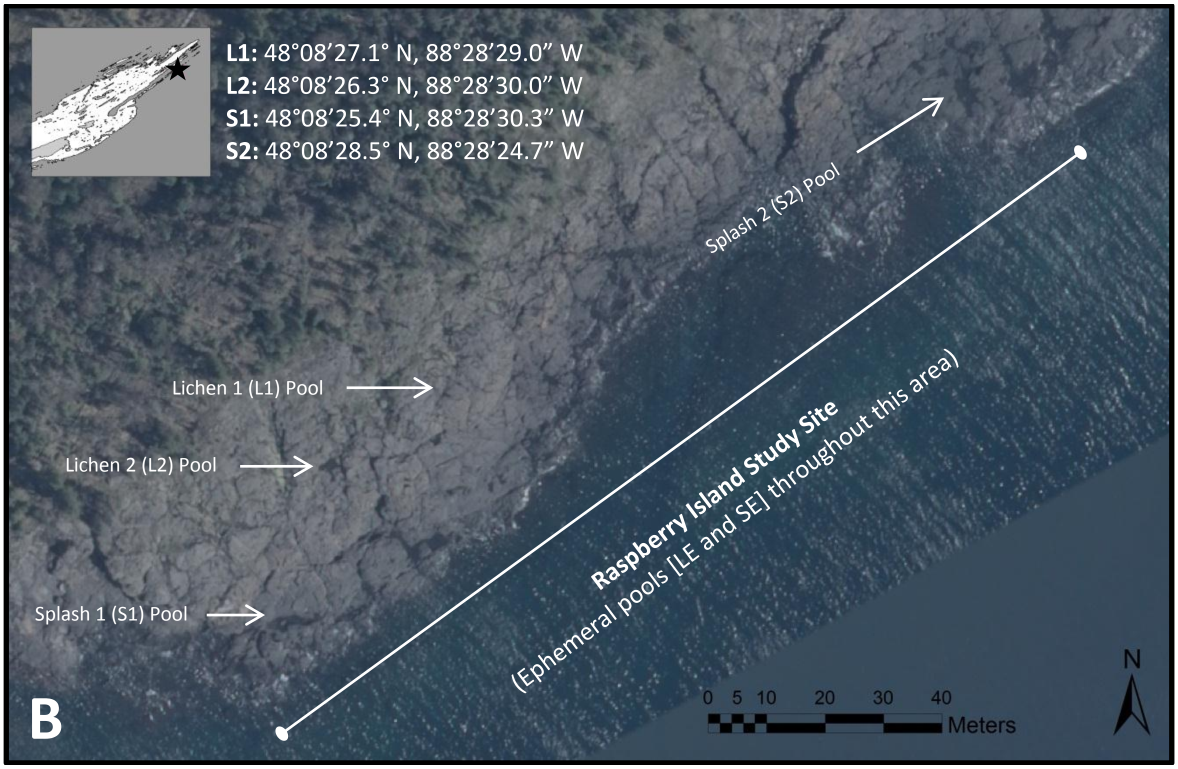
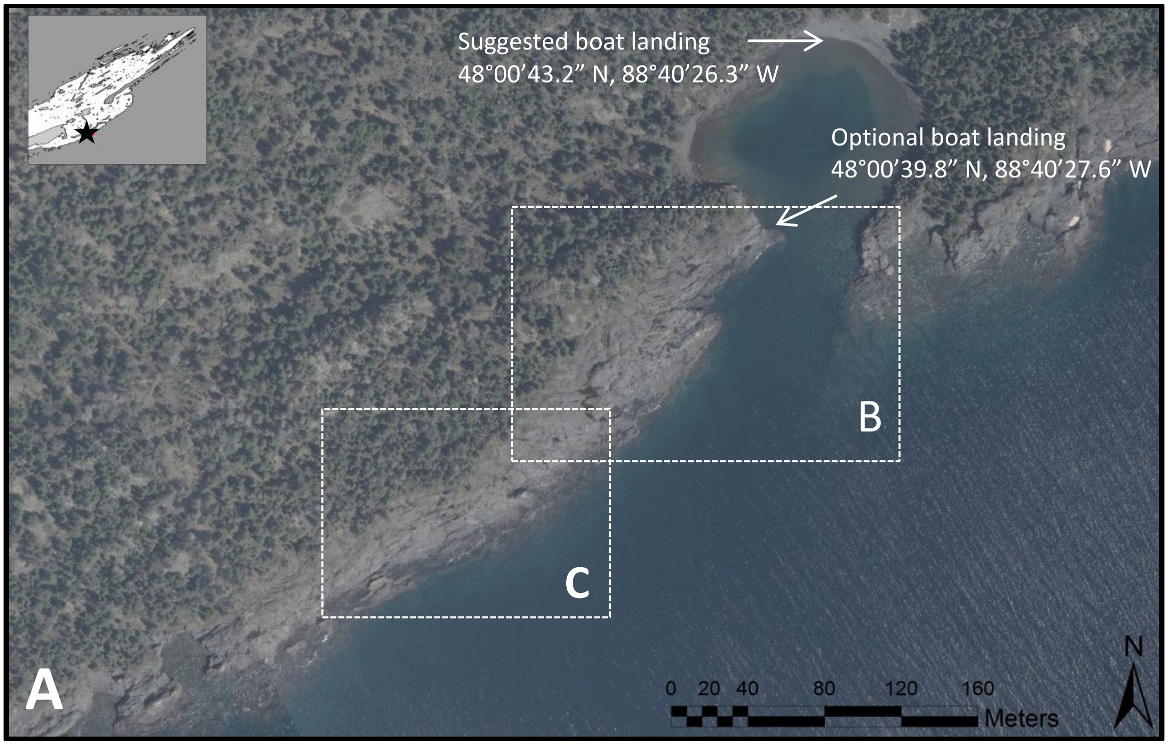


Figure A16. Raspberry Island (RA and RS) sample site overview (A) and detail (B), Isle Royale. From the dock on the north side of the island, cross the boardwalk to the short, steep section of trail leading to the shore.



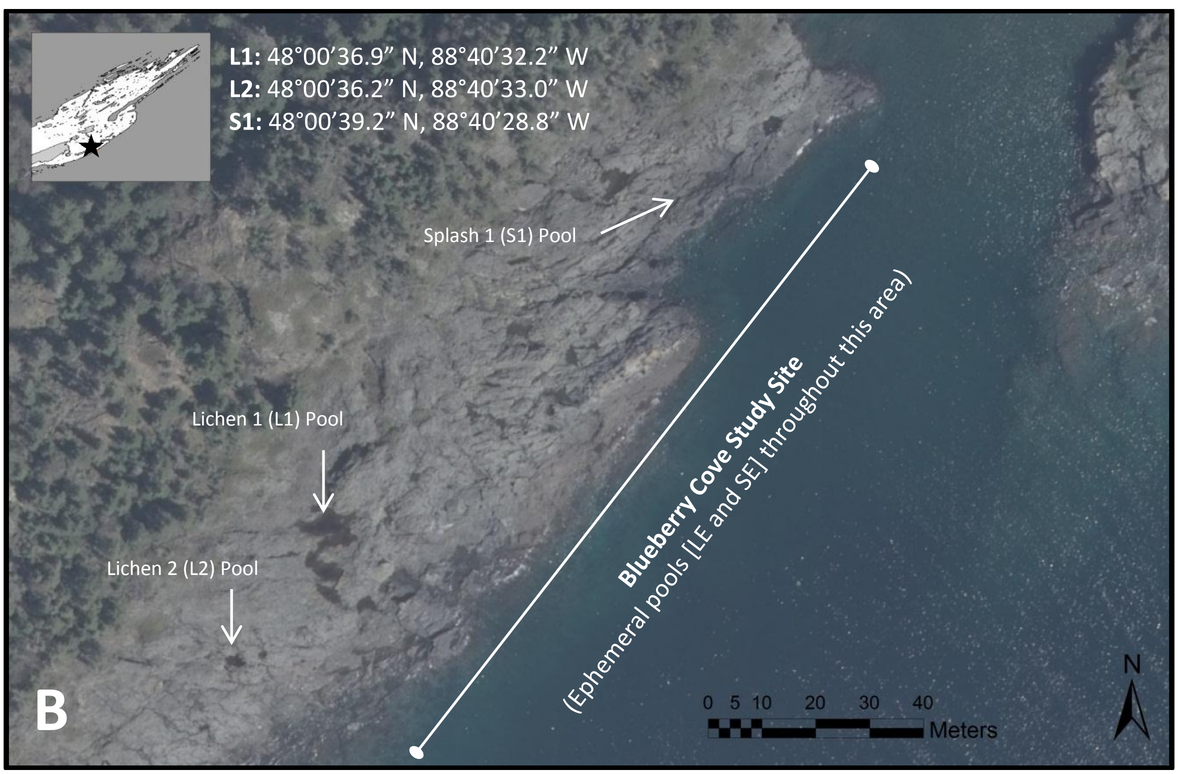


Figure A17. Blueberry Cove (BL) sample site overview (A) and detail (B and C), Isle Royale. Land in the back of the cove on gravel beach, then hike over steep ridge behind the site. If calm, landing can be made on southwest side of cove against bedrock.

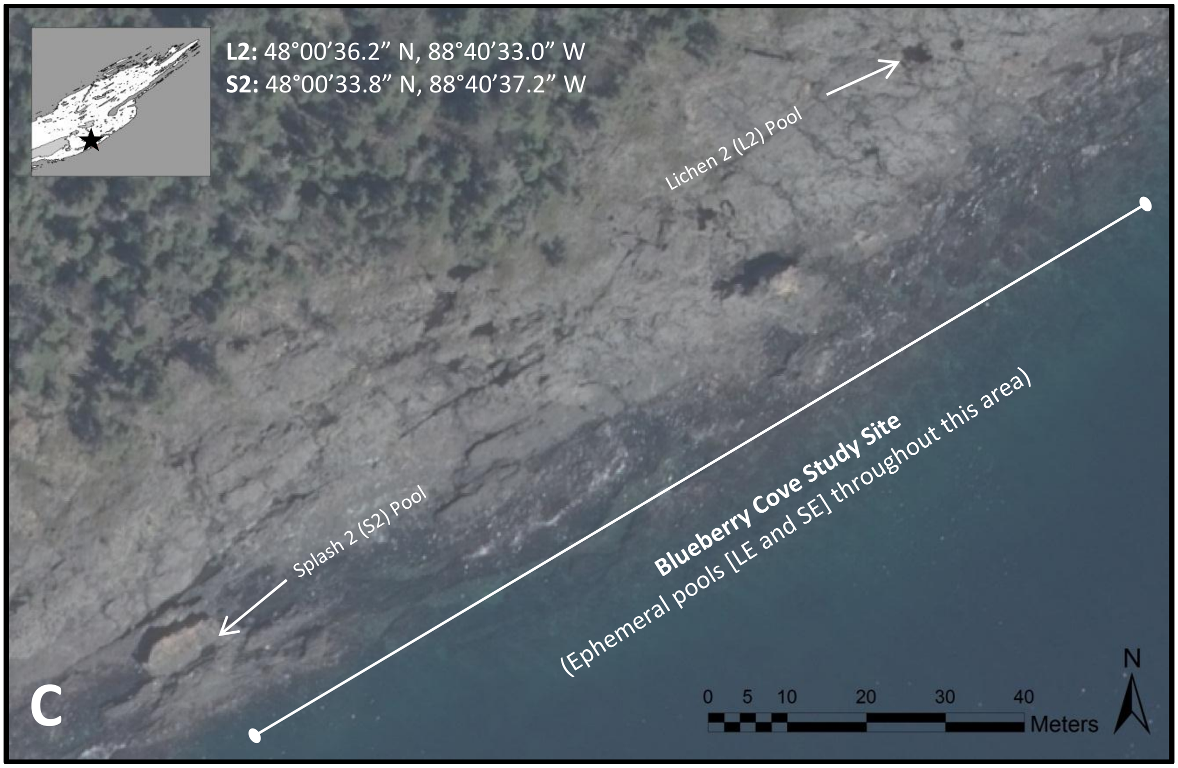


Figure A17. Blueberry Cove (BL) sample site overview (A) and detail (B and C), Isle Royale. Land in the back of the cove on gravel beach, then hike over steep ridge behind the site. If calm, landing can be made on southwest side of cove against bedrock (continued).

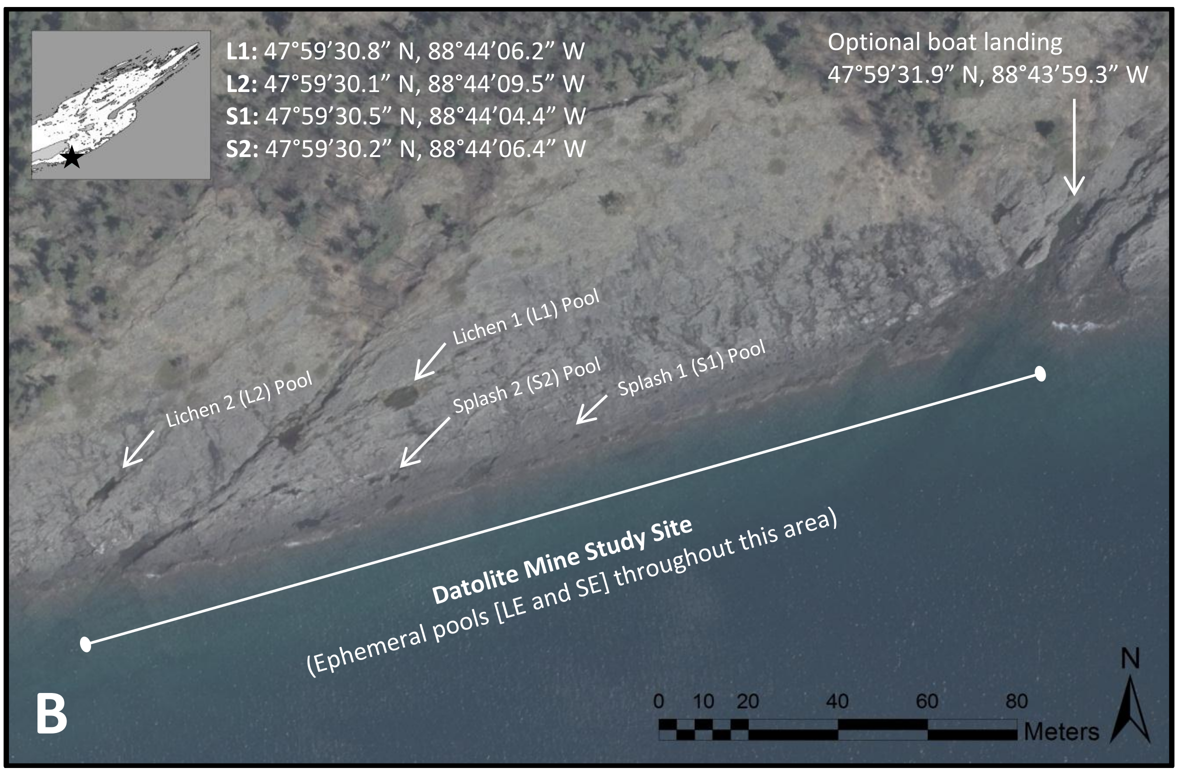


Figure A18. Datolite Mine (DM) sample site overview (A) and detail (B), Isle Royale. Optional landing in slot of bedrock near the study site works only in very calm conditions with a long anchor line; Little Greenstone Beach is a preferable landing site. Hiking along the shore of the study area is easy, but head inland to avoid steep cliffs near Little Greenstone Beach.

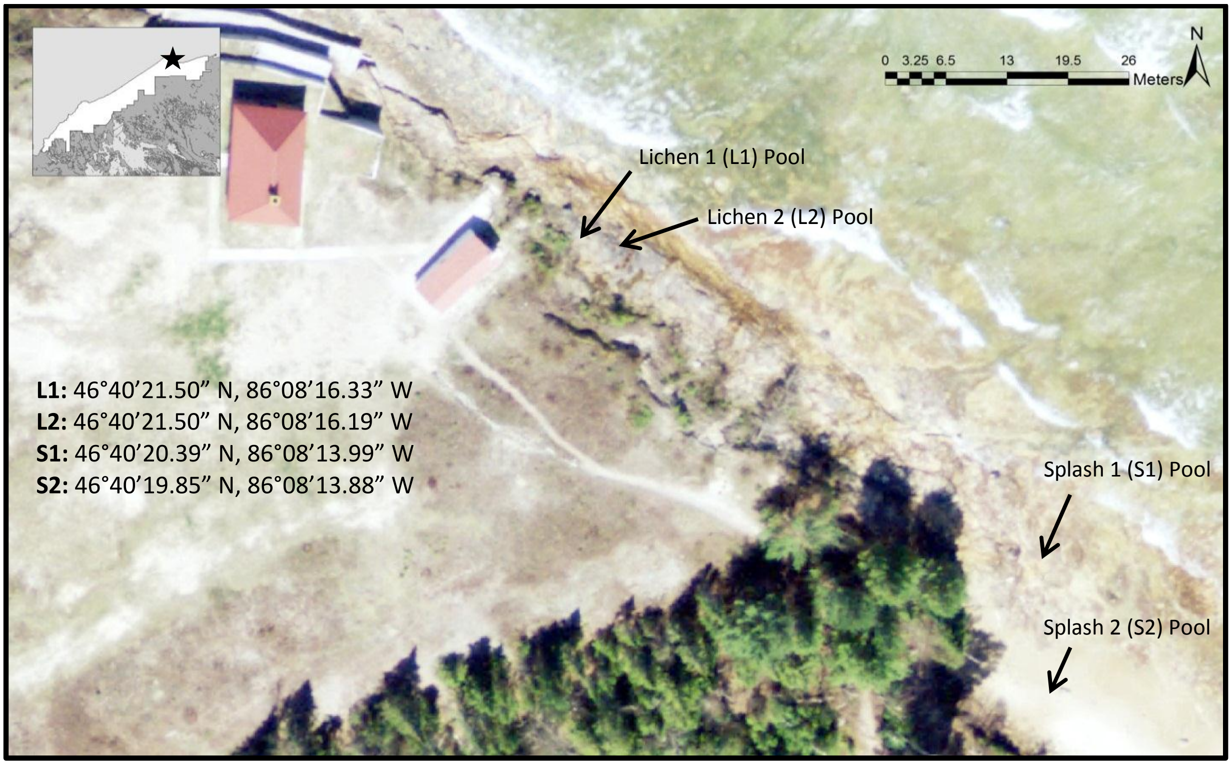


Figure A19. Au Sable Point (AU) sample site, Pictured Rocks.

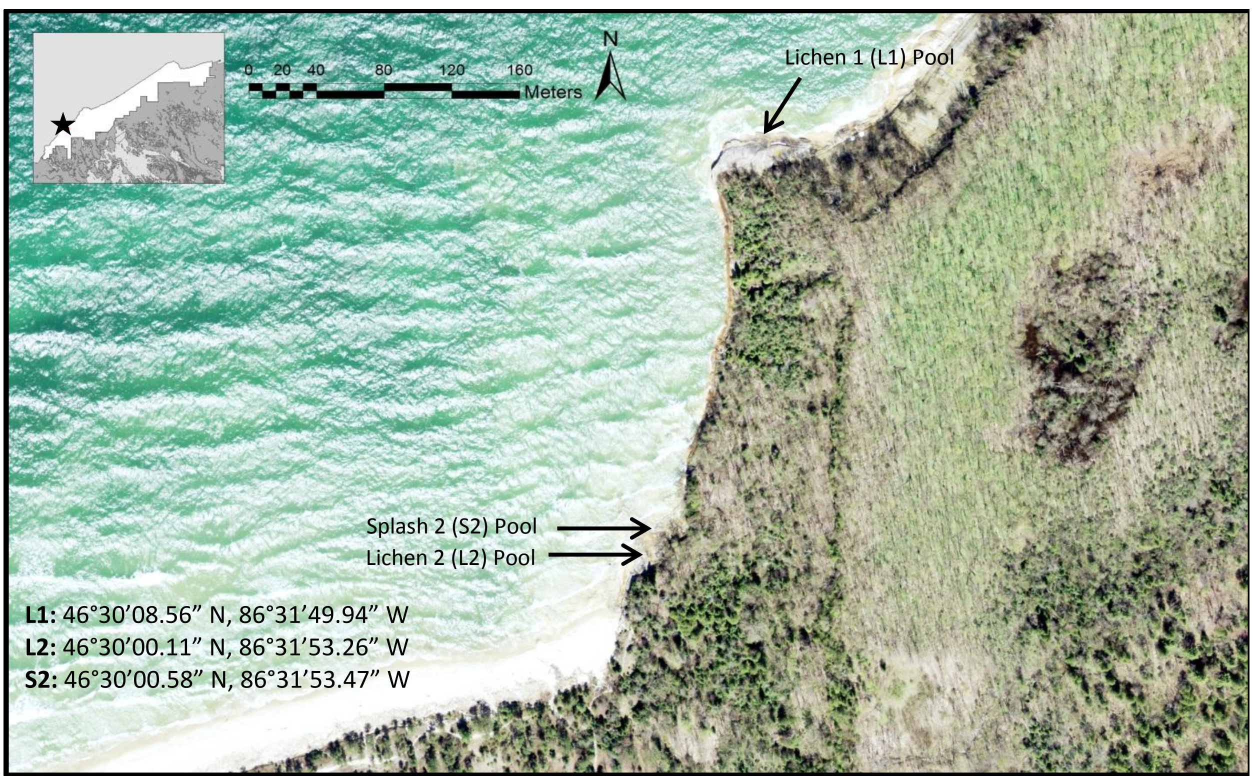


Figure A20. Miner's Beach (MB) sample site, Pictured Rocks.

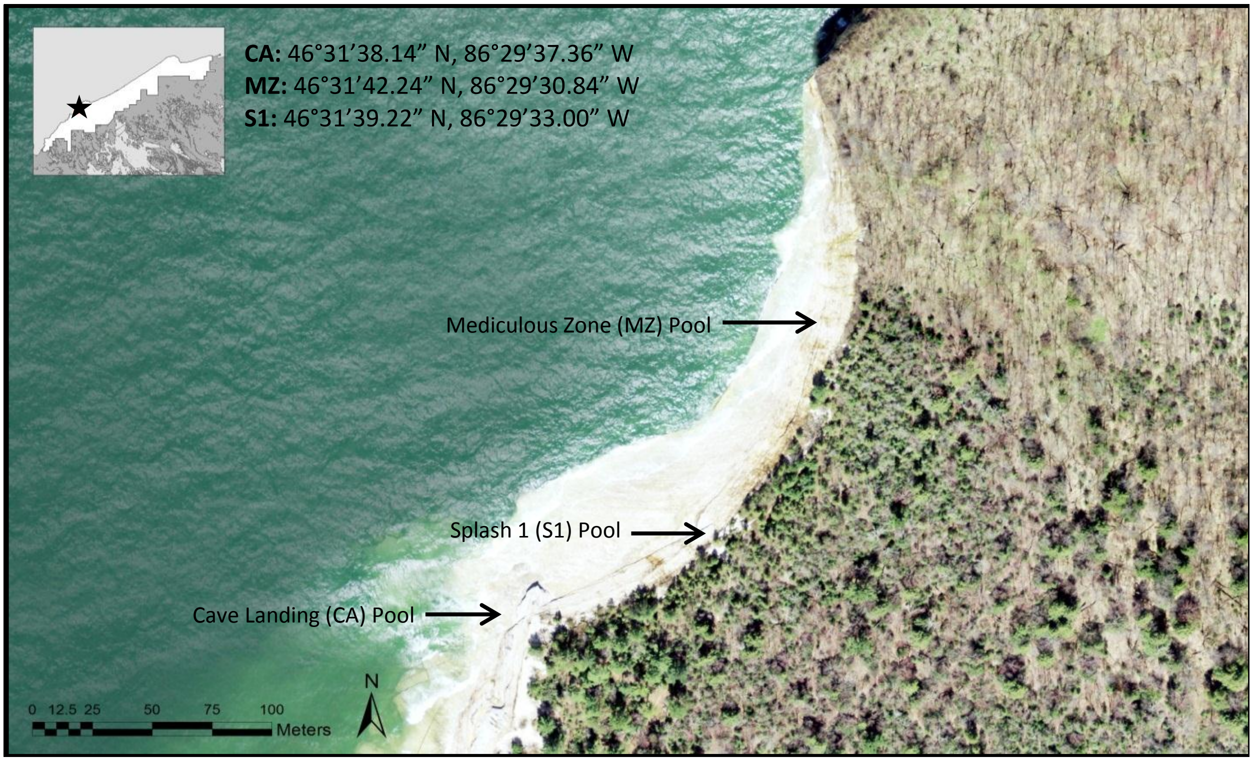


Figure A21. Mosquito Harbor (MH) sample site, Pictured Rocks.

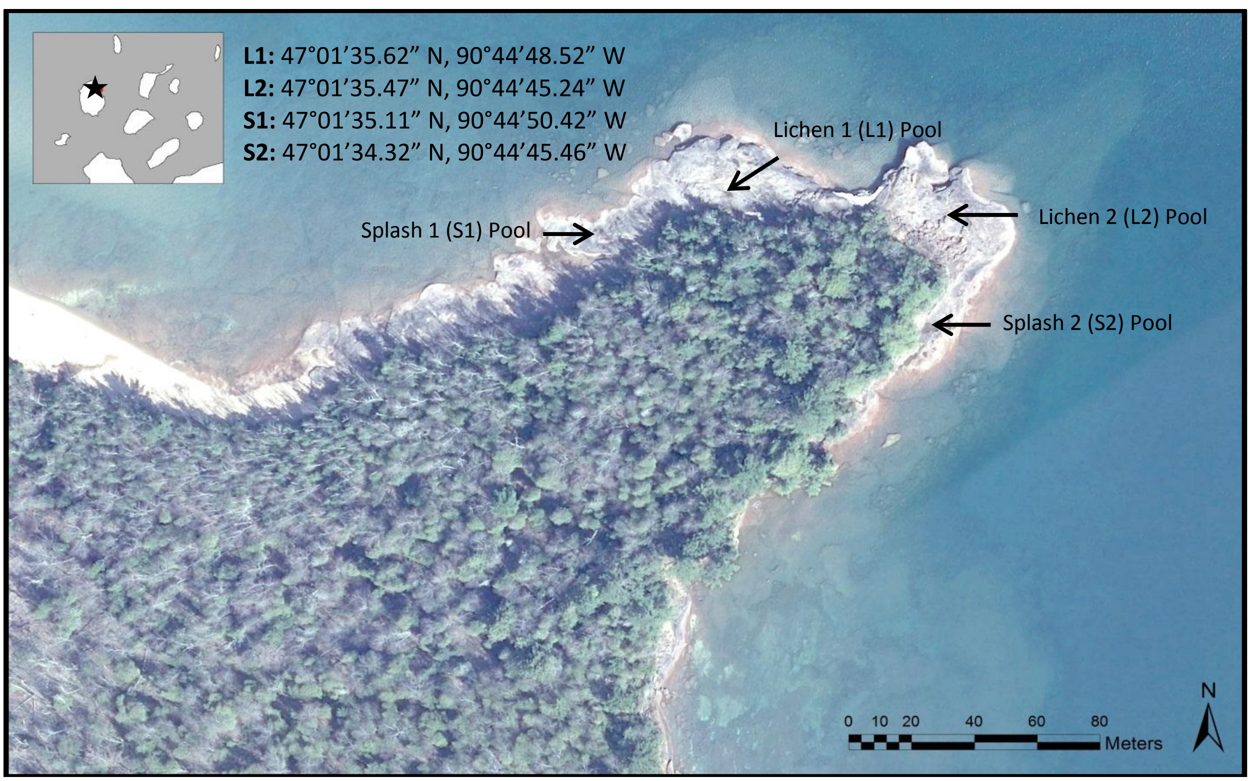


Figure A22. Bear Island (BI) sample site, Apostle Islands.

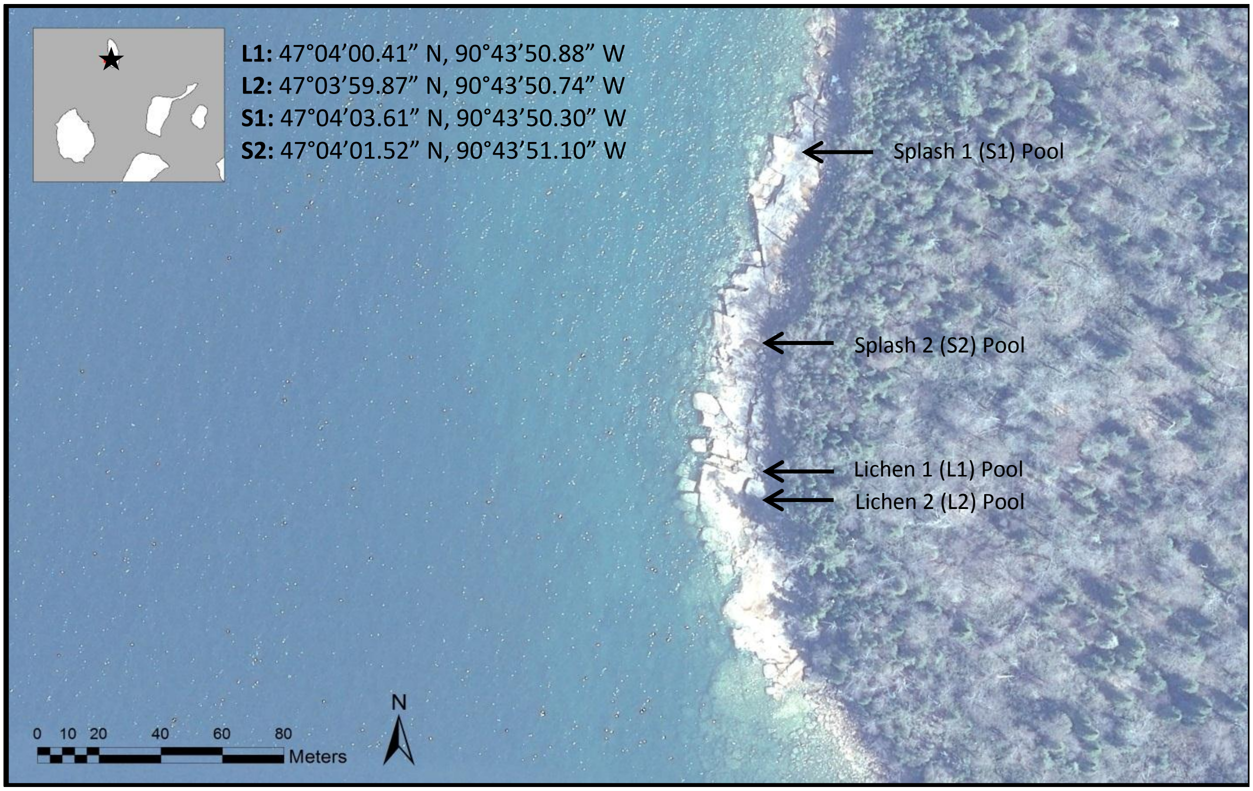


Figure A23. Devil's Island (DI) sample site, Apostle Islands.

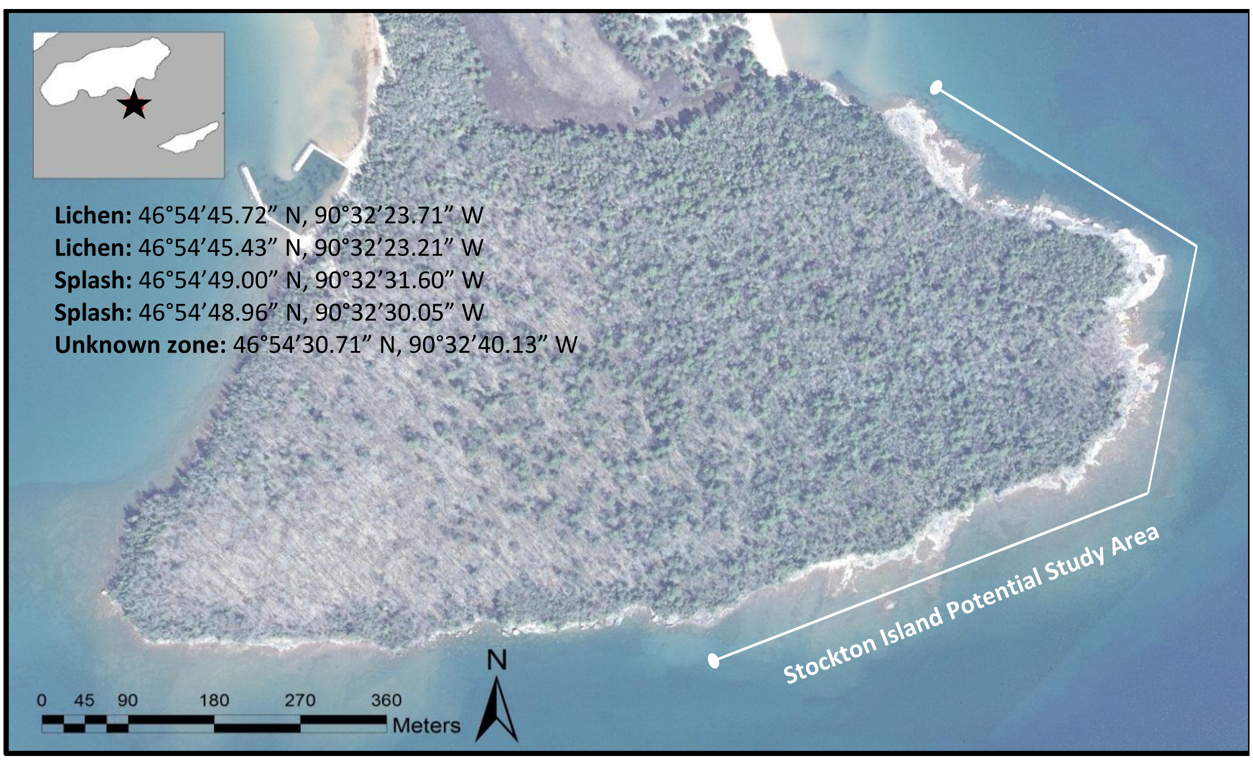


Figure A24. Stockton Island (SI) potential study area, Apostle Islands.

Table A1. Study sites and pool locations for biological collections, 2009-2010.

Site	Site and Sample Abbreviations	Sampling Year	Coordinates for permanent pools or ephemeral zones (NAD83)	Bedrock formation
Isle Royale				
Passage Island	PA	2010		Portage Lake Volcanics
Lichen 1 pool	L1		48°13'48.0" N, 88°21'10.9" W	
Lichen 2 pool	L2		48°13'48.4" N, 88°21'10.2" W	
Lichen ephemeral pools	LE			
Splash 1 pool	S1		48°13'48.0" N, 88°21'09.5" W	
Splash 2 pool	S2		48°13'46.6" N, 88°21'13.5" W	
Splash ephemeral pools	SE			
Raspberry Island	RS	2010		Portage Lake Volcanics
Lichen 1 pool	L1		48°08'27.1° N, 88°28'29.0" W	
Lichen 2 pool	L2		48°08'26.3° N, 88°28'30.0" W	
Lichen ephemeral pools	LE			
Splash 1 pool	S1		48°08'25.4° N, 88°28'30.3" W	
Splash 2 pool	S2		48°08'28.5° N, 88°28'24.7" W	
Splash ephemeral pools	SE			
Blueberry Cove	BL	2010		Portage Lake Volcanics
Lichen 1 pool	L1		48°00'36.9" N, 88°40'32.2" W	
Lichen 2 pool	L2		48°00'36.2" N, 88°40'33.0" W	
Lichen ephemeral pools	LE			
Splash 1 pool	S1		48°00'39.2" N, 88°40'28.8" W	
Splash 2 pool	S2		48°00'33.8" N, 88°40'37.2" W	
Splash ephemeral pools	SE			
Datolite Mine	DM	2010		Portage Lake Volcanics
Lichen 1 pool	L1		47°59'30.8" N, 88°44'06.2" W	
Lichen 2 pool	L2		47°59'30.1" N, 88°44'09.5" W	
Lichen ephemeral pools	LE			
Splash 1 pool	S1		47°59'30.5" N, 88°44'04.4" W	
Splash 2 pool	S2		47°59'30.2" N, 88°44'06.4" W	
Splash ephemeral pools	SE			
Blake's Point	BP	2009		Portage Lake Volcanics
Lichen zone			48°11'24.3" N, 88°25'21.4" W	
Splash zone			48°11'24.3" N, 88°25'21.4" W	
Third Island	TH	2009		Portage Lake Volcanics
Lichen zone			48°10'53.3" N, 88°25'37.7" W	
Splash zone			48°10'53.3" N, 88°25'37.7" W	
Edward's Island	ED	2009		Portage Lake Volcanics
Lichen zone			48°10'15.5" N, 88°26'08.4" W	
Splash zone			48°10'15.5" N, 88°26'08.4" W	
North Government Island	NG	2009		Portage Lake Volcanics
Lichen zone			48°10'45.0" N, 88°25'15.4" W	
Splash zone			48°10'45.0" N, 88°25'15.4" W	
South Government Island	SG	2009		Portage Lake Volcanics
Lichen zone			48°10'13.4" N, 88°25'13.7" W	
Splash zone			48°10'13.4" N, 88°25'13.7" W	
Scoville Point	SP	2009		Portage Lake Volcanics
Lichen zone			48°09'44.5" N, 88°27'04.0" W	
Splash zone			48°09'44.5" N, 88°27'04.0" W	
Bat Island	BA	2009		Portage Lake Volcanics
Lichen zone			48°08'44.7" N, 88°27'52.9" W	
Splash zone			48°08'44.7" N, 88°27'52.9" W	
Raspberry Island	RA	2009		Portage Lake Volcanics
Lichen zone			48°08'26.3" N, 88°28'30.2" W	
Splash zone			48°08'25.6" N, 88°28'30.6" W	
Smithwick Island	SM	2009		Portage Lake Volcanics
Lichen zone			48°08'17.7" N, 88°28'59.8" W	
Splash zone			48°08'17.7" N, 88°28'59.8" W	
Shaw Island	SH	2009		Portage Lake Volcanics
Lichen zone			48°07'59.3" N, 88°29'42.0" W	
Splash zone			48°07'59.3" N, 88°29'42.0" W	
Davidson Island	DA	2009		Portage Lake Volcanics
Lichen zone			48°07'30.3" N, 88°30'38.2" W	
Splash zone			48°07'30.3" N, 88°30'38.2" W	
Outer Hill Island	OH	2009		Portage Lake Volcanics
Lichen zone			48°07'03.2" N, 88°31'18.4" W	
Splash zone			48°07'03.2" N, 88°31'18.4" W	

Table A1. Study sites and pool locations for biological collections, 2009-2010 (continued).

Site	Site and Sample Abbreviations	Sampling Year	Coordinates for permanent pools or ephemeral zones (NAD83)	Bedrock formation
Mott Island	MO	2009		Portage Lake Volcanics
Lichen zone			48°06'32.5" N, 88°32'18.6" W	
Splash/Transition zone			48°06'32.4" N, 88°32'16.4" W	
East Caribou Island	EC	2009		Portage Lake Volcanics
Lichen/Transition zone			48°06'07.8" N, 88°33'04.3" W	
Splash zone			48°06'07.8" N, 88°33'04.3" W	
West Caribou Island	WC	2009		Portage Lake Volcanics
Lichen zone			48°05'49.4" N, 88°33'37.6" W	
Apostle Islands				
Bear Island	BI	2010		Chequamegon Sandstone
Lichen 1 pool	L1		47°01'35.62" N, 90°44'48.52" W	
Lichen 2 pool	L2		47°01'35.47" N, 90°44'45.24" W	
Lichen ephemeral pools	LE			
Splash 1 pool	S1		47°01'35.11" N, 90°44'50.42" W	
Splash 2 pool	S2		47°01'34.32" N, 90°44'45.46" W	
Splash ephemeral pools	SE			
Devil's Island	DI	2010		Devil's Island Sandstone
Lichen 1 pool	L1		47°04'00.41" N, 90°43'50.88" W	
Lichen 2 pool	L2		47°03'59.87" N, 90°43'50.74" W	
Lichen ephemeral pools	LE			
Splash 1 pool	S1		47°04'03.61" N, 90°43'50.30" W	
Splash 2 pool	S2		47°04'01.52" N, 90°43'51.10" W	
Splash ephemeral pools	SE			
Pictured Rocks				
Miner's Beach	MB	2010		Chapel Rock/Miners Castle
Lichen 1 pool	L1		46°30'08.56" N, 86°31'49.94" W	Sandstone
Lichen 2 pool	L2		46°30'00.11" N, 86°31'53.26" W	
Splash pool	S2		46°30'00.58" N, 86°31'53.47" W	
Lichen zone	LZ			
Splash zone	SZ			
AuSable Point	AS	2010		Jacobsville Sandstone
Lichen 1 pool	L1		46°40'21.50" N, 86°08'16.33" W	
Lichen 2 pool	L2		46°40'21.50" N, 86°08'16.19" W	
Splash 1 pool	S1		46°40'20.39" N, 86°08'13.99" W	
Splash 2 pool	S2		46°40'19.85" N, 86°08'13.88" W	
Mosquito Harbor	MH	2010		Chapel Rock/Miners Castle
Cave landing	CA		46°31'38.14" N, 86°29'37.36" W	Sandstone
Mediculous zone	MZ		46°31'42.24" N, 86°29'30.84" W	
Splash 1 pool	S1		46°31'39.22" N, 86°29'33.00" W	

Study Site Pool Photographs

Figure A25. Bear Island (BI) sample pool photographs, Apostle Islands.

Figure A26. Devil’s Island (DI) sample pool photographs, Apostle Islands.

Figure A27. Miner’s Beach (DI) sample pool photographs, Pictured Rocks.

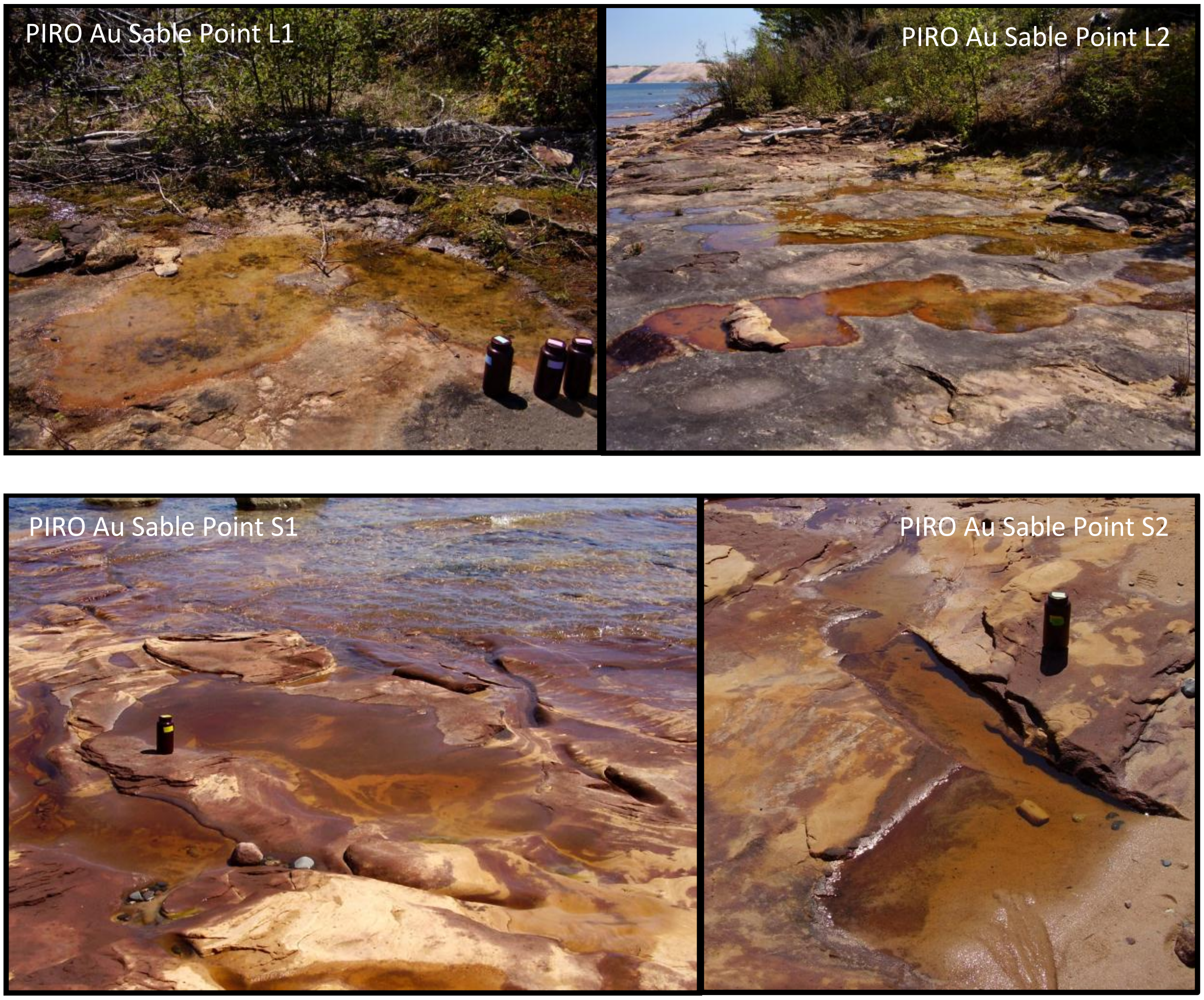


Figure A28. Au Sable Point (AS) sample pool photographs, Pictured Rocks.

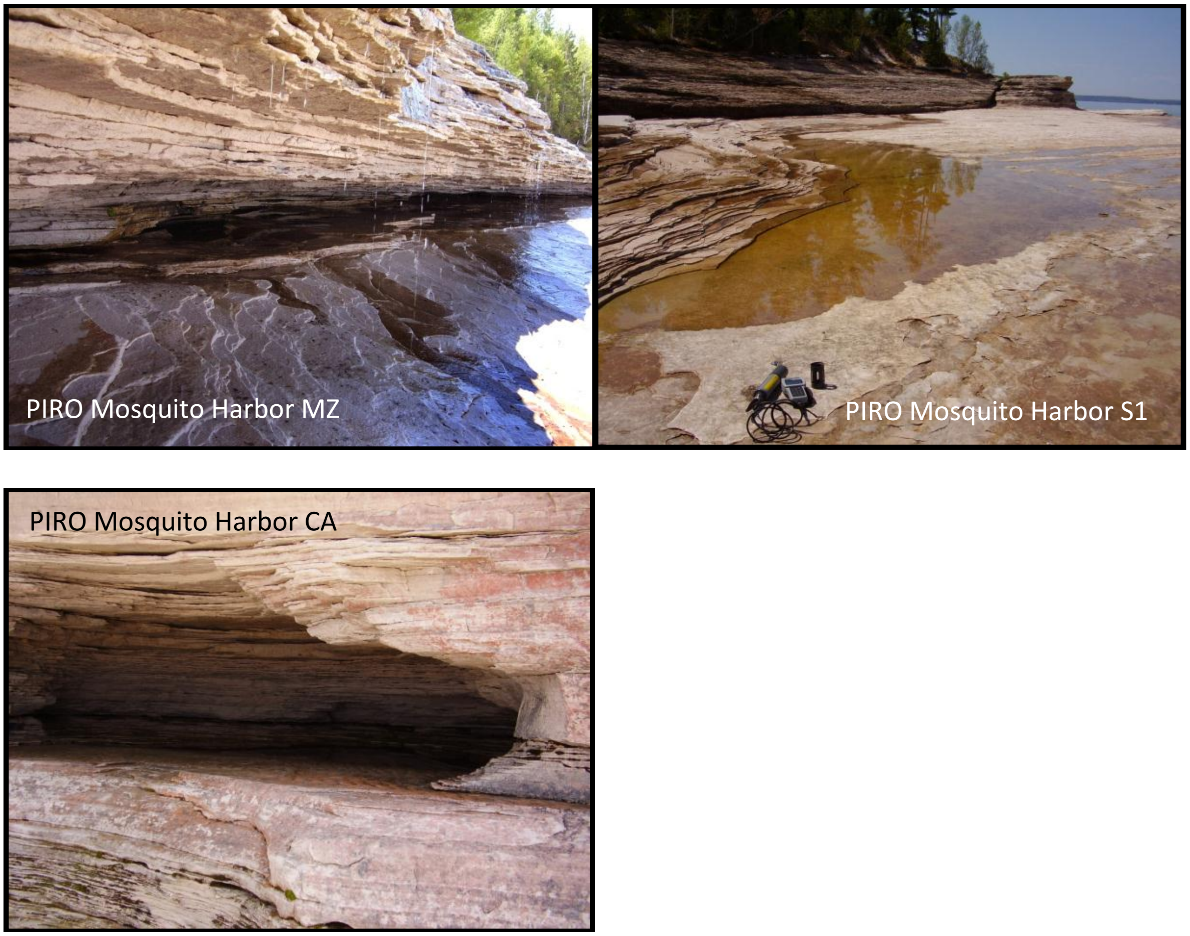


Figure A29. Mosquito Harbor (MH) sample pool photographs, Pictured Rocks.

Figure A30. Passage Island (PA) sample pool photographs, Isle Royale.

Figure A31. Raspberry Island (RS) sample pool photographs, Isle Royale.

Figure A32. Blueberry Cove (BL) sample pool photographs, Isle Royale.

Figure A33. Datolite Mine (DM) sample pool photographs, Isle Royale.

Appendix B: Biogeographic Mapping

Mapping of rock pool densities and characteristics offers managers many advantages: knowledge of fauna and general characteristics of pools that different taxa occupy, predicting where taxa of interest are likely to be found, and creation of a database for coastal monitoring and protection efforts. However, mapping may be an intensive task for shorelines dense with pools. Below are examples of analyses that can be accomplished using the geodatabase created from detailed mapping in 2011–2012 at Isle Royale. Range maps include all amphibians detected to be breeding in coastal pools. Analysis maps include chorus frog presence by ecological zone and by pool permanence, pool distribution and abundance in ecological zones, recharge sources, and distances covered daily during mapping based on pool density. All range maps and analyses were prepared in ArcGIS 10.1 (© 1995–2012 Esri Inc.).

Range Maps of Isle Royale Amphibians

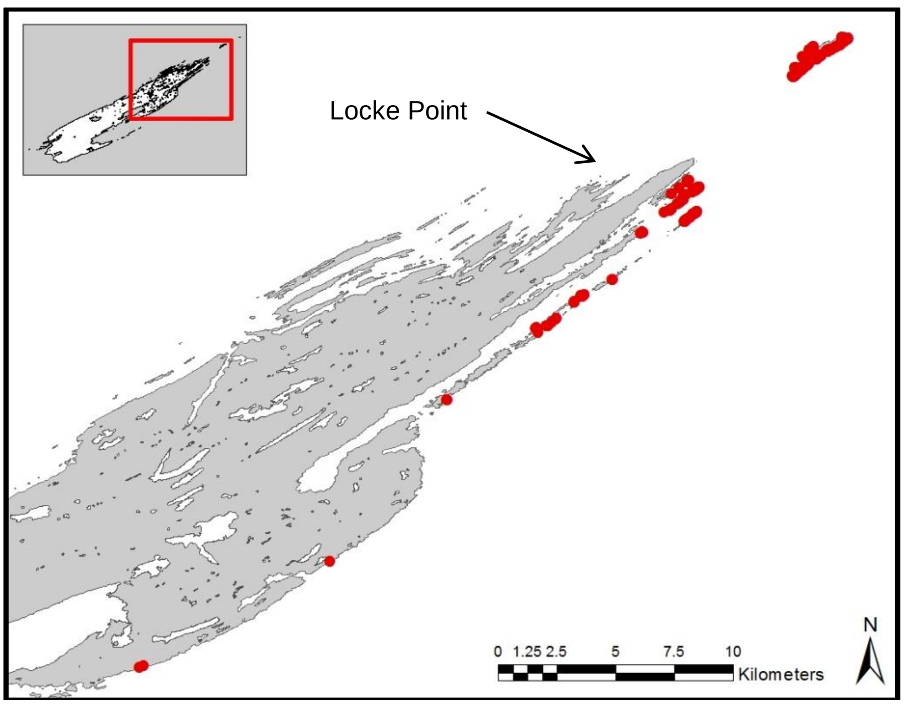


Figure B1. Known chorus frog (*Pseudacris triseriata*) coastal distribution at Isle Royale. All points indicate larval presence in pools. Additional range southwest is unknown, while north shore range is known to extend at least to small islands west of Locke Point.

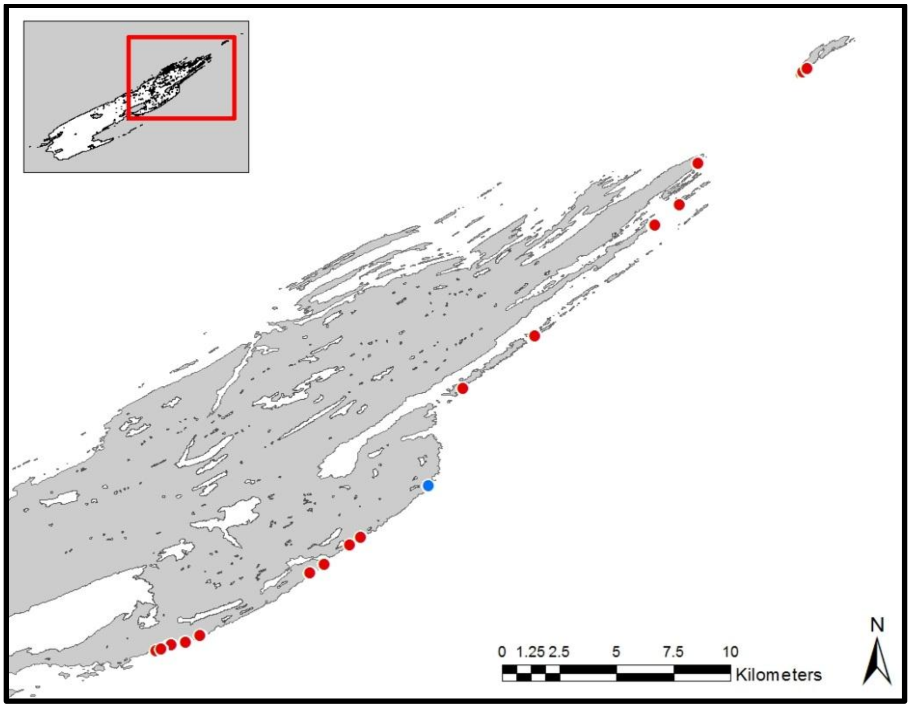


Figure B2. Known spring peeper (*Pseudacris crucifer*) coastal distribution at Isle Royale. Red points indicate larval presence in pools, blue points indicate adult presence. Additional range in rock pools on north shore and west end is unknown.

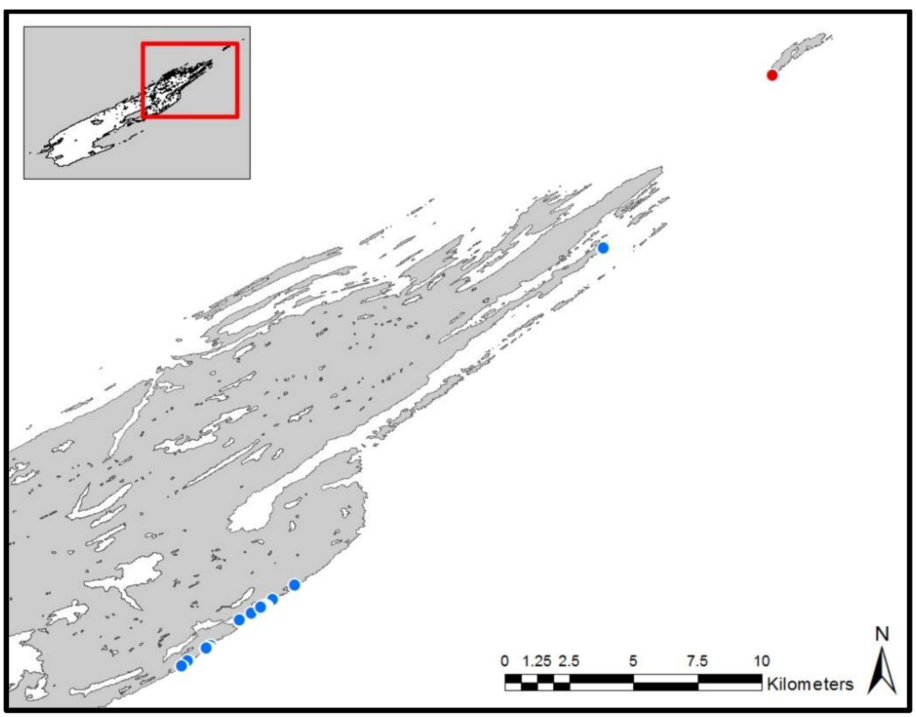


Figure B3. Known green frog (*Rana clamitans*) coastal distribution at Isle Royale. Red points indicate larval presence in pools, blue points indicate adult presence. Additional range in rock pools on north shore and west end is unknown.

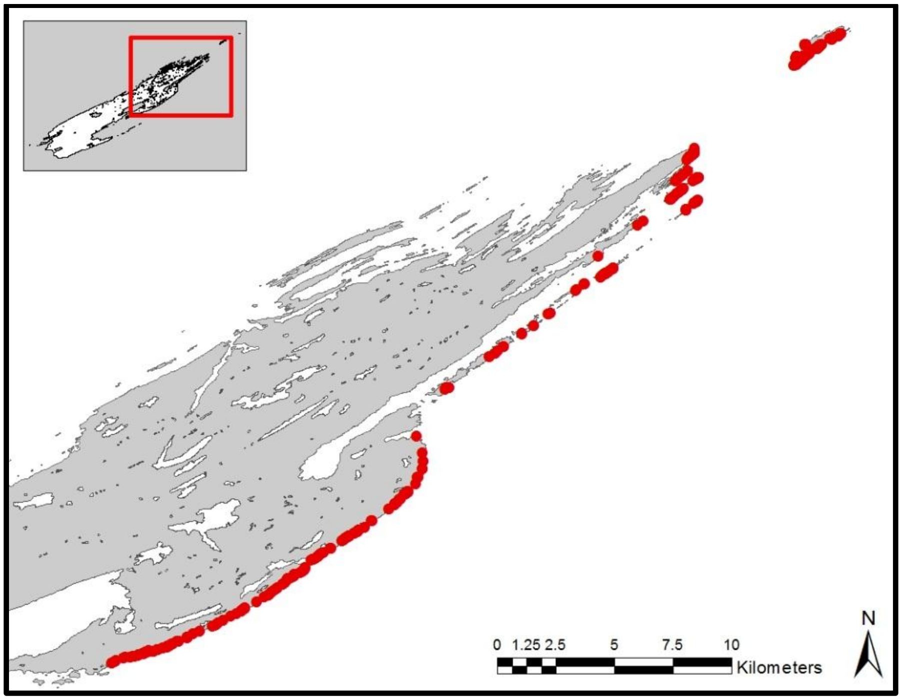


Figure B4. Known blue-spotted salamander (*Ambystoma laterale*) coastal distribution at Isle Royale. All points indicate larval presence in pools. Additional range in rock pools on north shore and west end is unknown.

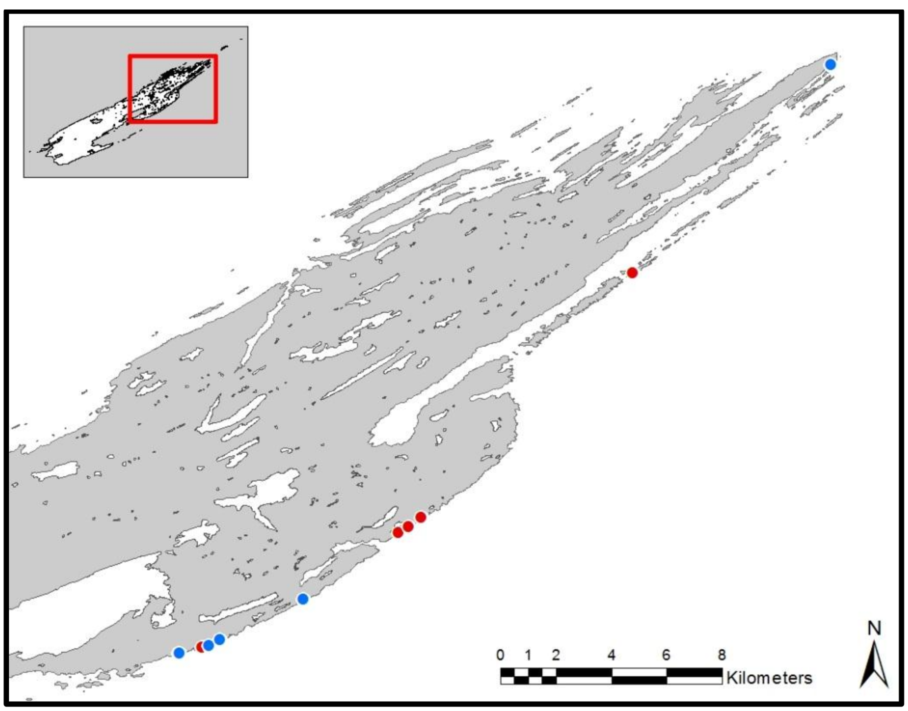


Figure B5. Known eastern newt (*Notophthalmus viridescens*) coastal distribution at Isle Royale. Red points indicate larval presence in pools, blue points indicate adult presence. Additional range in rock pools on north shore and west end is unknown.

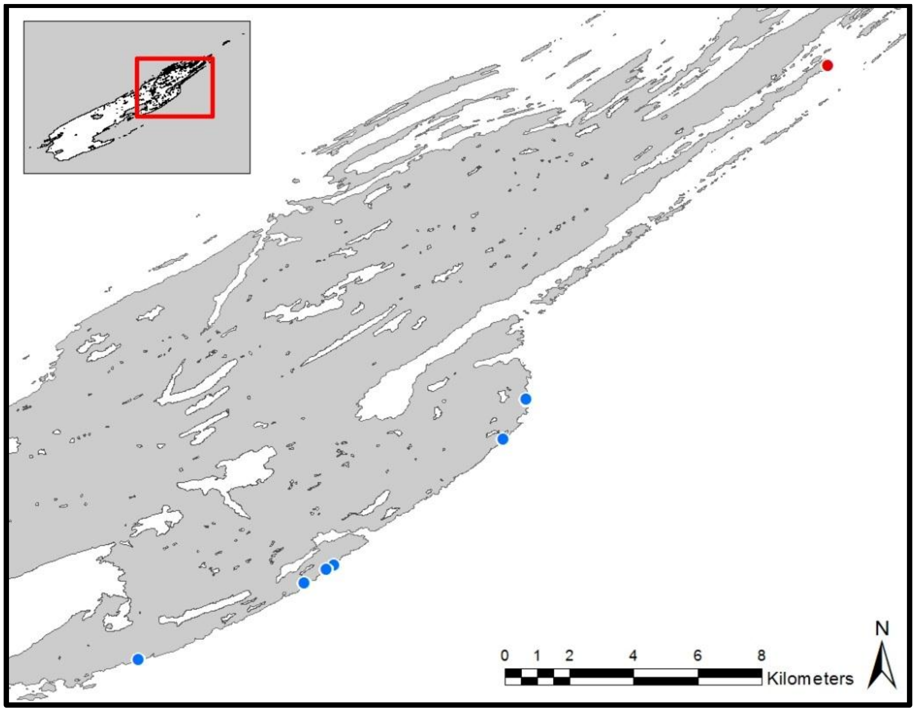


Figure B6. Known American toad (*Bufo americanus*) coastal distribution at Isle Royale. Red points indicate larval presence in pools, blue points indicate adult presence. Additional range in rock pools on north shore and west end of island is unknown.

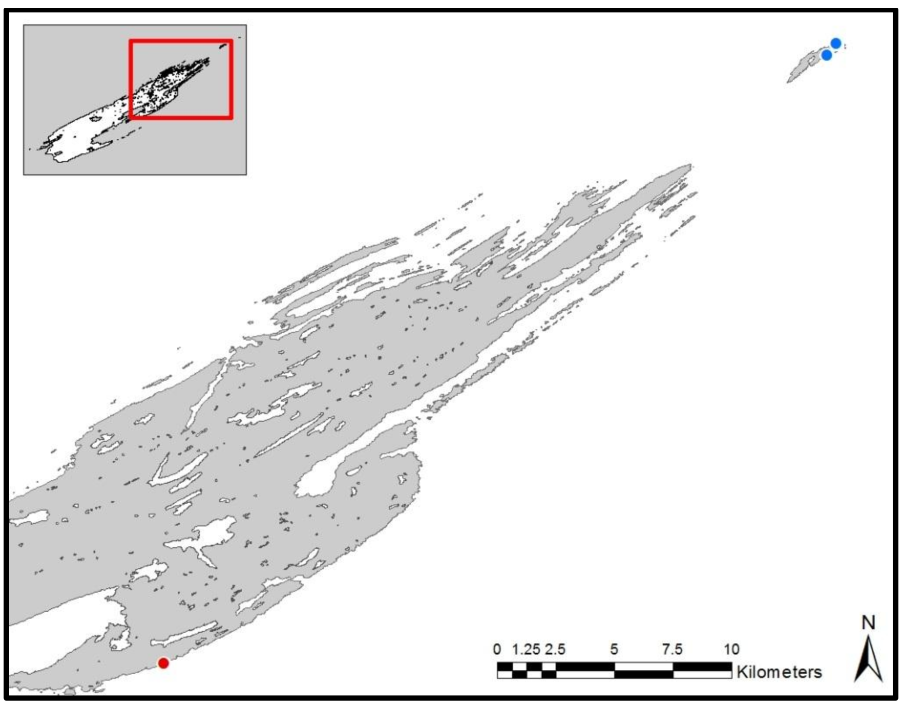


Figure B7. Known wood frog (*Rana sylvatica*) coastal distribution at Isle Royale. Red points indicate larval presence in pools, blue points indicate adult presence. Additional range in rock pools on north shore and west end of island is unknown.

Selected Analytical Examples for Biogeographic Database

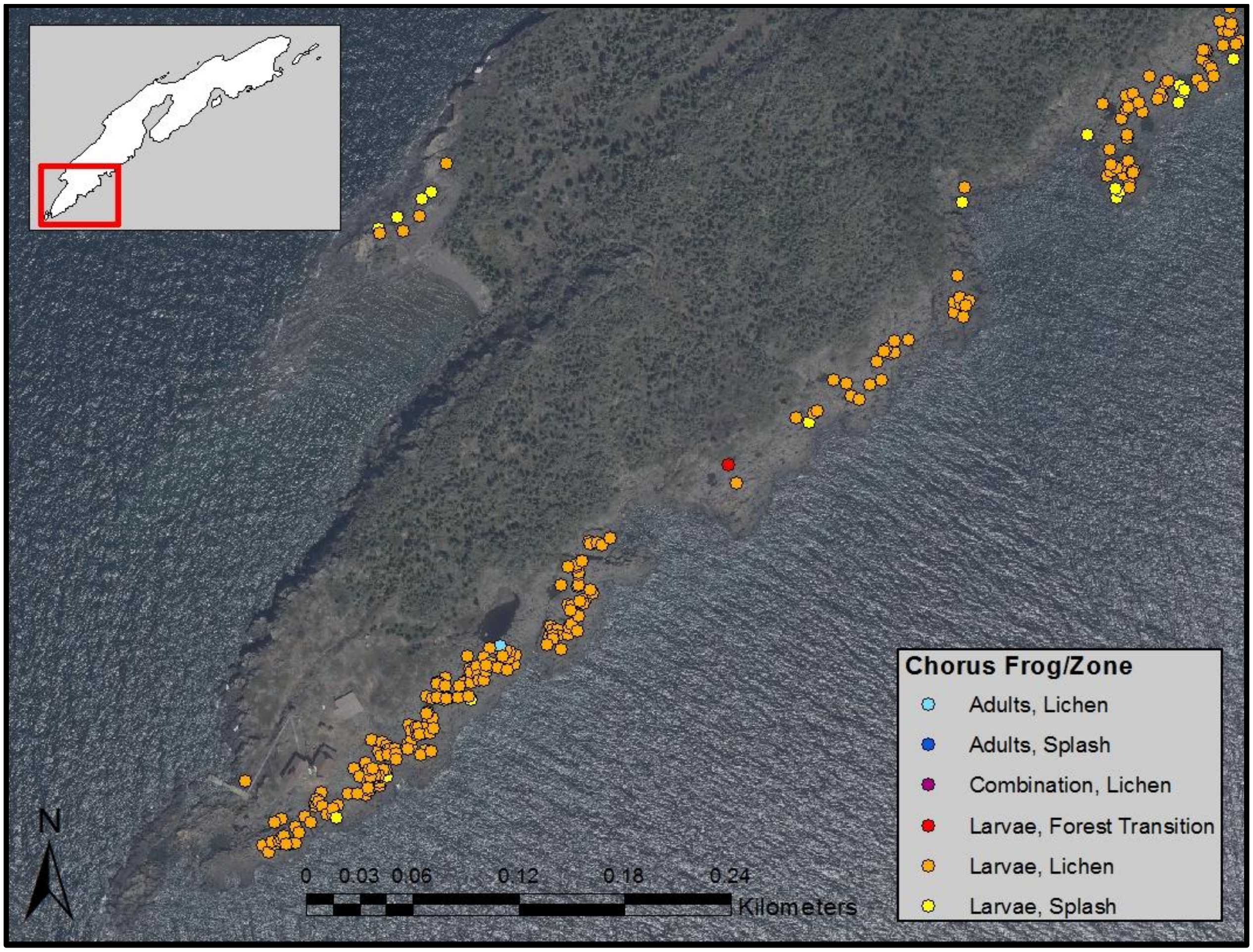


Figure B8. Chorus frog use of ecological zones at the west end of Passage Island, Isle Royale. Clearly chorus frogs prefer laying eggs in lichen zone pools and adults appear to return to forested habitats after laying eggs; therefore, during spill responses most of the chorus frog population should not be in contact with pollutants if wave conditions are low or moderate.

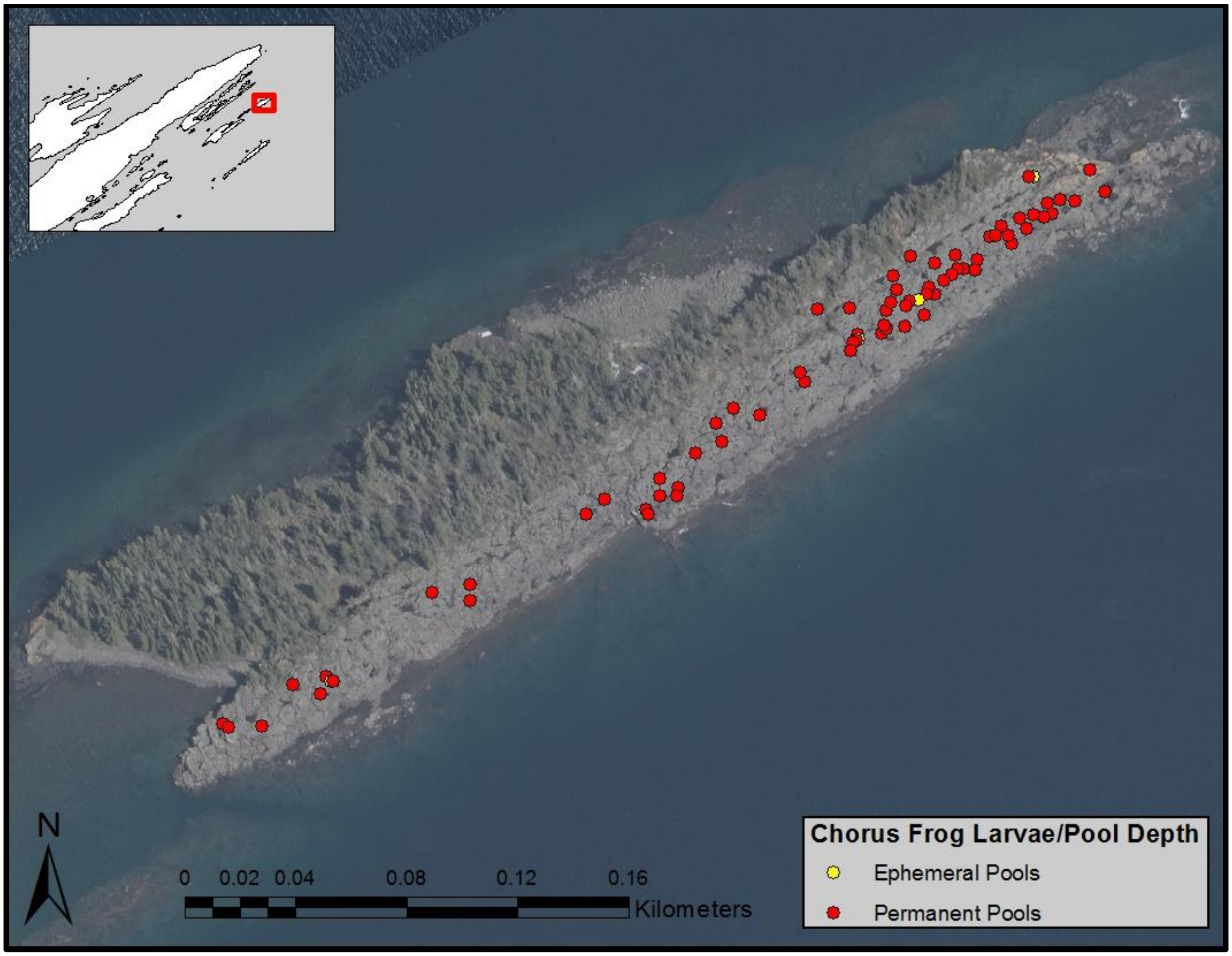
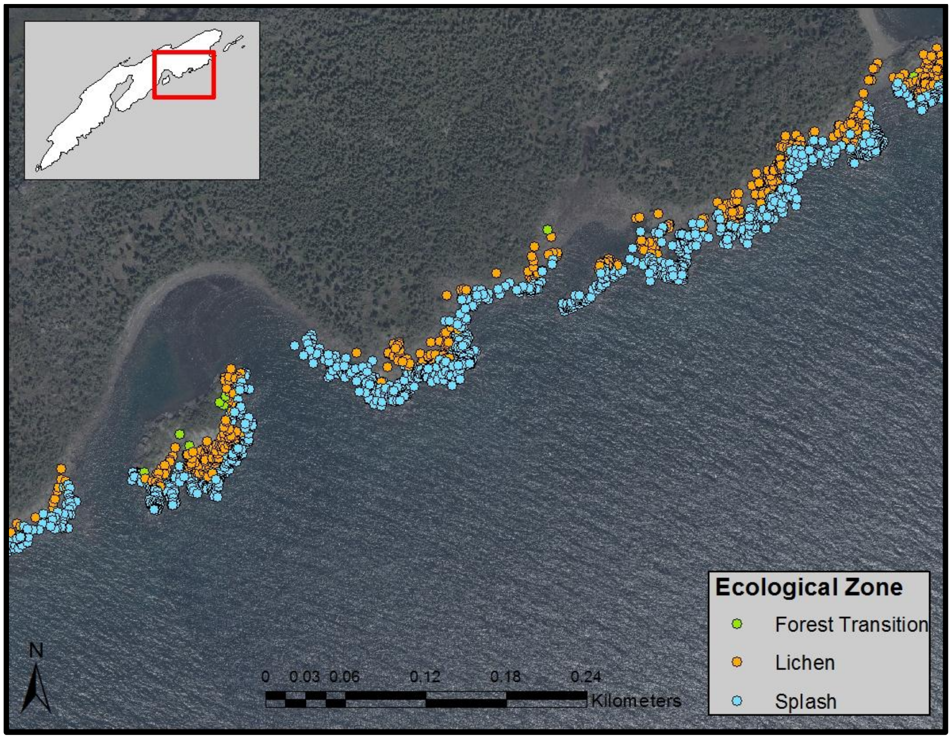


Figure B9. Chorus frog use of permanent versus ephemeral pools at North Government Island, Isle Royale. Based on a clear preference for permanent pools, which is typical of chorus frogs across their Isle Royale range, it is likely that they would not be strongly affected by limited or moderate droughts.



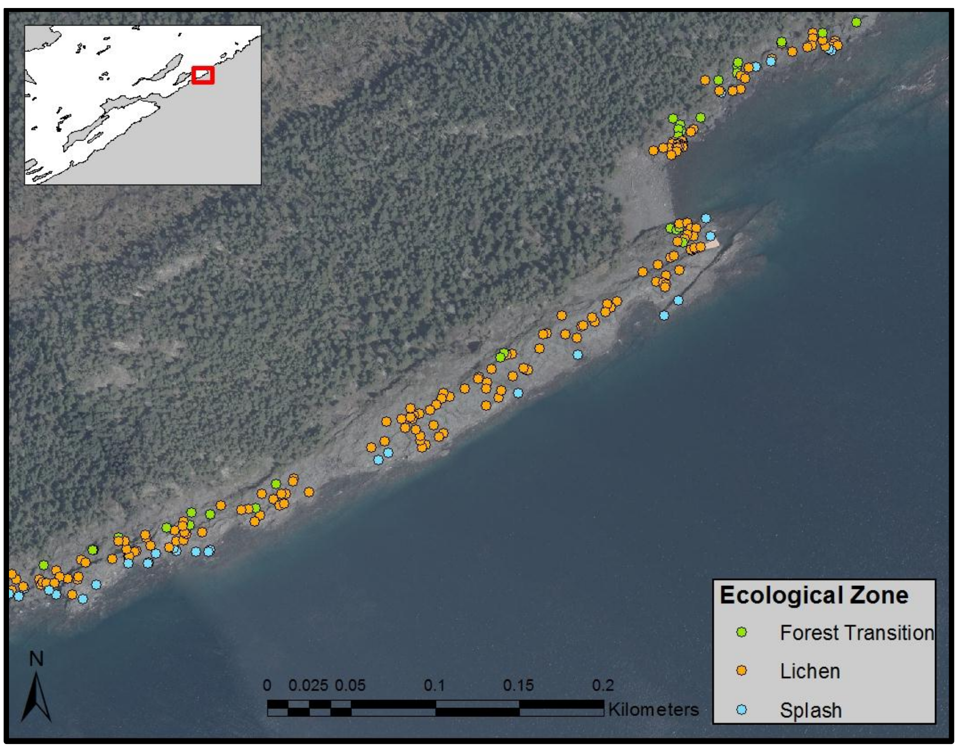


Figure B10. Pool distribution and abundance in ecological zones, Passage Island (A) and near Chippewa Harbor (B), Isle Royale. Zonal analysis helps estimate the potential pollutant (e.g., oil spills) impact to particular shorelines. Passage Island has many more splash zone pools than the Chippewa Harbor area (probably due to bedrock morphology and slope to the lake), adding to the importance of protecting this site from spills.

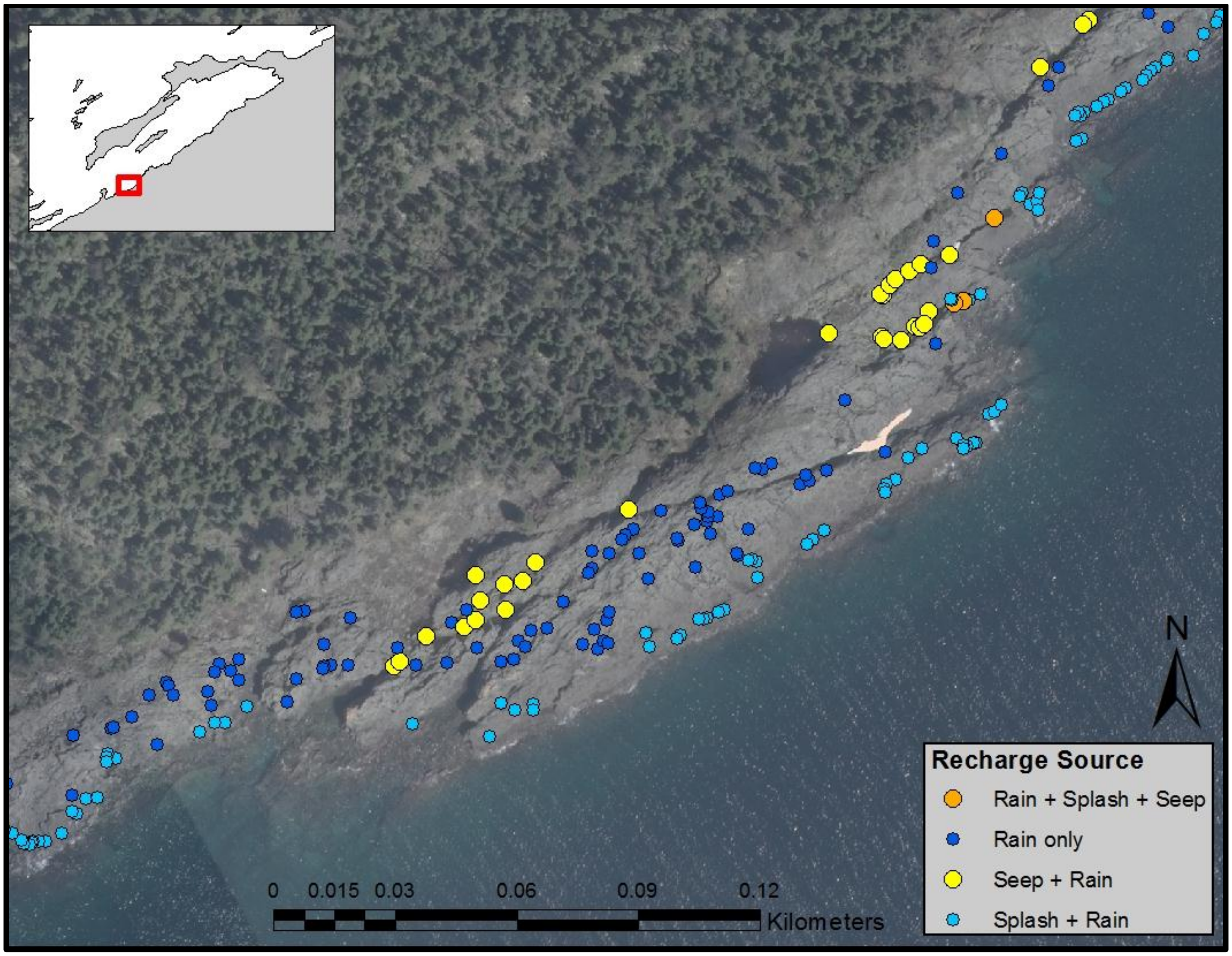


Figure B11. Recharge sources of pools south of Chippewa Harbor, Isle Royale. This type of analysis may be used to identify pools receiving overland flow, only rainfall, or splash from Lake Superior. Water chemistry is likely to be different in each of these pool types and may define the presence of particular species or communities. Seep and splash zone pools should be able to better withstand drought conditions and act as a community reservoir during extremely dry conditions.

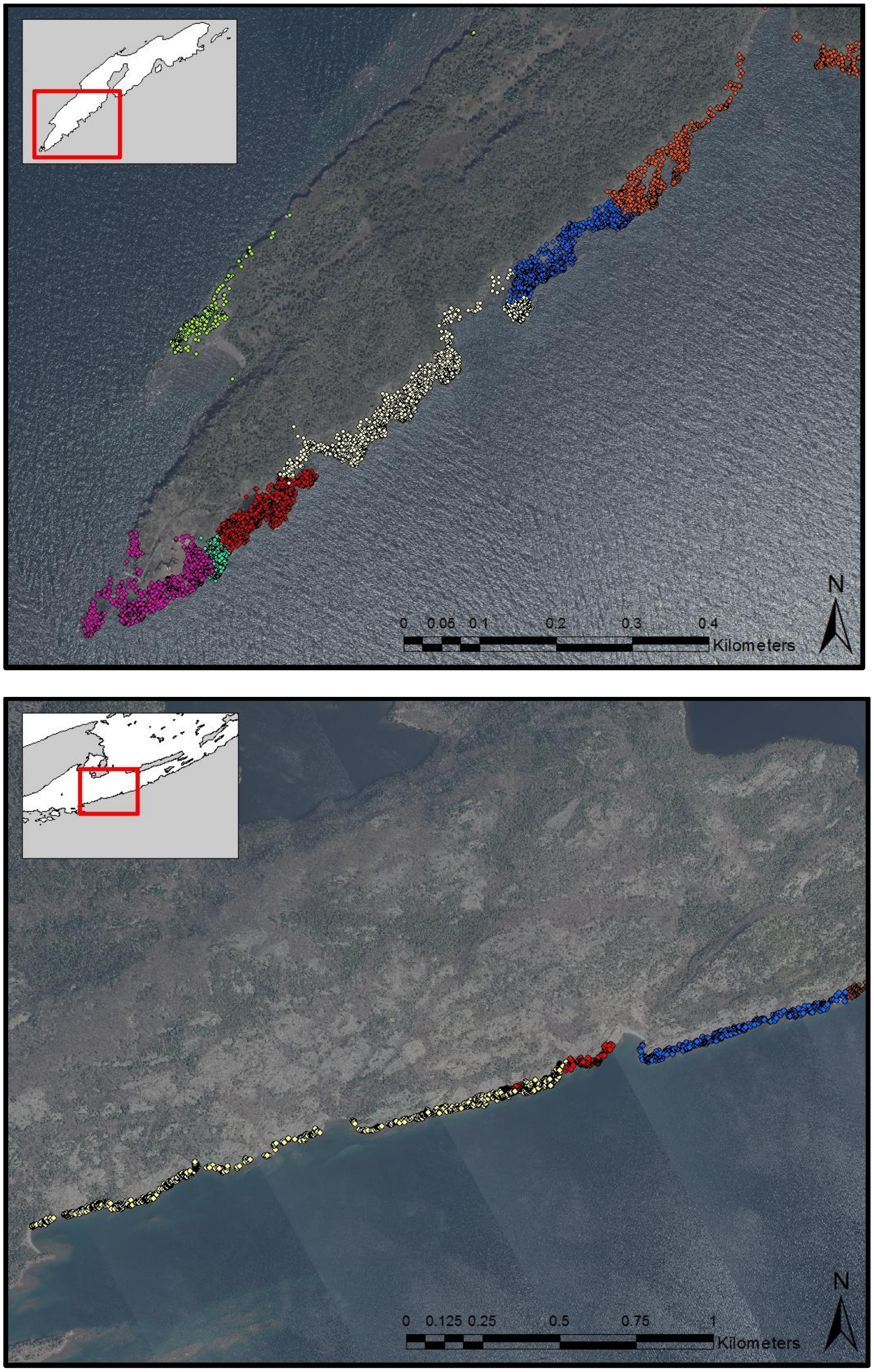


Figure B12. Daily distances covered while mapping the west half of Passage Island (A) and the Datolite Mine area (B), Isle Royale. With extremely high pool densities, Passage Island mapping was often limited to 0.2–0.4 km/day (indicated by different colors) with 2–3 people, while moderate densities near Datolite allowed up to 0.5–1.5 km/day with 1–2 people. In these maps, about 24,800 pools are represented at Passage Island (covering 1.5 km) and 1,700 pools around Datolite Mine (covering 3 km). This reveals almost a 30:1 ratio of pool density difference between Passage Island and the south shore of Isle Royale.

Appendix C: Field and Physical Measurements and Water Quality Data from Rock Pools, 2010.

The tables included in Appendix E represent data collected during the 2010 field sampling and analysis of waters from rock pools and Lake Superior at Isle Royale National Park, Apostle Island National Lakeshore, and Pictured Rocks National Lakeshore. Data are presented in seven tables.

Table C-1a. Metadata parameters, codes, and descriptors for park, site, pool, and sampling dates, 2010.

Table C-1b. Metadata parameters, codes, units, and descriptors for field and water quality parameters collected in 2010.

Table C-2. Sample inventory and codes for water quality samples.

Table C-3. Field and physical data.

Table C-4. Total and dissolved nutrients.

Table C-5. Anions in water samples.

Table C-6. Cations in water samples.

Table C-7a. Metals in water samples, molecular weights 7Li–66Zn.

Table C-7b. Metals in water samples, molecular weights 75As–235U.

Table C-1a. Metadata parameters, codes, and descriptors for park, site, pool, and sampling dates, 2010.

Parameter	Codes and Description		
SAMPLE NR	**7-character identifier for each sampling event by park, site, pool, month**		
PARK	**Four-letter or one-letter abbreviation for each GLKN park unit**		
	ISRO	I	Isle Royale National Park
	APIS	A	Apostle Islands National Lakeshore
	PIRO	P	Pictured Rocks National Lakeshore
SITE	**Two-letter abbreviation for each sampling site**		
	BL	Blueberry Cove (ISRO)	
	DM	Datolite Mine (ISRO)	
	PA	Passage Island (ISRO)	
	RS	Raspberry Island (ISRO	
	BI	Bear Island (APIS)	
	DI	Devils Island (APIS)	
	AS	AuSable Point (PIRO)	
	MB	Miners Bay (PIRO)	
	MH	Mosquito Harbor (PIRO)	
POOL	**two letter or letter-digit combination to identify specific pool or type of pool habitat, LK used for Lake Superior in stats data**		
	L1	Lichen zone pool 1	
	L2	Lichen zone pool 2	
	LE	Lichen zone ephemeral pools	
	LZ	Lichen zone pool (PIRO Miners Bay only)	
	S1	Splash zone pool 1	
	S2	Splash zone pool 2	
	SE	Splash zone ephemeral pools	
	CA	Cave pool (PIRO Mosquito Harbor only)	
	MZ	Mediculous zone (PIRO Mosquito Harbor only)	
	LSUP or LK	Lake Superior	
DATE	**six digit number for sampling date YYMMDD, set up this way they can be sorted in chronological order most easily**		
MONTH	**Three-letter abbreviation or two-digit number for sampling month**		
	MAY	05	May
	JUN	06	June
	JUL	07	July
	AUG	08	August
	SEP	09	September
	OCT	10	October

Table C-1b. Metadata parameters, codes, units, and descriptors for field and water quality parameters collected in 2010.

Nutrients/Chl-*a*	**units**	**Description**
TP	µg P/L	total phosphorus
TN	mg N/L	total nitrogen
DOC	ppm	dissolved organic carbon
DIC	mg C/L	dissolved inorganic carbon
NO-X	mg N/L	nitrate-nitrite N
NH4	mg N/L as NH4	ammonium
SRP	µg P/L	soluble reactive phosphorus
Chl-a	µg/L	chlorophyll-*a*

Physical/Field	**units**	**Description**
Lat	°N	decimal degrees North
Long	°W	decimal degrees W
Dist from treeline	m	distance from pool to treeline, in m
Dist from lake	m	distance from pool to lake, in m
Length	m	longest axis of pool, in m
Width	m	orthogonal to length, in m
Depth	m	depth of pool at diatom collection site, in m
Spec Cond	mS/cm	specific conductivity, mS/cm
DO	mg/L	dissolved oxygen, mg/L
DO%	% sat	percent DO saturation, %
Temp	°C	temperature, °C
pH	pH	pH of pool

Cations	**units**	**Description**
Al1670	ng/g	Aluminum
Ba4554	ng/g	Barium
Ca3158	ng/g	Calcium
Fe2382	ng/g	Iron
K_7664	ng/g	Potassium
Li6707	ng/g	Lithium
Mg2852	ng/g	Magnesium
Mn2576	ng/g	Manganese
Na5895	ng/g	Sodium
P_1774	ng/g	Phosphorus
Si2516	ng/g	Silica
Sr4215	ng/g	Strontium

Anions	**units**
Fluoride	µg/g
Lactate	µg/g
Acetate	µg/g
Formate	µg/g
Chloride	µg/g
Nitrite - N	µg/g
Bromide	µg/g
Nitrate - N	µg/g

Table C-1b. Metadata parameters, codes, units, and descriptors for field and water quality parameters collected in 2010 (continued).

Anions	units
Sulfate	µg/g
Oxalate	µg/g
Thiosulfate	µg/g
Phosphate - P	µg/g

Trace Metals	units	Description
7Li	ppb	Lithium
9Be	ppb	Beryllium
11B	ppb	Boron
27Al	ppb	Aluminum
31P	ppb	Phosphorus
47Ti	ppb	Titanium
51V	ppb	Vanadium
52Cr	ppb	Chromium
55Mn	ppb	Manganese
56Fe	ppb	Iron
59Co	ppb	Cobalt
60Ni	ppb	Nickel
63Cu	ppb	Copper
66Zn	ppb	Zinc
75As	ppb	Arsenic
78Se	ppb	Selenium
85Rb	ppb	Rubidium
88Sr	ppb	Strontium
90Zr	ppb	Zirconium
93Nb	ppb	Niobium
95Mo	ppb	Molybdenum
111Cd	ppb	Cadmium
138Ba	ppb	Barium
181Ta	ppb	Tantalum
182W	ppb	Tungsten
205Tl	ppb	Thallium
208Pb	ppb	Lead
238U	ppb	Uranium

Table C-2. Sample inventory and codes for water quality samples collected from rock pools and Lake Superior at Apostle Islands National Lakeshore (APIS), Isle Royale National Park (ISRO), and Pictured Rocks National Lakeshore (PIRO), 2010. See Table C-1 for metadata.

PARK		SITE	POOL	POOL	DATE	MONTH		SAMPLE NR
APIS	A	BI	L1	L1	100505	MAY	05	ABIL105
APIS	A	BI	L2	L2	100505	MAY	05	ABIL205
APIS	A	BI	S1	S1	100505	MAY	05	ABIS105
APIS	A	BI	S2	S2	100505	MAY	05	ABIS205
APIS	A	BI	L1	L1	100914	SEP	09	ABIL109
APIS	A	BI	L2	L2	100914	SEP	09	ABIL209
APIS	A	BI	S1	S1	100914	SEP	09	ABIS109
APIS	A	BI	S2	S2	100914	SEP	09	ABIS209
APIS	A	DI	L1	L1	100506	MAY	05	ADIL105
APIS	A	DI	L2	L2	100506	MAY	05	ADIL205
APIS	A	DI	S1	S1	100506	MAY	05	ADIS105
APIS	A	DI	S2	S2	100506	MAY	05	ADIS205
APIS	A	DI	L1	L1	100915	SEP	09	ADIL109
APIS	A	DI	L2	L2	100915	SEP	09	ADIL209
APIS	A	DI	S1	S1	100915	SEP	09	ADIS109
APIS	A	DI	S2	S2	100915	SEP	09	ADIS209
ISRO	I	BL	L1	L1	100517	MAY	05	IBLL105
ISRO	I	BL	L2	L2	100517	MAY	05	IBLL205
ISRO	I	BL	S1	S1	100517	MAY	05	IBLS105
ISRO	I	BL	S2	S2	100517	MAY	05	IBLS205
ISRO	I	BL	L1	L1	100706	JUL	07	IBLL107
ISRO	I	BL	L2	L2	100706	JUL	07	IBLL207
ISRO	I	BL	S1	S1	100706	JUL	07	IBLS107
ISRO	I	BL	S2	S2	100706	JUL	07	IBLS207
ISRO	I	BL	L1	L1	101004	OCT	10	IBLL110
ISRO	I	BL	L2	L2	101004	OCT	10	IBLL210
ISRO	I	BL	S1	S1	101004	OCT	10	IBLS110
ISRO	I	BL	S2	S2	101004	OCT	10	IBLS210
ISRO	I	DM	L1	L1	100516	MAY	05	IDML105
ISRO	I	DM	L2	L2	100516	MAY	05	IDML205
ISRO	I	DM	S1	S1	100516	MAY	05	IDMS105
ISRO	I	DM	L1	L1	100707	JUL	07	IDML107
ISRO	I	DM	L2	L2	100707	JUL	07	IDML207
ISRO	I	DM	S1	S1	100707	JUL	07	IDMS107
ISRO	I	DM	S2	S2	100707	JUL	07	IDMS207
ISRO	I	DM	L1	L1	101003	OCT	10	IDML110
ISRO	I	DM	L2	L2	101003	OCT	10	IDML210
ISRO	I	DM	S1	S1	101003	OCT	10	IDMS110
ISRO	I	DM	S2	S2	101003	OCT	10	IDMS210
ISRO	I	PA	L1	L1	100518	MAY	05	IPAL105
ISRO	I	PA	L2	L2	100518	MAY	05	IPAL205
ISRO	I	PA	S1	S1	100518	MAY	05	IPAS105
ISRO	I	PA	S2	S2	100518	MAY	05	IPAS205
ISRO	I	PA	L1	L1	100708	JUL	07	IPAL107

Table C-2. Sample inventory and codes for water quality samples collected from rock pools and Lake Superior at Apostle Islands National Lakeshore (APIS), Isle Royale National Park (ISRO), and Pictured Rocks National Lakeshore (PIRO), 2010. See Table C-1 for metadata (continued).

PARK		SITE	POOL	POOL	DATE	MONTH		SAMPLE NR
ISRO	I	PA	L2	L2	100708	JUL	07	IPAL207
ISRO	I	PA	S1	S1	100708	JUL	07	IPAS107
ISRO	I	PA	S2	S2	100708	JUL	07	IPAS207
ISRO	I	PA	L1	L1	101001	OCT	10	IPAL110
ISRO	I	PA	L2	L2	101001	OCT	10	IPAL210
ISRO	I	PA	S1	S1	101001	OCT	10	IPAS110
ISRO	I	PA	S2	S2	101001	OCT	10	IPAS210
ISRO	I	RS	L1	L1	100518	MAY	05	IRSL105
ISRO	I	RS	L2	L2	100518	MAY	05	IRSL205
ISRO	I	RS	S1	S1	100518	MAY	05	IRSS105
ISRO	I	RS	S2	S2	100518	MAY	05	IRSS205
ISRO	I	RS	L1	L1	100703	JUL	07	IRSL107
ISRO	I	RS	L2	L2	100703	JUL	07	IRSL207
ISRO	I	RS	S1	S1	100703	JUL	07	IRSS107
ISRO	I	RS	S2	S2	100703	JUL	07	IRSS207
ISRO	I	RS	L1	L1	101002	OCT	10	IRSL110
ISRO	I	RS	L2	L2	101002	OCT	10	IRSL210
ISRO	I	RS	S1	S1	101002	OCT	10	IRSS110
ISRO	I	RS	S2	S2	101002	OCT	10	IRSS210
PIRO	P	AS	L1	L1	100519	MAY	05	PASL105
PIRO	P	AS	L2	L2	100519	MAY	05	PASL205
PIRO	P	AS	S1	S1	100519	MAY	05	PASS105
PIRO	P	AS	S2	S2	100519	MAY	05	PASS205
PIRO	P	AS	L1	L1	100823	AUG	08	PASL108
PIRO	P	AS	L2	L2	100823	AUG	08	PASL208
PIRO	P	MB	L1	L1	100518	MAY	05	PMBL105
PIRO	P	MB	LZ	LZ	100518	MAY	05	PMBLZ05
PIRO	P	MB	S2	S2	100518	MAY	05	PMBS205
PIRO	P	MB	L1	L1	100825	AUG	08	PMBL108
PIRO	P	MB	L2	L2	100825	AUG	08	PMBL208
PIRO	P	MB	LZ	LZ	100825	AUG	08	PMBLZ08
PIRO	P	MH	CA	CA	100520	MAY	05	PMHCA05
PIRO	P	MH	MZ	MZ	100520	MAY	05	PMHMZ05
PIRO	P	MH	S1	S1	100520	MAY	05	PMHS105
PIRO	P	MH	CA	CA	100824	AUG	08	PMHCA08
PIRO	P	MH	MZ	MZ	100824	AUG	08	PMHMZ08
PIRO	P	MH	S1	S1	100824	AUG	08	PMHS108

Table C-3. Field and physical data collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. **Bold** numbers represent estimated values. See Table C-1 for metadata.

PARAMETER: UNITS:	Distance from treeline (m)	Distance from lake (m)	Length (m)	Width (m)	Depth (m)	Spec Cond (mS/cm)	DO (mg/L)	DO% (% sat)	Temp (°C)	pH
SAMPLE NR					***APOSTLE ISLANDS NATIONAL LAKESHORE (APIS, A)***					
ABIL105	6	13.7	11.5	5	0.19	0.105	11.05	101.8	12.08	7.91
ABIL109	6	15.5	5.16	1.6	0.12	0.047	11.52	118.8	16.08	7.96
ABIL205	5	6.5	8	4	0.22	0.098	11.75	101	8.5	7.84
ABIL209	3	6.5	10.1	4.4	0.19	0.057	11.03	108.5	13.93	8.66
ABILK05	N/A	N/A	N/A	N/A	N/A	0.095	N/A	N/A	N/A	8.01
ABILK09	N/A	N/A	N/A	N/A	N/A	0.100	N/A	N/A	N/A	7.97
ABIS105	5	7	10	2.5	0.24	0.1	11.91	102.8	8.77	8.12
ABIS109	8	3.5	4.55	1.5	0.15	0.101	12.39	123.1	14.46	8.68
ABIS205	2	5	4	4	0.29	0.096	11.59	108.4	12.1	7.96
ABIS209	2	4.2	5.2	3.5	0.27	0.077	10.82	110.4	15.5	8.94
ADIL105	2.5	16	1.3	1	0.18	0.027	10.93	92.6	7.97	7.49
ADIL109	2.5	6.5	1.75	1.3	0.1	0.022	9.61	93.6	12.37	5.35
ADIL205	3	2	5	5	0.15	0.029	10.13	80.6	5.35	6.87
ADIL209	2.75	3	3.1	3	0.25	0.026	7.07	66.9	11.18	5.21
ADILK05	N/A	N/A	N/A	N/A	N/A	0.101	N/A	N/A	N/A	7.82
ADILK09	N/A	N/A	N/A	N/A	N/A	0.100	N/A	N/A	N/A	7.63
ADIS105	4	8	6	2.8	0.46	0.05	11.36	98.9	7.84	8.26
ADIS109	3.5	5.6	8.5	5	0.3	0.049	9.43	90.6	12.06	7.59
ADIS205	1.75	5	8	6.5	0.27	0.065	10.87	89.6	6.93	7.99
ADIS209	3.5	2	11.5	8.3	0.3	0.07	9.38	88.9	11.26	7.35
					ISLE ROYALE NATIONAL PARK (ISRO, I)					
IBLL105	0.1	19	12.5	11	0.44	0.0763	10.26	121.7	22.78	8.54
IBLL107	0.1	19	12.5	11	0.44	0.0843	8.25	94.9	20.68	8.35
IBLL110	0.1	19	12.5	11	0.44	0.0515	10.24	97.4	11.59	7.56
IBLL205	11	16	3.5	2.8	0.42	0.0299	9.82	109.7	19.79	7.43
IBLL207	11	16	3.5	2.8	0.42	0.0267	8.34	93.5	19.41	8.22
IBLL210	11	16	3.5	2.8	0.42	0.0247	9.84	92.9	11.3	7.07
IBLLK05	N/A	N/A	N/A	N/A	N/A	**0.100**	N/A	N/A	N/A	**8.00**

Table C-3. Field and physical data collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. **Bold** numbers represent estimated values. See Table C-1 for metadata (continued).

PARAMETER: UNITS:	Distance from treeline (m)	Distance from lake (m)	Length (m)	Width (m)	Depth (m)	Spec Cond (mS/cm)	DO (mg/L)	DO% (% sat)	Temp (°C)	pH
SAMPLE NR					***ISLE ROYALE NATIONAL PARK (ISRO, I)***					
IBLLK07	N/A	N/A	N/A	N/A	N/A	**0.100**	N/A	N/A	N/A	**8.00**
IBLLK10	N/A	N/A	N/A	N/A	N/A	0.096	N/A	N/A	N/A	8.07
IBLS105	15	1.8	8.05	2.7	0.5	0.107	9.83	118.9	23.84	9.02
IBLS107	15	1.8	8.05	2.7	0.5	0.1051	8.33	94.3	19.81	8.83
IBLS110	15	1.8	8.05	2.7	0.5	0.0364	10.81	102.9	11.7	8.12
IBLS205	28	6	2.3	1.17	0.32	0.1176	10.96	132	23.6	9.11
IBLS207	28	6	2.3	1.17	0.32	0.09	9.11	99.8	18.19	8.92
IBLS210	28	6	2.3	1.17	0.32	0.0831	10.99	104.5	11.5	8.65
IDML105	13	21	7.8	4.4	0.4	0.019	8.98	95	16.36	7.69
IDML107	13	21	7.8	4.4	0.4	0.0202	8.21	95.1	21.03	8.17
IDML110	13	21	7.8	4.4	0.4	0.012	10.14	94.7	10.81	7.59
IDML205	14	11	7	2.6	0.63	0.1227	9.41	96.3	14.81	8.35
IDML207	14	11	7	2.6	0.63	0.1297	9.16	102.9	19.48	8.71
IDML210	14	11	7	2.6	0.63	0.0756	10.22	96.1	11.14	7.90
IDMLK05	N/A	N/A	N/A	N/A	N/A	**0.100**	N/A	N/A	N/A	**8.00**
IDMLK07	N/A	N/A	N/A	N/A	N/A	**0.100**	N/A	N/A	N/A	**8.00**
IDMLK10	N/A	N/A	N/A	N/A	N/A	0.096	N/A	N/A	N/A	8.15
IDMS105	50	3	1.9	0.95	0.23	0.1644	10	108.4	17.53	8.71
IDMS107	50	3	1.9	0.95	0.23	0.1117	9.32	111.1	22.51	8.97
IDMS110	50	3	1.9	0.95	0.23	0.0952	11.22	104.1	10.46	8.05
IDMS207	26	7	2	0.55	0.17	0.0832	9.14	109.4	22.71	8.59
IDMS210	26	7	2	0.55	0.17	0.0234	10.32	95.8	10.63	7.96
IPAL105	2.5	17	4	0.48	0.52	0.0419	9.93	94.7	12.29	7.44
IPAL107	2.5	17	4	0.48	0.52	0.0403	8	89.1	19.01	7.45
IPAL110	2.5	17	4	0.48	0.52	0.0347	9.69	95.6	13.27	6.65
IPAL205	3	15	4.6	1.6	0.47	0.0425	9.38	91.3	13.21	7.10
IPAL207	3	15	4.6	1.6	0.47	0.042	6.53	73.3	19.43	6.93
IPAL210	3	15	4.6	1.6	0.47	0.0296	10.56	103.7	13.05	6.67

Table C-3. Field and physical data collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. **Bold** numbers represent estimated values. See Table C-1 for metadata (continued).

PARAMETER: **UNITS:**	**Distance from treeline** **(m)**	**Distance from lake** **(m)**	**Length** **(m)**	**Width** **(m)**	**Depth** **(m)**	**Spec Cond** **(mS/cm)**	**DO** **(mg/L)**	**DO%** **(% sat)**	**Temp** **(°C)**	**pH**
SAMPLE NR					***ISLE ROYALE NATIONAL PARK (ISRO, I)***					
IPALK05	N/A	N/A	N/A	N/A	N/A	**0.100**	N/A	N/A	N/A	**8.00**
IPALK07	N/A	N/A	N/A	N/A	N/A	**0.100**	N/A	N/A	N/A	**8.00**
IPALK10	N/A	N/A	N/A	N/A	N/A	0.097	N/A	N/A	N/A	8.05
IPAS105	22	3.5	1	0.77	0.27	0.1226	11.19	110.2	13.74	8.98
IPAS107	22	3.5	1	0.77	0.27	0.0741	8.38	94.2	19.46	8.27
IPAS110	22	3.5	1	0.77	0.27	0.083	11.49	116.4	14.48	8.35
IPAS205	25	9.5	3.9	3.54	0.6	0.1103	10.08	100.8	14.44	8.65
IPAS207	25	9.5	3.9	3.54	0.6	0.0953	8.78	101.8	21.08	8.64
IPAS210	25	9.5	3.9	3.54	0.6	0.0888	10.51	107.7	14.99	8.18
IRSL105	4.5	25	1.25	1.15	0.28	0.0395	10.73	119.8	19.72	9.01
IRSL107	4.5	25	1.25	1.15	0.28	0.0451	7.78	81.4	15.98	7.48
IRSL110	4.5	25	1.25	1.15	0.28	0.0335	11.12	114	15.16	7.56
IRSL205	14	24	3.2	1.4	0.28	0.0433	11.59	129.9	19.92	8.87
IRSL207	14	24	3.2	1.4	0.28	0.0304	8.52	90.4	16.65	7.59
IRSL210	14	24	3.2	1.4	0.28	0.0177	10.27	113.1	18.38	7.25
IRSLK05	N/A	N/A	N/A	N/A	N/A	**0.100**	N/A	N/A	N/A	**8.00**
IRSLK07	N/A	N/A	N/A	N/A	N/A	**0.100**	N/A	N/A	N/A	**8.00**
IRSLK10	N/A	N/A	N/A	N/A	N/A	0.096	N/A	N/A	N/A	7.98
IRSS105	25	5	10	2.72	0.63	0.1007	11.06	122.1	19.12	8.54
IRSS107	25	5	10	2.72	0.63	0.0968	12.06	111	10.23	7.99
IRSS110	25	5	10	2.72	0.63	0.0944	11.07	111.8	14.04	7.93
IRSS205	24	4	6.8	2.55	0.39	0.1047	10.06	115.4	21.06	8.61
IRSS207	24	4	6.8	2.55	0.39	0.0968	11.84	110.2	10.7	8.08
IRSS210	24	4	6.8	2.55	0.39	0.0899	11.14	115	15.18	8.00
					PICTURED ROCKS NATIONAL LAKESHORE (PIRO, P)					
PASL105	0.2	7	2.23	1.08	0.05	0.039	9.12	114.5	27.07	8.36
PASL108	0.1	4.3	6.93	1.35	0.053	0.017	5.99	67.9	24.34	7.33
PASL205	1.5	7	5.5	3.5	0.095	0.039	9.53	111.1	22.92	7.85

Table C-3. Field and physical data collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. **Bold** numbers represent estimated values. See Table C-1 for metadata (continued).

PARAMETER: **UNITS:**	**Distance from treeline (m)**	**Distance from lake (m)**	**Length (m)**	**Width (m)**	**Depth (m)**	**Spec Cond (mS/cm)**	**DO (mg/L)**	**DO% (% sat)**	**Temp (°C)**	**pH**
SAMPLE NR	***PICTURED ROCKS NATIONAL LAKESHORE (PIRO, P)***									
PASL208	1.5	3.98	3.46	1.4	0.063	0.019	5.98	70.2	24.83	6.87
PASLK05	N/A	N/A	N/A	N/A	N/A	0.100	N/A	N/A	N/A	7.89
PASLK08	N/A	N/A	N/A	N/A	N/A	0.099	N/A	N/A	N/A	7.82
PASS105	15	1	3.82	1.27	0.082	0.093	8.12	95.2	23.24	7.89
PASS205	10.5	5.1	4.48	0.57	0.087	0.081	7.28	80.1	19.73	7.90
PMBL105	0.75	11	10.5	9	0.09	0.378	11.44	114.3	15.32	8.28
PMBL108	0.5	12	6.83	6.25	0.063	0.378	11.44	114.3	15.32	8.28
PMBL208	0.1	9	0.5	0.071	0.089	0.384	8.54	84.3	15.88	8.21
PMBLK05	N/A	N/A	N/A	N/A	N/A	0.105	N/A	N/A	N/A	7.88
PMBLZ05	0.1	9	10.25	5	0.105	0.399	11.96	103.1	8.56	7.85
PMBLZ08	0.1	10	12	6	0.1	0.342	9.64	96	15.72	8.42
PMBS205	9.5	6.5	5	3.9	0.075	0.381	11.8	102.2	8.69	7.87
PMHCA05	8	14	3.3	1.53	0.06	0.105	13.99	119.5	8.43	8.83
PMHCA08	8	13	3.95	2.29	0.007	0.103	8.66	89.4	18.5	8.40
PMHLK05	N/A	N/A	N/A	N/A	N/A	0.104	N/A	N/A	N/A	8.07
PMHLK08	N/A	N/A	N/A	N/A	N/A	0.109	N/A	N/A	N/A	8.31
PMHMZ05	9	8	11	2.1	0.01	0.143	10.04	95.5	12.74	7.74
PMHMZ08	9	8	9.3	2	0.001	0.059	7.8	78.3	19.5	8.28
PMHS105	14.5	22	17	4.5	9.4	0.172	9.1	106.6	23.14	8.10
PMHS108	15	22	20.2	5.79	0.037	0.139	9.22	112.5	23.88	8.50

Table C-4. Total and dissolved nutrients in water samples collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata.

PARAMETER:	TP	TN	DOC	DIC	NO-X	NH4	SRP	Chl-a		
UNITS:	µg P/L	mg N/L	ppm	mg C/L	mg N/L	mg N/L	µg P/L	µg/L	DIN:TP	TN:TP
SAMPLE NR	***APOSTLE ISLANDS NATIONAL LAKESHORE (APIS, A)***									
ABIL105	6.78	0.43	3.43	10.20	0.194	0.07	0.676	0.933	39.4	64.0
ABIL109	31.46	0.71	10.47	4.77	0.005	0.07	6.363	4.396	2.5	22.7
ABIL205	2.53	0.40	1.88	9.99	0.302	0.06	0.842	0.686	141.6	157.4
ABIL209	5.93	0.29	3.17	5.29	0.010	0.07	1.068	0.154	13.6	49.7
ABILK05	2.66	0.44	2.33	9.98	0.350	0.07	3.472	0.810	157.5	164.3
ABILK09	4.24	0.45	1.76	10.28	0.360	0.06	2.675	0.930	99.9	106.6
ABIS105	2.91	0.44	1.98	9.93	0.331	0.05	1.338	0.496	130.5	150.1
ABIS109	6.09	0.37	2.58	10.10	0.201	0.06	2.733	0.859	43.2	61.1
ABIS205	5.78	0.41	2.99	9.84	0.305	0.05	0.624	0.888	61.7	71.4
ABIS209	12.56	0.43	6.12	7.08	0.022	0.09	2.620	1.028	8.7	34.6
ADIL105	12.02	0.27	9.12	1.62	0.000	0.07	3.421	0.146	5.8	22.3
ADIL109	43.70	0.71	11.93	0.88	0.000	0.07	5.994	9.920	1.5	16.2
ADIL205	17.76	0.58	7.71	4.99	0.190	0.07	3.009	7.684	14.4	32.9
ADIL209	13.43	0.32	6.98	4.53	0.059	0.07	3.150	1.529	9.6	23.5
ADILK05	2.82	0.43	1.75	10.07	0.345	0.06	0.958	0.961	142.6	153.2
ADILK09	6.09	0.45	5.79	10.31	0.357	0.06	0.959	0.762	68.5	73.2
ADIS105	16.51	0.54	8.92	2.96	0.011	0.08	3.362	2.296	5.8	32.5
ADIS109	11.34	0.30	5.78	4.72	0.021	0.06	4.614	0.461	7.3	26.7
ADIS205	4.50	0.33	5.48	5.81	0.158	0.06	0.477	0.367	48.4	74.4
ADIS209	3.238	0.36	2.95	8.46	0.207	0.04	1.011	0.525	77.7	112.3
	ISLE ROYALE NATIONAL PARK (ISRO, I)									
IBLL105	12.047	0.65	27.26	5.12	0.002	0.12	2.044	0.277	10.2	53.8
IBLL107	10.987	0.87	32.14	8.28	0.001	0.13	1.676	2.495	11.8	79.5
IBLL110	11.008	0.69	29.43	5.90	0.003	0.11	2.104	0.302	10.0	62.3
IBLL205	13.569	0.58	23.83	2.67	0.002	0.12	1.645	0.977	8.7	42.5
IBLL207	25.432	0.90	21.71	2.05	0.001	0.11	1.606	11.487	4.3	35.5
IBLL210	9.937	0.55	23.84	2.16	0.004	0.10	2.312	0.933	10.9	55.6
IBLLK05	1.567	0.44	5.26	9.61	0.376	0.07	2.480	0.520	286.7	283.3
IBLLK07	1.748	0.41	3.07	9.77	0.354	0.10	1.649	0.346	259.3	235.6
IBLLK10	3.505	0.42	4.72	10.00	0.343	0.08	2.296	0.717	121.9	120.2

Table C-4. Total and dissolved nutrients in water samples collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	TP	TN	DOC	DIC	NO-X	NH4	SRP	Chl-a		
UNITS:	µg P/L	mg N/L	ppm	mg C/L	mg N/L	mg N/L	µg P/L	µg/L	DIN:TP	TN:TP
SAMPLE NR				***ISLE ROYALE NATIONAL PARK (ISRO, I)***						
IBLS105	4.452	0.32	12.95	6.86	0.084	0.08	-0.002	0.000	37.8	72.6
IBLS107	4.415	0.28	5.39	10.13	0.052	0.09	1.895	0.382	32.4	64.3
IBLS110	2.306	0.38	4.97	9.78	0.270	0.08	2.181	0.570	153.9	163.1
IBLS205	1.719	0.39	4.39	9.70	0.271	0.10	1.566	1.048	215.6	224.6
IBLS207	6.664	0.49	7.12	8.85	0.007	0.10	1.657	3.910	15.5	74.1
IBLS210	2.847	0.24	4.50	8.65	0.108	0.10	2.027	0.406	73.7	85.8
IDML105	8.235	0.40	7.48	1.32	0.004	0.11	1.568	0.967	13.6	48.0
IDML107	13.907	0.80	11.24	1.82	0.003	0.11	2.664	5.180	8.4	57.7
IDML110	4.620	0.23	6.95	1.19	0.007	0.08	1.023	0.231	18.4	49.5
IDML205	9.329	0.48	14.66	5.94	0.006	0.13	2.052	0.294	14.2	51.9
IDML207	50.176	0.57	15.23	14.68	0.002	0.12	1.707	0.337	2.4	11.4
IDML210	4.786	0.35	11.84	12.12	0.026	0.11	2.089	0.162	28.6	73.6
IDMLK05	2.053	0.39	12.32	9.46	0.370	0.10	1.163	0.323	226.7	191.0
IDMLK07	5.147	0.44	2.97	9.76	0.354	0.10	0.920	0.499	88.9	85.6
IDMLK10	2.669	0.37	2.96	9.85	0.338	0.08	0.226	0.867	154.8	139.1
IDMS105	2.093	0.39	7.90	9.84	0.303	0.08	2.169	0.244	184.5	185.1
IDMS107	8.361	0.36	5.93	10.76	0.039	0.09	1.819	4.016	15.0	43.3
IDMS110	1.863	0.23	3.42	10.27	0.286	0.10	1.135	0.667	205.8	122.9
IDMS207	28.832	1.15	13.13	8.50	0.006	0.13	1.745	1.145	4.6	40.0
IDMS210	2.206	0.40	5.04	2.92	0.007	0.08	0.603	0.243	39.6	183.4
IPAL105	10.563	1.16	21.99	3.20	0.583	0.12	2.575	0.327	66.6	109.6
IPAL107	13.765	0.76	25.64	3.41	0.006	0.13	2.535	0.439	10.1	55.5
IPAL110	13.730	0.65	29.30	3.80	0.008	0.11	2.739	0.320	8.7	47.1
IPAL205	10.887	0.61	22.92	3.37	0.014	0.12	2.541	0.184	11.9	56.0
IPAL207	16.341	0.87	27.12	4.05	0.003	0.13	2.803	0.677	8.2	53.5
IPAL210	11.095	0.55	27.12	3.20	0.002	0.11	2.269	0.313	9.9	50.0
IPALK05	2.477	0.44	3.04	9.71	0.357	0.09	1.711	0.433	181.1	176.5
IPALK07	3.206	0.44	2.97	9.82	0.345	0.10	1.784	0.483	139.7	137.2
IPALK10	2.742	0.43	4.82	9.87	0.337	0.09	4.216	0.845	154.4	156.3
SAMPLE NR				***ISLE ROYALE NATIONAL PARK (ISRO, I)***						
IPAS105	4.284	0.39	3.26	10.10	0.108	0.09	1.964	0.195	46.2	90.2

Table C-4. Total and dissolved nutrients in water samples collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	**TP**	**TN**	**DOC**	**DIC**	**NO-X**	**NH4**	**SRP**	**Chl-a**		
UNITS:	**µg P/L**	**mg N/L**	**ppm**	**mg C/L**	**mg N/L**	**mg N/L**	**µg P/L**	**µg/L**	**DIN:TP**	**TN:TP**
IPAS107	9.711	0.69	7.32	7.73	0.009	0.10	1.596	1.161	11.7	71.0
IPAS110	2.454	0.29	4.80	8.66	0.160	0.08	0.615	0.363	98.1	116.4
IPAS205	1.958	0.39	3.85	10.01	0.189	0.08	1.777	0.145	137.1	197.7
IPAS207	5.045	0.39	6.18	9.38	0.021	0.10	2.293	0.338	24.4	77.1
IPAS210	2.854	0.34	3.71	9.26	0.263	0.08	2.061	0.404	121.2	119.6
IRSL105	10.849	0.53	6.41	3.42	0.014	0.11	2.182	4.263	11.0	48.7
IRSL107	29.519	1.29	16.42	5.22	0.022	0.17	3.731	1.055	6.6	43.6
IRSL110	9.057	0.35	9.22	4.23	0.004	0.11	2.834	1.543	12.5	38.8
IRSL205	12.512	0.73	7.85	3.34	0.007	0.11	3.135	1.465	9.2	58.1
IRSL207	30.527	1.13	14.11	3.37	0.006	0.12	2.036	2.215	4.3	37.0
IRSL210	8.826	0.39	8.89	2.24	0.005	0.09	1.919	2.259	10.6	44.1
IRSLK05	2.151	0.43	3.80	9.71	0.371	0.10	1.821	0.611	219.8	200.1
IRSLK07	4.728	0.49	3.51	9.90	0.353	0.09	2.143	0.460	94.1	102.7
IRSLK10	1.804	0.43	3.11	9.93	0.360	0.10	2.500	0.709	253.8	238.9
IRSS105	1.882	0.41	4.94	9.54	0.152	0.10	1.580	0.451	131.7	215.6
IRSS107	2.007	0.42	3.99	9.86	0.346	0.09	2.467	0.347	218.7	210.6
IRSS110	2.745	0.41	3.49	9.84	0.345	0.09	2.577	0.871	157.0	148.5
IRSS205	2.556	0.42	4.44	10.65	0.130	0.12	1.873	0.468	98.5	165.3
IRSS207	4.600	0.42	4.77	9.87	0.349	0.08	1.087	0.534	93.4	91.8
IRSS210	1.701	0.42	7.47	9.94	0.352	0.10	2.454	0.886	266.2	245.8
				PICTURED ROCKS NATIONAL LAKESHORE (PIRO, P)						
PASL105	78.74	1.10	22.38	3.05	0.000	0.08	9.920	10.190	1.0	14.0
PASL108	210.28	3.04	20.34	2.43	0.000	0.11	7.103	123.840	0.5	14.5
PASL205	18.93	0.54	23.77	2.75	0.000	0.08	4.501	1.594	4.0	28.3
PASL208	2060.73	5.13	22.08	3.22	0.000	0.10	44.860	393.760	0.0	2.5
PASLK05	7.50	0.46	3.86	10.11	0.336	0.05	0.407	0.940	52.1	61.7
PASLK08	3.99	0.44	1.75	10.17	0.309	0.08	2.415	0.599	97.4	111.0
PASS105	12.98	0.83	29.26	8.43	0.005	0.09	3.789	0.119	7.3	64.1
PASS205	22.79	1.06	30.28	7.68	0.007	0.09	10.063	0.379	4.5	46.6
SAMPLE NR				***PICTURED ROCKS NATIONAL LAKESHORE (PIRO, P)***						
PMBL105	9.58	0.31	7.52	38.89	0.000	0.06	4.039	1.182	6.2	32.6
PMBL108	14.25	0.53	9.61	35.43	0.016	0.10	1.768	2.241	7.9	37.5
PMBL208	8.46	0.20	5.38	41.06	0.003	0.07	2.132	5.816	9.1	23.7

Table C-4. Total and dissolved nutrients in water samples collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	TP	TN	DOC	DIC	NO-X	NH4	SRP	Chl-a		
UNITS:	µg P/L	mg N/L	ppm	mg C/L	mg N/L	mg N/L	µg P/L	µg/L	DIN:TP	TN:TP
PMBLK05	4.33	0.47	4.48	10.68	0.338	0.08	3.787	0.993	96.6	109.3
PMBLZ05	26.31	0.24	5.20	50.62	0.000	0.02	3.505	1.626	0.8	9.0
PMBLZ08	51.84	0.88	6.07	37.67	0.010	0.09	0.269	3.464	1.9	17.0
PMBS205	7.91	0.30	13.03	47.15	0.000	0.06	3.717	0.337	7.7	37.6
PMHCA05	7.15	0.25	3.50	10.00	0.000	0.09	3.302	0.368	12.8	34.8
PMHCA08	29.26	0.91	3.64	10.37	0.020	0.12	5.203	4.772	4.6	31.1
PMHLK05	33.70	0.66	1.72	10.44	0.352	0.07	3.988	1.446	12.4	19.5
PMHLK08	4.57	0.44	2.59	10.31	0.300	0.07	1.857	0.555	82.1	96.0
PMHMZ05	4.45	0.21	7.14	13.25	0.000	0.05	1.141	0.058	10.8	47.6
PMHMZ08	4.61	0.28	9.76	16.49	0.012	0.08	2.083	0.227	20.2	60.1
PMHS105	8.22	0.29	9.95	17.73	0.003	0.07	3.606	0.059	8.7	34.9
PMHS108	9.38	0.54	14.60	12.91	0.006	0.10	0.086	0.609	11.1	57.5

Table C-5. Anions in water samples collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. **Bold** values represent analysis detection limits. See Table C-1 for metadata.

PARAMETER:	Fluoride	Lactate	Acetate	Formate	Chloride	Nitrite - N	Bromide	Nitrate - N	Sulfate	Oxalate	Thiosulfate	Phosphate - P
UNITS:	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g
Detection Limit:	0.002	0.005	0.005	0.005	0.010	0.001	0.005	0.001	0.050	0.010	0.010	0.010
SAMPLE NR					***APOSTLE ISLANDS NATIONAL LAKESHORE (APIS, A)***							
ABIL105	0.036	**0.005**	**0.005**	**0.005**	1.625	**0.001**	**0.005**	0.192	3.831	**0.010**	**0.010**	0.023
ABIL109	0.018	**0.005**	**0.005**	**0.005**	0.417	**0.001**	**0.005**	**0.001**	0.628	**0.010**	**0.010**	0.018
ABIL205	0.032	**0.005**	**0.005**	**0.005**	1.567	**0.001**	**0.005**	0.300	3.934	**0.010**	**0.010**	0.024
ABIL209	0.032	**0.005**	**0.005**	**0.005**	1.023	**0.001**	**0.005**	0.006	2.341	**0.010**	**0.010**	0.032
ABILK05	0.035	**0.005**	**0.005**	**0.005**	1.668	**0.001**	**0.005**	0.283	3.942	**0.010**	**0.010**	0.028
ABILK09	0.034	**0.005**	**0.005**	**0.005**	1.614	**0.001**	**0.005**	0.362	3.922	**0.010**	**0.010**	0.022
ABIS105	0.031	**0.005**	**0.005**	**0.005**	1.628	**0.001**	**0.005**	0.334	3.832	**0.010**	**0.010**	0.064
ABIS109	0.034	**0.005**	**0.005**	**0.005**	1.357	**0.001**	**0.005**	0.110	3.114	**0.010**	**0.010**	0.034
ABIS205	0.033	**0.005**	**0.005**	**0.005**	1.631	0.007	**0.005**	0.307	3.826	**0.010**	**0.010**	0.030
ABIS209	0.032	**0.005**	**0.005**	**0.005**	1.379	**0.001**	**0.005**	0.019	3.323	**0.010**	**0.010**	0.016
ADIL105	0.039	**0.005**	**0.005**	**0.005**	0.624	**0.001**	**0.005**	**0.001**	6.079	**0.010**	**0.010**	0.019
ADIL109	0.034	**0.005**	**0.005**	**0.005**	0.644	**0.001**	**0.005**	**0.001**	4.745	**0.010**	**0.010**	**0.010**
ADIL205	0.032	**0.005**	**0.005**	**0.005**	0.705	**0.001**	**0.005**	**0.001**	5.863	**0.010**	**0.010**	0.025
ADIL209	0.027	**0.005**	**0.005**	**0.005**	0.466	**0.001**	**0.005**	0.062	5.508	**0.010**	**0.010**	0.013
ADILK05	0.038	**0.005**	**0.005**	**0.005**	1.560	**0.001**	**0.005**	0.383	3.783	**0.010**	**0.010**	0.030
ADILK09	0.034	**0.005**	**0.005**	**0.005**	1.604	**0.001**	**0.005**	0.447	3.825	**0.010**	**0.010**	0.033
ADIS105	0.040	**0.005**	**0.005**	**0.005**	1.146	**0.001**	**0.005**	0.013	6.647	**0.010**	**0.010**	0.017
ADIS109	0.018	**0.005**	**0.005**	**0.005**	0.896	**0.001**	**0.005**	**0.001**	2.931	**0.010**	**0.010**	0.033
ADIS205	0.033	**0.005**	**0.005**	**0.005**	1.089	**0.001**	**0.005**	0.165	5.025	**0.010**	**0.010**	0.014
ADIS209	0.030	**0.005**	**0.005**	**0.005**	1.409	**0.001**	**0.005**	0.253	4.215	**0.010**	**0.010**	0.037
					ISLE ROYALE NATIONAL PARK (ISRO, I)							
IBLL105	0.031	**0.005**	**0.005**	**0.005**	0.299	**0.001**	**0.005**	**0.001**	2.478	**0.010**	**0.010**	0.033
IBLL107	0.041	**0.005**	**0.005**	**0.005**	0.096	**0.001**	**0.005**	0.030	1.608	**0.010**	**0.010**	0.019
IBLL110	0.038	**0.005**	**0.005**	**0.005**	0.273	**0.001**	**0.005**	**0.001**	2.713	**0.010**	**0.010**	0.017
IBLL205	0.024	**0.005**	**0.005**	**0.005**	0.224	**0.001**	**0.005**	**0.001**	1.985	**0.010**	**0.010**	**0.010**
IBLL207	0.026	**0.005**	**0.005**	**0.005**	0.160	**0.001**	**0.005**	**0.001**	1.343	**0.010**	**0.010**	0.013
IBLL210	0.027	**0.005**	**0.005**	**0.005**	0.259	**0.001**	**0.005**	**0.001**	1.458	**0.010**	**0.010**	**0.010**
IBLLK05	0.035	**0.005**	**0.005**	**0.005**	1.539	**0.001**	**0.005**	0.256	3.750	**0.010**	**0.010**	0.047
IBLLK07	0.033	**0.005**	**0.005**	**0.005**	1.502	**0.001**	**0.005**	0.378	3.624	**0.010**	**0.010**	0.030

Table C-5. Anions in water samples collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. **Bold** values represent analysis detection limits. See Table C-1 for metadata (continued).

PARAMETER:	Fluoride	Lactate	Acetate	Formate	Chloride	Nitrite - N	Bromide	Nitrate - N	Sulfate	Oxalate	Thiosulfate	Phosphate - P
UNITS:	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g	ug/g
Detection Limit:	0.002	0.005	0.005	0.005	0.010	0.001	0.005	0.001	0.050	0.010	0.010	0.010
SAMPLE NR	***ISLE ROYALE NATIONAL PARK (ISRO, I)***											
IBLLK10	0.031	**0.005**	**0.005**	**0.005**	1.446	**0.001**	**0.005**	0.389	3.576	**0.010**	**0.010**	0.042
IBLS105	0.031	**0.005**	**0.005**	**0.005**	1.191	**0.001**	**0.005**	0.089	3.606	**0.010**	**0.010**	**0.010**
IBLS107	0.036	**0.005**	**0.005**	**0.005**	1.541	**0.001**	**0.005**	0.056	3.678	**0.010**	**0.010**	0.073
IBLS110	0.032	**0.005**	**0.005**	**0.005**	1.454	**0.001**	**0.005**	0.265	3.434	**0.010**	**0.010**	0.029
IBLS205	0.033	**0.005**	**0.005**	**0.005**	1.369	**0.001**	**0.005**	0.183	3.718	**0.010**	**0.010**	0.023
IBLS207	0.034	**0.005**	**0.005**	0.077	1.480	**0.001**	**0.005**	**0.001**	3.238	**0.010**	**0.010**	0.035
IBLS210	0.032	**0.005**	**0.005**	**0.005**	1.261	**0.001**	**0.005**	0.103	3.039	**0.010**	**0.010**	0.031
IDML105	0.015	**0.005**	**0.005**	**0.005**	0.147	**0.001**	**0.005**	**0.001**	0.955	**0.010**	**0.010**	0.032
IDML107	0.023	**0.005**	**0.005**	**0.005**	0.152	**0.001**	**0.005**	0.003	0.546	**0.010**	**0.010**	0.021
IDML110	0.013	**0.005**	**0.005**	**0.005**	0.022	**0.001**	**0.005**	0.004	0.346	**0.010**	**0.010**	**0.010**
IDML205	0.013	**0.005**	**0.005**	**0.005**	0.135	**0.001**	**0.005**	**0.001**	0.991	**0.010**	**0.010**	0.029
IDML207	0.018	**0.005**	**0.005**	**0.005**	0.114	**0.001**	**0.005**	**0.001**	0.626	**0.010**	**0.010**	**0.010**
IDML210	0.015	**0.005**	**0.005**	**0.005**	0.142	**0.001**	**0.005**	0.006	0.811	**0.010**	**0.010**	0.016
IDMLK05	0.031	**0.005**	**0.005**	**0.005**	1.494	0.049	**0.005**	0.380	3.898	**0.010**	**0.010**	0.039
IDMLK07	0.031	**0.005**	**0.005**	**0.005**	1.503	**0.001**	**0.005**	0.356	3.716	**0.010**	**0.010**	0.036
IDMLK10	0.030	**0.005**	**0.005**	**0.005**	1.483	0.048	**0.005**	0.325	3.627	**0.010**	**0.010**	0.065
IDMS105	0.035	**0.005**	**0.005**	**0.005**	1.553	0.006	**0.005**	0.291	3.993	**0.010**	**0.010**	0.050
IDMS107	0.041	**0.005**	**0.005**	**0.005**	1.726	**0.001**	**0.005**	0.036	4.024	**0.010**	**0.010**	0.032
IDMS110	0.031	**0.005**	**0.005**	**0.005**	1.551	**0.001**	**0.005**	0.284	3.732	**0.010**	**0.010**	0.021
IDMS205	0.038	**0.005**	**0.005**	**0.005**	1.543	**0.001**	**0.005**	0.244	3.864	**0.010**	**0.010**	0.011
IDMS207	0.032	**0.005**	**0.005**	**0.005**	0.826	**0.001**	**0.005**	**0.001**	2.116	**0.010**	**0.010**	0.015
IDMS210	0.011	**0.005**	**0.005**	**0.005**	0.234	**0.001**	**0.005**	0.007	0.600	**0.010**	**0.010**	0.024
IPAL105	0.028	**0.005**	**0.005**	**0.005**	0.205	**0.001**	**0.005**	0.583	3.890	**0.010**	**0.010**	0.018
IPAL107	0.025	**0.005**	**0.005**	**0.005**	0.100	**0.001**	**0.005**	**0.001**	2.223	**0.010**	**0.010**	**0.010**
IPAL110	0.023	**0.005**	**0.005**	**0.005**	0.316	**0.001**	**0.005**	0.014	2.073	**0.010**	**0.010**	**0.010**
IPAL205	0.021	**0.005**	**0.005**	**0.005**	0.147	**0.001**	**0.005**	0.042	3.601	**0.010**	**0.010**	0.013
IPAL207	0.023	**0.005**	**0.005**	**0.005**	0.158	**0.001**	**0.005**	**0.001**	1.492	**0.010**	**0.010**	**0.010**
IPAL210	0.020	**0.005**	**0.005**	**0.005**	0.259	**0.001**	**0.005**	0.004	1.635	**0.010**	**0.010**	**0.010**
IPALK05	0.034	**0.005**	**0.005**	**0.005**	1.465	**0.001**	**0.005**	0.353	3.631	**0.010**	**0.010**	0.051

Table C-5. Anions in water samples collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. **Bold** values represent analysis detection limits. See Table C-1 for metadata (continued).

PARAMETER:	**Fluoride**	**Lactate**	**Acetate**	**Formate**	**Chloride**	**Nitrite - N**	**Bromide**	**Nitrate - N**	**Sulfate**	**Oxalate**	**Thiosulfate**	**Phosphate - P**
UNITS:	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**
Detection Limit:	**0.002**	**0.005**	**0.005**	**0.005**	**0.010**	**0.001**	**0.005**	**0.001**	**0.050**	**0.010**	**0.010**	**0.010**
SAMPLE NR					***ISLE ROYALE NATIONAL PARK (ISRO, I)***							
IPALK07	0.033	**0.005**	**0.005**	**0.005**	1.507	**0.001**	**0.005**	0.126	3.656	**0.010**	**0.010**	0.047
IPALK10	0.034	**0.005**	**0.005**	**0.005**	1.520	0.036	**0.005**	0.034	3.694	**0.010**	**0.010**	0.053
IPAS105	0.036	**0.005**	**0.005**	**0.005**	1.606	**0.001**	**0.005**	0.120	3.987	**0.010**	**0.010**	0.028
IPAS107	0.028	**0.005**	**0.005**	**0.005**	1.002	**0.001**	**0.005**	0.005	2.224	**0.010**	**0.010**	**0.010**
IPAS110	0.030	**0.005**	**0.005**	**0.005**	1.268	**0.001**	**0.005**	0.161	3.090	**0.010**	**0.010**	**0.010**
IPAS205	0.037	**0.005**	**0.005**	**0.005**	1.603	**0.001**	**0.005**	0.261	3.906	**0.010**	**0.010**	0.052
IPAS207	0.032	**0.005**	**0.005**	**0.005**	1.618	0.118	**0.005**	0.018	3.370	**0.010**	**0.010**	0.040
IPAS210	0.030	**0.005**	**0.005**	**0.005**	1.362	**0.001**	**0.005**	0.265	3.339	**0.010**	**0.010**	0.033
IRSL105	0.017	**0.005**	**0.005**	**0.005**	0.255	**0.001**	**0.005**	**0.001**	2.091	**0.010**	**0.010**	0.023
IRSL107	0.020	**0.005**	**0.005**	**0.005**	0.279	**0.001**	**0.005**	0.017	1.444	**0.010**	**0.010**	**0.010**
IRSL110	0.014	**0.005**	**0.005**	**0.005**	0.215	**0.001**	**0.005**	0.028	1.121	**0.010**	**0.010**	0.018
IRSL205	0.016	**0.005**	**0.005**	**0.005**	0.592	**0.001**	**0.005**	0.020	2.138	**0.010**	**0.010**	0.031
IRSL207	0.022	**0.005**	**0.005**	**0.005**	0.245	**0.001**	**0.005**	**0.001**	0.708	**0.010**	**0.010**	**0.010**
IRSL210	0.012	**0.005**	**0.005**	**0.005**	0.147	**0.001**	**0.005**	**0.001**	0.634	**0.010**	**0.010**	0.014
IRSLK05	0.031	**0.005**	**0.005**	**0.005**	1.509	**0.001**	**0.005**	0.373	3.791	**0.010**	**0.010**	0.054
IRSLK07	0.035	**0.005**	**0.005**	**0.005**	1.506	**0.001**	**0.005**	0.438	3.701	**0.010**	**0.010**	0.108
IRSLK10	0.031	**0.005**	**0.005**	**0.005**	1.521	**0.001**	**0.005**	0.414	3.731	**0.010**	**0.010**	0.056
IRSS105	0.035	**0.005**	**0.005**	**0.005**	1.488	**0.001**	**0.005**	0.146	3.844	**0.010**	**0.010**	0.029
IRSS107	0.032	**0.005**	**0.005**	**0.005**	1.486	**0.001**	**0.005**	0.239	3.644	**0.010**	**0.010**	0.057
IRSS110	0.036	**0.005**	**0.005**	**0.005**	1.531	**0.001**	**0.005**	0.347	3.859	**0.010**	**0.010**	**0.010**
IRSS205	0.041	**0.005**	**0.005**	**0.005**	1.598	**0.001**	**0.005**	0.137	4.027	**0.010**	**0.010**	0.061
IRSS207	0.035	**0.005**	**0.005**	**0.005**	1.440	**0.001**	**0.005**	0.335	3.543	**0.010**	**0.010**	0.027
IRSS210	0.032	**0.005**	**0.005**	**0.005**	1.542	**0.001**	**0.005**	0.341	3.725	**0.010**	**0.010**	0.044
					PICTURED ROCKS NATIONAL LAKESHORE (PIRO, P)							
PASL105	0.075	**0.005**	**0.005**	**0.005**	1.216	0.034	**0.005**	0.207	27.342	**0.010**	**0.010**	0.041
PASL108	0.025	**0.005**	**0.005**	**0.005**	0.699	**0.001**	**0.005**	0.010	3.460	**0.010**	**0.010**	0.039
PASL205	0.033	**0.005**	**0.005**	**0.005**	0.516	**0.001**	**0.005**	0.004	4.576	**0.010**	**0.010**	**0.010**
PASL208	0.021	**0.005**	**0.005**	**0.005**	0.462	**0.001**	**0.005**	**0.001**	2.442	**0.010**	**0.010**	0.023
PASLK05	0.036	**0.005**	**0.005**	**0.005**	1.555	**0.001**	**0.005**	0.302	3.795	**0.010**	**0.010**	0.040

Table C-5. Anions in water samples collected from rock pools and Lake Superior at Apostle Islands National Lakeshore, Isle Royale National Park, and Pictured Rocks National Lakeshore, 2010. **Bold** values represent analysis detection limits. See Table C-1 for metadata (continued).

PARAMETER:	Fluoride	Lactate	Acetate	Formate	Chloride	Nitrite - N	Bromide	Nitrate - N	Sulfate	Oxalate	Thiosulfate	Phosphate - P
UNITS:	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**	**ug/g**
Detection Limit:	**0.002**	**0.005**	**0.005**	**0.005**	**0.010**	**0.001**	**0.005**	**0.001**	**0.050**	**0.010**	**0.010**	**0.010**
SAMPLE NR	***PICTURED ROCKS NATIONAL LAKESHORE (PIRO, P)***											
PASLK08	0.031	**0.005**	**0.005**	**0.005**	1.706	**0.001**	**0.005**	0.324	3.611	**0.010**	**0.010**	0.047
PASS105	0.035	**0.005**	**0.005**	**0.005**	0.586	**0.001**	**0.005**	0.008	3.918	**0.010**	**0.010**	0.014
PASS205	0.034	**0.005**	**0.005**	**0.005**	1.031	**0.001**	**0.005**	0.100	3.002	**0.010**	**0.010**	0.023
PMBL105	0.115	**0.005**	**0.005**	**0.005**	0.745	**0.001**	**0.005**	**0.001**	50.544	**0.010**	**0.010**	**0.010**
PMBL108	0.088	**0.005**	**0.005**	**0.005**	0.603	**0.001**	**0.005**	0.037	34.660	**0.010**	**0.010**	0.021
PMBL208	0.058	**0.005**	**0.005**	**0.005**	0.543	**0.001**	**0.005**	**0.001**	30.386	**0.010**	**0.010**	0.071
PMBLK05	0.034	**0.005**	**0.005**	**0.005**	1.687	0.066	**0.005**	0.412	4.139	**0.010**	**0.010**	0.071
PMBLZ05	0.054	**0.005**	**0.005**	**0.005**	0.740	**0.001**	**0.005**	0.041	22.711	**0.010**	**0.010**	0.024
PMBLZ08	0.047	**0.005**	**0.005**	**0.005**	0.830	**0.001**	**0.005**	0.077	14.210	**0.010**	**0.010**	0.024
PMBS205	0.057	**0.005**	**0.005**	**0.005**	0.776	**0.001**	**0.005**	**0.001**	22.857	**0.010**	**0.010**	0.027
PMHCA05	0.039	**0.005**	**0.005**	**0.005**	1.844	**0.001**	**0.005**	**0.001**	4.046	**0.010**	**0.010**	0.028
PMHCA08	0.036	**0.005**	**0.005**	**0.005**	3.602	0.042	**0.005**	0.045	5.230	**0.010**	**0.010**	0.017
PMHLK05	0.034	**0.005**	**0.005**	**0.005**	1.581	**0.001**	**0.005**	0.342	3.832	**0.010**	**0.010**	0.062
PMHLK08	0.033	**0.005**	**0.005**	**0.005**	1.558	**0.001**	**0.005**	0.302	3.834	**0.010**	**0.010**	0.032
PMHMZ05	0.064	**0.005**	**0.005**	**0.005**	0.413	**0.001**	**0.005**	**0.001**	12.320	**0.010**	**0.010**	0.035
PMHMZ08	0.068	**0.005**	**0.005**	**0.005**	0.301	**0.001**	**0.005**	0.009	12.373	**0.010**	**0.010**	0.042
PMHS105	0.043	**0.005**	**0.005**	**0.005**	1.313	0.015	**0.005**	0.020	19.692	**0.010**	**0.010**	0.025
PMHS108	0.042	**0.005**	**0.005**	**0.005**	0.925	**0.001**	**0.005**	0.004	13.866	**0.010**	**0.010**	0.025

Table C-6. Cations in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata.

PARAMETER:	Al1670	Ba4554	Ca3158	Fe2382	K_7664	Li6707	Mg2852	Mn2576	Na5895	P_1774	Si2516	Sr4215
UNITS:	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g
SAMPLE NR					***APOSTLE ISLANDS NATIONAL LAKESHORE (APIS, A)***							
ABIL105	1.56	10.85	14175.0	2.36	552.85	1.09	2873.0	0.41	1657.5	0.78	888.15	23.95
ABIL109	18.19	6.92	6931.5	61.97	643.95	-0.78	1413.5	21.22	543.5	6.86	1048.00	11.61
ABIL205	4.03	10.21	13730.0	1.79	525.80	6.26	2819.0	0.10	1570.5	2.06	982.80	22.52
ABIL209	16.17	5.02	7648.0	8.22	469.80	-0.61	1572.0	0.84	1056.0	0.78	960.90	12.51
ABILK05	1.18	10.35	13760.0	2.64	743.80	3.72	2838.0	2.10	1830.5	4.48	989.15	23.25
ABILK09	5.65	11.70	13885.0	11.84	567.35	-0.07	2858.0	1.85	1647.0	5.82	1023.00	22.68
ABIS105	-0.74	10.05	13670.0	4.07	556.60	3.67	2791.5	-0.07	1595.0	1.29	1006.50	22.21
ABIS109	-0.35	9.84	13555.0	1.70	547.15	4.11	2798.5	-0.04	1586.0	-0.21	882.95	22.28
ABIS205	46.20	7.67	10500.0	12.05	813.25	1.64	2233.0	0.76	1360.5	6.49	845.70	17.63
ABIS209	4.44	9.87	13595.0	4.63	510.20	0.81	2852.5	0.26	1695.5	3.85	1074.00	22.54
ADIL105	761.75	24.97	1400.5	62.84	417.55	2.93	485.2	65.74	896.8	5.59	4882.50	8.78
ADIL109	504.85	21.62	1494.0	26.80	324.30	-1.37	452.3	74.76	1047.5	9.93	5030.50	8.34
ADIL205	615.40	26.06	1589.0	43.18	520.00	4.05	548.2	59.84	1059.0	4.91	4837.00	10.18
ADIL209	476.75	23.86	1437.5	61.76	356.05	-4.08	567.9	71.86	1085.5	7.99	5581.00	9.17
ADILK05	-2.25	10.97	13790.0	1.16	541.25	3.16	2812.5	0.29	1549.5	-1.17	990.40	22.39
ADILK09	3.77	10.08	13640.0	2.92	548.60	0.80	2827.0	0.16	1619.5	3.85	984.15	22.46
ADIS105	195.25	13.75	5611.5	17.69	479.95	0.26	1182.5	1.33	1271.5	7.58	2162.00	13.84
ADIS109	24.57	7.44	6301.0	20.80	355.25	0.49	1376.5	0.74	1002.0	7.60	414.60	12.66
ADIS205	323.40	19.85	7951.0	20.44	499.15	0.93	1732.5	29.48	1225.5	0.45	2701.50	16.45
ADIS209	90.59	13.63	11235.0	10.49	516.45	1.04	2358.5	7.92	1466.0	3.98	1750.50	20.02
						ISLE ROYALE NATIONAL PARK (ISRO, I)						
IBLL105	516.90	5.96	8693.0	115.15	68.14	0.16	2533.0	1.06	634.7	8.39	2295.50	11.82
IBLL107	417.15	1.52	14565.0	67.35	23.56	-3.81	3689.5	3.77	786.4	8.63	2051.00	16.91
IBLL110	596.85	6.13	10720.0	103.80	40.99	-1.38	3113.0	2.11	735.9	8.74	3705.50	13.85
IBLL205	590.65	2.64	3406.5	119.95	259.25	-0.81	1410.0	2.81	543.6	9.92	2004.50	7.45
IBLL207	349.00	1.68	3763.0	64.07	47.05	-5.53	1475.0	4.94	382.0	6.83	849.00	7.30
IBLL210	619.25	2.31	4237.0	124.40	99.22	3.14	1654.0	2.02	575.3	6.97	2559.50	8.66
IBLLK05	4.59	10.10	13830.0	2.37	575.45	-0.57	2793.5	0.16	1538.5	4.57	1047.00	22.19
IBLLK07	1.60	10.76	13815.0	0.75	549.50	-1.92	2786.5	0.21	1517.0	2.37	1018.00	22.28
IBLLK10	1.42	9.96	13510.0	-0.84	534.95	-1.65	2766.0	0.07	1507.0	1.82	968.20	22.06
IBLS105	231.80	8.07	10585.0	45.61	499.15	0.99	2304.5	1.17	1276.0	3.04	1320.50	18.53

Table C-6. Cations in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	**Al1670**	**Ba4554**	**Ca3158**	**Fe2382**	**K_7664**	**Li6707**	**Mg2852**	**Mn2576**	**Na5895**	**P_1774**	**Si2516**	**Sr4215**
UNITS:	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**
SAMPLE NR					***ISLE ROYALE NATIONAL PARK (ISRO, I)***							
IBLS107	26.87	10.61	14825.0	-0.68	615.20	2.05	2971.5	0.16	1620.0	1.42	940.60	24.87
IBLS110	42.62	9.21	13680.0	4.10	542.15	-2.54	2830.5	0.26	1513.5	-0.38	1006.00	22.67
IBLS205	4.83	10.11	13880.0	2.31	568.95	-0.80	2773.5	0.05	1540.5	3.37	984.75	22.56
IBLS207	3.75	10.45	12805.0	0.70	585.70	1.61	2441.0	0.20	1482.0	4.98	415.45	20.84
IBLS210	6.84	7.68	12145.0	1.44	512.10	-1.01	2444.0	0.13	1368.0	3.09	669.00	19.26
IDML105	78.16	6.47	2192.0	19.38	146.25	1.29	283.0	0.85	221.1	7.21	347.30	2.36
IDML107	117.35	4.76	3448.5	37.79	122.90	-0.32	352.6	2.12	259.3	9.65	604.50	3.36
IDML110	79.14	1.06	2515.0	20.58	59.63	-0.76	234.5	0.61	189.1	2.68	591.75	2.27
IDML205	82.93	28.20	10415.0	24.04	92.41	-0.11	826.7	0.63	353.7	7.83	993.95	6.86
IDML207	57.42	58.35	24915.0	30.64	106.30	2.21	1653.5	1.20	588.7	5.45	304.00	16.04
IDML210	59.02	52.22	19530.0	19.45	80.31	2.65	1384.0	0.44	550.6	4.06	1311.50	12.15
IDMLK05	4.45	9.82	13600.0	1.78	571.70	1.58	2776.5	-0.14	1510.0	1.41	1055.00	22.10
IDMLK07	2.33	9.86	13895.0	1.27	535.10	5.02	2774.0	0.36	1501.0	-1.07	1030.50	22.14
IDMLK10	2.67	9.74	13515.0	-0.21	544.00	-1.32	2746.0	0.12	1511.0	2.90	947.50	22.00
IDMS105	3.62	10.98	14215.0	1.27	589.95	-0.18	2849.0	0.06	1573.0	7.68	1032.00	23.23
IDMS107	6.84	11.79	15520.0	1.69	665.35	2.15	3083.0	0.33	1731.5	0.73	1234.50	25.18
IDMS110	2.26	10.62	14150.0	0.13	566.50	-1.38	2873.5	0.25	1557.0	5.03	959.35	22.82
IDMS205	10.36	12.14	13895.0	0.86	572.30	6.82	2749.5	-0.16	1538.0	1.72	1071.50	22.59
IDMS207	42.66	11.35	12690.0	2.59	500.95	4.44	2032.0	0.19	1082.5	6.40	977.45	19.52
IDMS210	25.10	3.99	4366.0	6.41	164.85	-6.45	746.1	0.03	358.5	0.79	426.15	6.54
IPAL105	630.65	4.17	4526.0	112.75	65.39	-0.20	2060.5	2.25	1522.0	5.87	8248.00	18.18
IPAL107	884.60	2.55	4259.5	139.50	106.70	3.61	1931.5	4.56	1464.0	3.71	9855.50	17.34
IPAL110	611.35	2.27	4385.0	105.45	65.50	1.34	1848.5	1.36	1446.5	4.97	6659.00	16.86
IPAL205	737.50	1.84	3756.5	126.30	83.08	-1.73	1614.0	3.10	1215.5	4.21	7026.00	14.71
IPAL207	0.06	9.63	13825.0	1.90	562.35	0.36	2793.5	-0.23	1532.0	5.33	1010.50	22.24
IPAL210	1.15	9.63	13675.0	0.60	542.45	-2.99	2787.0	-0.15	1515.0	0.89	984.00	22.11
IPALK05	11.43	9.96	14545.0	0.89	631.75	6.90	2856.0	-0.09	1661.0	4.63	627.15	24.66
IPALK07	4.64	7.83	11755.0	2.74	494.25	-1.94	2409.0	0.04	1366.5	0.22	622.10	20.06
IPALK10	9.54	10.25	14410.0	2.87	646.30	2.26	2779.5	-0.16	1644.5	4.65	805.25	24.28
IPAS105	2.91	8.88	12655.0	2.84	512.20	-3.64	2598.5	-0.17	1437.0	0.42	874.80	21.06
IPAS107	615.40	2.54	4614.5	82.94	99.21	4.19	1922.5	3.72	1727.0	3.80	8268.00	18.43

Table C-6. Cations in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	**Al1670**	**Ba4554**	**Ca3158**	**Fe2382**	**K_7664**	**Li6707**	**Mg2852**	**Mn2576**	**Na5895**	**P_1774**	**Si2516**	**Sr4215**
UNITS:	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**	**ng/g**
SAMPLE NR	***ISLE ROYALE NATIONAL PARK (ISRO, I)***											
IPAS110	530.40	3.32	4916.0	225.90	106.40	4.24	2030.0	9.02	1671.5	8.82	6977.00	19.02
IPAS205	2.68	9.68	13985.0	3.04	544.40	3.04	2772.5	0.30	1498.5	0.12	992.70	22.12
IPAS207	8.67	8.32	10190.0	3.59	680.95	4.34	1900.5	0.20	1529.0	5.73	112.20	18.71
IPAS210	9.32	10.34	13340.0	1.65	749.85	3.06	2440.5	0.01	1739.0	1.41	832.75	23.90
IRSL105	50.85	2.80	3938.5	11.83	148.40	-1.76	1037.0	0.48	1789.0	5.10	1129.00	16.19
IRSL107	46.77	2.90	5403.0	32.43	289.95	4.00	1357.0	1.91	2666.5	14.73	760.55	22.28
IRSL110	65.20	2.35	4728.0	18.87	129.20	-1.26	1254.0	0.41	2117.0	2.60	2704.50	20.08
IRSL205	149.40	8.63	4366.0	18.80	212.20	2.91	1089.0	0.90	1405.5	8.56	996.80	17.41
IRSL207	152.15	2.68	3687.0	55.76	245.60	-0.94	851.8	3.06	1737.0	20.15	558.40	15.38
IRSL210	114.80	1.39	2559.5	25.40	152.95	-3.57	669.9	0.81	995.6	3.64	1424.50	10.97
IRSLK05	0.78	9.90	13485.0	0.90	558.80	1.69	2748.0	-0.13	1511.0	1.62	1030.50	22.05
IRSLK07	1.32	9.75	13590.0	-0.35	547.85	0.44	2762.5	0.32	1509.5	3.67	1026.50	22.04
IRSLK10	6.64	9.54	13815.0	4.77	626.35	4.82	2801.0	0.16	1571.0	2.81	978.45	22.26
IRSS105	14.46	9.47	13655.0	3.77	586.00	1.07	2653.5	0.17	1965.5	4.13	771.65	23.61
IRSS107	10.58	9.49	12720.0	7.78	590.35	4.67	2545.0	0.27	1658.0	3.16	970.40	21.29
IRSS110	3.34	10.39	13560.0	2.96	546.30	-5.16	2798.5	-0.10	1530.0	-0.29	984.70	22.15
IRSS205	7.50	11.33	14930.0	2.33	655.70	6.30	2914.5	0.00	1878.5	4.37	975.35	25.39
IRSS207	3.01	10.20	13790.0	1.59	554.80	-1.42	2743.5	0.36	1516.5	2.92	1025.50	22.13
IRSS210	3.80	9.56	13605.0	4.28	563.05	-4.10	2776.5	-0.03	1532.0	3.28	978.60	22.09
	PICTURED ROCKS NATIONAL LAKESHORE (PIRO, P)											
PASL105	484.15	18.25	4401.5	220.30	517.25	5.60	1001.0	4.13	1166.5	17.26	4747.50	13.50
PASL108	571.40	18.17	4236.5	688.00	655.85	2.87	1018.5	29.12	1308.0	14.74	5175.50	13.14
PASL205	573.95	17.78	4692.5	200.55	629.30	0.13	1049.0	2.88	1191.0	8.61	5283.50	13.76
PASL208	484.30	20.47	4620.5	1363.00	533.60	4.56	1300.0	92.51	1115.0	16.45	4944.50	11.64
PASLK05	5.60	10.81	13810.0	4.86	621.70	-0.40	2841.0	0.78	1538.0	3.07	1010.00	22.66
PASLK08	3.14	10.36	13730.0	9.19	569.70	-1.48	2838.5	0.53	1573.5	1.37	881.85	23.07
PASS105	194.10	33.96	13570.0	208.85	851.05	0.14	4490.0	1.52	1150.0	8.13	4417.50	21.63
PASS205	161.15	40.39	11535.0	171.95	1157.50	1.76	3492.0	3.15	1373.5	13.02	4901.00	21.25
PMBL105	4.66	50.63	43050.0	20.88	836.15	2.88	23715.0	3.00	581.1	3.80	1042.00	26.45
PMBL108	3.67	55.00	38070.0	17.90	837.70	7.68	23735.0	19.83	590.8	6.33	764.15	29.48
PMBL208	0.72	82.98	46355.0	7.39	797.75	4.48	22470.0	5.70	409.4	3.60	1149.50	26.84

Table C-6. Cations in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	Al1670	Ba4554	Ca3158	Fe2382	K_7664	Li6707	Mg2852	Mn2576	Na5895	P_1774	Si2516	Sr4215
UNITS:	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g	ng/g
SAMPLE NR					***ISLE ROYALE NATIONAL PARK (ISRO, I)***							
PMBLK05	14.57	12.42	14435.0	3.90	609.65	8.51	3214.5	2.36	1585.5	8.03	1049.50	23.16
PMBLZ05	7.76	56.15	48780.0	4.11	356.75	-4.34	23320.0	4.23	503.2	7.65	1820.00	30.02
PMBLZ08	0.07	44.78	37595.0	11.37	442.35	-1.96	20545.0	6.73	626.3	6.93	1385.00	25.65
PMBS205	0.19	52.33	45870.0	2.25	307.60	1.14	23405.0	0.96	501.9	3.08	1579.00	29.71
PMHCA05	39.10	8.98	15125.0	2.62	574.90	0.71	3068.0	0.52	1823.0	3.64	52.92	24.78
PMHCA08	8.77	8.47	14900.0	6.94	1006.05	-1.83	3057.0	0.72	2893.0	6.88	864.65	24.20
PMHLK05	29.94	10.66	14075.0	3.18	589.35	-2.81	3031.5	1.25	1583.5	2.95	1042.50	23.36
PMHLK08	3.09	12.34	14410.0	3.17	560.45	1.06	3255.0	1.60	1541.5	3.69	924.05	23.05
PMHMZ05	54.85	13.38	16775.0	20.56	437.10	-3.68	6827.0	2.53	510.4	3.97	2459.00	13.88
PMHMZ08	35.57	18.65	20610.0	7.99	552.95	7.31	8137.0	2.89	508.4	3.24	1102.00	17.80
PMHS105	58.61	31.11	24265.0	32.04	708.90	-0.23	9091.0	5.48	1169.0	1.99	2682.50	25.33
PMHS108	125.35	25.76	19550.0	63.60	508.35	5.06	7195.5	5.18	1003.6	2.81	2163.00	22.68

Table C-7a. Metals (molecular weights 7Li – 66Zn) in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata.

PARAMETER:	7Li	9Be	11B	27Al	31P	47Ti	51V	52Cr	55Mn	56Fe	59Co	60Ni	63Cu	66Zn
UNITS:	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**
SAMPLE NR	***APOSTLE ISLANDS NATIONAL LAKESHORE (APIS, A)***													
ABIL105	0.734	0.001	9.318	4.051	2.641	0.097	0.286	0.112	0.741	3.333	0.016	0.182	1.612	2.227
ABIL109	0.480	0.002	40.168	18.405	8.558	0.604	0.339	0.160	20.680	58.345	0.120	0.876	3.384	4.209
ABIL205	0.785	0.000	28.675	1.645	2.033	0.029	0.218	0.104	0.248	1.238	0.005	0.124	0.740	1.589
ABIL209	0.411	0.001	68.375	16.450	1.818	0.211	0.395	0.099	0.941	6.984	0.025	0.448	1.636	1.652
ABILK05	2.456	-0.001	30.495	5.829	5.310	0.045	0.222	0.144	2.384	3.017	0.012	0.388	1.347	9.537
ABILK09	0.773	0.001	84.230	5.986	2.988	0.214	0.232	0.111	1.806	10.245	0.037	2.251	3.622	8.926
ABIS105	0.686	0.003	1.072	1.245	1.402	0.050	0.208	0.111	0.206	1.134	0.005	0.179	0.874	2.925
ABIS109	0.687	-0.001	103.725	5.389	1.464	0.162	0.259	0.122	0.601	4.000	0.010	0.392	1.459	2.366
ABIS205	0.704	0.001	7.546	1.182	1.553	0.040	0.221	0.095	0.209	0.834	0.007	0.127	0.788	1.818
ABIS209	0.669	0.001	116.795	45.125	4.445	0.284	0.312	0.141	0.722	12.275	0.019	0.404	1.624	2.238
ADIL105	0.958	0.103	30.890	702.900	3.109	0.521	0.243	0.781	63.730	60.995	0.768	0.979	0.942	12.520
ADIL109	1.161	0.089	76.775	473.900	7.223	0.294	0.188	0.815	70.805	22.380	0.743	1.406	2.736	16.360
ADIL205	0.895	0.102	23.680	566.400	3.017	0.380	0.242	0.660	58.305	41.480	0.718	1.054	1.374	19.135
ADIL209	0.965	0.095	55.106	442.900	3.483	0.471	0.250	0.587	68.810	56.770	1.018	1.324	1.897	9.912
ADILK05	0.690	-0.001	15.782	1.772	1.566	0.083	0.215	0.115	0.331	1.400	0.004	0.154	0.744	2.309
ADILK09	0.722	0.001	38.615	4.475	1.082	0.070	0.215	0.101	0.322	3.808	0.007	0.332	0.831	1.450
ADIS105	0.594	0.043	47.155	187.850	7.116	0.450	0.371	0.483	1.782	18.515	0.059	0.638	2.374	2.218
ADIS109	0.482	0.006	50.235	26.690	7.758	0.810	0.235	0.138	0.976	18.040	0.036	0.459	1.420	1.659
ADIS205	0.967	0.062	42.480	303.400	1.303	0.254	0.225	0.409	29.165	19.660	0.358	0.567	1.394	4.149
ADIS209	0.861	0.029	65.980	88.175	1.170	0.229	0.240	0.220	7.762	8.577	0.097	0.389	1.397	2.614
IBLL105	0.457	0.026	200.100	505.100	8.335	7.185	0.861	0.679	1.187	112.600	0.134	1.653	26.490	4.710
IBLL107	0.617	0.039	1182.050	406.100	10.548	2.758	1.272	0.703	3.853	66.100	0.080	1.513	27.765	3.597
IBLL110	0.475	0.039	293.050	580.450	6.590	6.639	1.230	0.809	1.984	99.040	0.140	1.974	31.005	5.090
IBLL205	0.600	0.019	61.255	572.600	8.229	5.791	0.528	0.554	3.086	118.000	0.196	1.890	6.929	13.080
IBLL207	0.463	0.013	208.400	349.700	6.369	1.862	0.515	0.483	4.845	63.655	0.257	1.605	6.102	5.882
IBLL210	0.495	0.021	62.920	605.000	4.785	4.617	0.547	0.567	1.862	121.700	0.142	1.699	6.716	6.468
IBLLK05	1.581	0.001	12.745	6.081	5.505	0.252	0.221	0.144	0.377	2.524	0.010	0.296	1.256	6.152
IBLLK07	0.727	-0.001	78.460	1.478	2.027	0.024	0.229	0.127	0.212	0.914	0.004	0.174	0.933	4.068
IBLLK10	0.700	0.001	7.703	1.884	1.361	0.101	0.227	0.109	0.086	0.827	0.002	0.142	1.135	2.502
IBLS105	0.711	0.009	61.790	223.100	2.998	0.746	0.455	0.206	1.450	44.960	0.096	0.911	5.125	7.037

Table C-7a. Metals (molecular weights 7Li – 66Zn) in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	**7Li**	**9Be**	**11B**	**27Al**	**31P**	**47Ti**	**51V**	**52Cr**	**55Mn**	**56Fe**	**59Co**	**60Ni**	**63Cu**	**66Zn**
UNITS:	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**
SAMPLE NR						***ISLE ROYALE NATIONAL PARK (ISRO, I)***								
IBLS107	0.870	0.002	143.875	27.450	1.560	0.079	0.370	0.115	0.176	1.178	0.018	0.657	1.884	3.069
IBLS110	0.683	0.003	31.650	43.140	1.369	0.070	0.251	0.136	0.242	4.279	0.013	0.241	2.096	3.419
IBLS205	0.704	0.001	26.890	4.939	1.176	0.087	0.252	0.112	0.194	1.237	0.006	0.169	1.175	2.603
IBLS207	0.824	0.000	77.635	4.331	3.561	0.074	0.383	0.118	0.126	0.907	0.008	0.257	1.824	1.778
IBLS210	0.623	0.001	8.780	5.897	2.062	0.027	0.263	0.097	0.173	1.458	0.006	0.229	1.441	3.634
IDML105	0.229	0.003	123.065	77.235	5.809	0.675	0.320	0.162	0.979	19.250	0.027	0.309	2.039	4.722
IDML107	0.218	0.003	100.630	119.650	7.756	0.913	0.610	0.207	2.029	36.000	0.049	0.388	2.612	5.090
IDML110	0.162	0.021	3.130	79.960	3.554	0.582	0.318	0.128	0.634	18.930	0.027	0.272	2.157	4.931
IDML205	0.092	0.003	215.370	80.985	5.596	1.224	0.346	0.200	0.724	25.170	0.039	0.461	9.596	4.790
IDML207	0.409	0.001	1762.000	117.000	10.560	0.412	0.440	0.214	1.347	28.215	0.034	0.349	17.850	8.135
IDML210	0.145	0.004	60.145	56.975	2.930	0.622	0.415	0.199	0.505	19.080	0.035	0.358	14.130	5.613
IDMLK05	0.846	0.001	37.995	4.414	1.775	0.061	0.230	0.131	0.221	2.253	0.011	0.279	1.058	4.767
IDMLK07	0.494	0.003	-58.610	2.389	1.843	0.010	0.228	0.121	0.172	0.658	0.004	0.153	0.922	2.992
IDMLK10	0.743	0.001	7.588	2.888	1.995	-0.018	0.233	0.142	0.186	0.995	0.004	0.211	1.217	3.087
IDMS105	0.782	0.001	82.430	3.999	2.059	0.055	0.256	0.136	0.222	1.671	0.006	0.247	1.274	4.010
IDMS107	0.624	-0.001	9.050	6.709	3.646	0.033	0.393	0.129	0.176	4.928	0.007	0.240	2.271	3.730
IDMS110	0.708	0.001	8.998	1.887	2.146	0.013	0.227	0.102	0.129	0.769	0.003	0.147	0.984	1.990
IDMS205	0.764	0.001	69.875	6.106	1.746	0.123	0.292	0.130	0.199	1.928	0.008	0.217	1.223	3.731
IDMS207	0.524	-0.001	-30.570	43.755	9.096	0.169	0.767	0.162	0.259	3.220	0.024	0.585	5.370	7.237
IDMS210	0.238	0.003	9.000	25.780	2.262	0.201	0.330	0.097	0.193	6.644	0.011	0.193	1.841	3.009
IPAL105	0.079	0.009	69.185	604.850	2.672	1.108	0.394	0.341	2.625	110.100	0.220	2.970	7.632	5.736
IPAL107	-0.127	0.008	-39.470	625.350	3.860	0.976	0.397	0.352	3.623	81.020	0.162	3.381	8.159	6.810
IPAL110	-0.109	0.010	-65.355	900.250	3.187	1.494	0.457	0.420	4.483	139.350	0.339	3.627	8.285	7.868
IPAL205	0.088	0.010	69.325	584.550	2.438	0.986	0.346	0.291	1.597	103.700	0.171	2.756	9.010	4.230
IPAL207	-0.101	0.007	2.750	533.300	6.988	0.856	0.503	0.328	9.195	226.750	0.246	3.233	8.377	7.062
IPAL210	0.293	0.153	8.962	712.100	2.877	1.121	0.327	0.304	3.005	120.300	0.236	2.890	8.385	6.112
IPALK05	0.679	0.001	4.735	1.049	1.452	0.024	0.237	0.139	0.162	1.244	0.012	0.292	1.939	2.977
IPALK07	0.483	-0.001	-14.800	4.726	1.701	0.059	0.236	0.109	0.201	1.139	0.004	0.158	0.994	2.315
IPALK10	0.658	0.001	1.400	1.563	1.540	0.059	0.231	0.111	0.142	0.884	0.004	0.155	1.010	2.914
IPAS105	0.762	0.001	35.675	7.731	1.339	0.048	0.310	0.115	0.171	1.402	0.006	0.210	1.623	1.726
IPAS107	0.482	0.019	3.550	9.834	5.220	0.074	0.473	0.118	0.211	2.221	0.018	0.417	3.749	2.544
IPAS110	0.604	0.003	3.592	4.822	1.390	0.150	0.267	0.090	0.201	1.321	0.008	0.147	1.259	3.321

Table C-7a. Metals (molecular weights 7Li – 66Zn) in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	7Li	9Be	11B	27Al	31P	47Ti	51V	52Cr	55Mn	56Fe	59Co	60Ni	63Cu	66Zn
UNITS:	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb
SAMPLE NR	***ISLE ROYALE NATIONAL PARK (ISRO, I)***													
IPAS205	0.758	0.001	20.485	9.325	1.646	0.176	0.308	0.119	0.206	1.646	0.006	0.198	1.670	2.113
IPAS207	0.584	0.000	-4.500	9.048	2.474	0.248	0.476	0.102	0.217	1.719	0.011	0.276	3.489	2.507
IPAS210	0.666	0.002	2.480	3.083	1.561	0.054	0.231	0.100	0.151	1.471	0.003	0.118	1.226	2.644
IRSL105	0.119	0.009	210.400	50.010	4.746	0.114	0.159	0.080	0.660	11.265	0.034	0.300	8.594	8.636
IRSL107	0.127	0.003	143.800	42.310	16.600	0.233	0.237	0.136	2.531	33.265	0.401	0.924	15.415	10.910
IRSL110	0.014	0.016	137.100	64.785	3.147	0.225	0.156	0.086	0.816	18.220	0.056	0.299	12.045	10.820
IRSL205	0.145	0.005	246.165	138.800	7.749	0.218	0.195	0.109	1.008	16.820	0.032	0.353	6.735	6.713
IRSL207	0.143	0.004	478.050	150.900	15.275	0.285	0.275	0.215	2.950	52.925	0.061	0.613	8.194	11.095
IRSL210	0.017	0.003	50.440	115.650	5.268	0.285	0.161	0.071	1.159	25.340	0.037	0.339	6.017	6.192
IRSLK05	0.684	0.001	67.635	1.916	1.990	0.050	0.219	0.110	0.146	1.091	0.005	0.200	0.950	3.762
IRSLK07	0.695	0.001	169.850	1.823	2.224	0.070	0.231	0.219	0.226	0.877	0.004	0.158	0.947	4.285
IRSLK10	0.830	0.008	6.136	7.704	2.869	0.071	0.236	0.126	0.329	3.038	0.013	0.366	1.736	9.207
IRSS105	0.798	0.001	223.215	14.955	2.075	0.228	0.342	0.132	0.282	2.754	0.013	0.230	3.028	2.946
IRSS107	0.672	0.001	126.005	12.335	3.272	0.442	0.256	0.209	0.507	6.524	0.043	0.530	1.970	10.695
IRSS110	0.663	0.000	5.087	3.233	1.750	0.159	0.239	0.126	0.156	1.409	0.005	0.233	1.245	5.011
IRSS205	0.777	0.001	117.045	5.893	2.108	0.082	0.416	0.177	0.365	2.243	0.054	0.342	2.674	2.908
IRSS207	0.690	0.002	137.400	3.252	1.909	0.057	0.233	0.183	0.168	1.612	0.004	0.164	1.131	5.094
IRSS210	0.744	-0.001	4.081	3.892	2.263	0.076	0.240	0.120	0.181	1.512	0.005	0.223	1.439	4.396
	PICTURED ROCKS NATIONAL LAKESHORE (PIRO, P)													
PASL105	0.556	0.044	7.092	471.800	16.145	4.653	1.951	0.965	3.886	220.000	0.272	0.477	1.271	60.025
PASL108	0.521	0.059	9.606	578.000	12.660	6.355	1.514	0.944	28.095	692.250	0.414	0.873	1.939	17.440
PASL205	0.521	0.072	7.842	561.200	6.208	5.495	1.771	0.879	2.666	199.900	0.214	0.567	2.297	20.310
PASL208	0.442	0.144	7.285	470.550	15.320	6.912	3.287	0.990	87.095	1328.500	0.447	0.457	0.693	5.182
PASLK05	0.717	0.006	9.238	7.176	3.364	0.122	0.260	0.136	0.607	4.064	0.015	0.173	1.616	3.957
PASLK08	0.666	0.002	6.625	4.624	0.830	0.152	0.268	0.136	0.690	7.494	0.006	0.155	3.304	1.162
PASS105	0.303	0.355	5.368	35.335	3.803	6.588	3.090	0.902	1.443	176.500	0.095	0.833	3.834	4.233
PASS205	0.367	0.233	6.348	59.720	10.474	8.613	2.950	0.966	2.915	163.650	0.225	1.507	7.005	78.255
PMBL105	0.667	0.008	5.232	6.214	3.584	0.152	0.105	0.109	3.175	21.000	0.330	2.267	7.202	1.771
PMBL108	0.874	0.002	7.064	5.343	6.415	0.544	0.079	0.077	20.175	18.380	0.478	1.812	5.617	2.553
PMBL208	0.565	0.004	4.970	2.377	2.160	0.099	0.036	0.060	5.795	6.392	0.318	1.378	9.543	2.422
PMBLK05	0.733	0.005	7.853	8.995	5.766	0.080	0.242	0.132	2.276	4.121	0.027	0.484	1.487	3.905

Table C-7a. Metals (molecular weights 7Li – 66Zn) in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	**7Li**	**9Be**	**11B**	**27Al**	**31P**	**47Ti**	**51V**	**52Cr**	**55Mn**	**56Fe**	**59Co**	**60Ni**	**63Cu**	**66Zn**
UNITS:	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**
SAMPLE NR					***PICTURED ROCKS NATIONAL LAKESHORE (PIRO, P)***									
PMBLZ05	0.542	0.007	5.991	9.823	5.739	0.031	0.173	0.082	4.312	4.758	0.191	1.038	3.668	2.720
PMBLZ08	0.544	0.001	5.202	2.082	4.924	0.105	0.353	0.073	7.207	9.937	0.286	1.001	4.232	4.993
PMBS205	0.509	0.011	6.168	1.864	3.111	0.033	0.145	0.063	0.983	2.089	0.110	0.717	3.165	1.391
PMHCA05	0.815	0.001	3.214	41.790	4.172	0.117	0.374	0.150	0.352	2.161	0.078	0.343	7.002	13.115
PMHCA08	0.902	0.005	6.634	10.500	7.462	0.711	0.322	0.141	0.568	7.058	0.463	1.120	10.925	2.408
PMHLK05	0.756	0.003	7.440	33.160	6.637	0.050	0.254	0.115	1.146	3.361	0.021	0.169	1.048	6.628
PMHLK08	0.801	0.001	7.214	4.368	2.973	0.032	0.235	0.246	1.663	3.622	0.016	0.295	3.329	4.185
PMHMZ05	0.604	0.012	6.042	56.700	4.214	0.418	0.107	0.138	2.569	20.335	0.099	0.809	5.448	3.843
PMHMZ08	0.730	0.009	6.262	37.915	1.423	0.215	0.086	0.096	2.701	7.576	0.139	0.748	6.283	2.752
PMHS105	0.930	0.031	6.698	59.310	3.513	0.580	0.330	0.268	5.218	31.250	0.177	1.706	29.070	5.388
PMHS108	0.927	0.039	5.963	125.650	2.151	0.899	0.325	0.272	5.008	61.270	0.173	1.537	27.080	2.107

Table C-7b. Metals (molecular weights 75As – 235U) in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata.

PARAMETER: **UNITS:**	**75As** **ppb**	**78Se** **ppb**	**85Rb** **ppb**	**88Sr** **ppb**	**90Zr** **ppb**	**93Nb** **ppb**	**95Mo** **ppb**	**111Cd** **ppb**	**138Ba** **ppb**	**181Ta** **ppb**	**182W** **ppb**	**205Tl** **ppb**	**208Pb** **ppb**	**238U** **ppb**
SAMPLE NR					***APOSTLE ISLANDS NATIONAL LAKESHORE (APIS, A)***									
ABIL105	0.446	0.140	0.666	23.650	0.025	-0.022	0.197	0.024	10.310	-0.011	-0.130	0.001	0.057	0.047
ABIL109	0.659	0.188	0.695	11.275	0.233	0.045	0.083	0.047	6.263	0.023	-0.083	0.014	0.081	0.026
ABIL205	0.355	0.057	0.675	22.220	0.010	-0.027	0.134	0.015	9.755	-0.011	-0.138	0.001	0.018	0.046
ABIL209	0.265	0.176	0.568	12.270	0.109	0.023	0.095	0.020	4.659	0.004	-0.104	0.003	0.041	0.021
ABILK05	0.411	0.107	0.918	22.865	0.036	-0.027	0.146	0.045	10.134	-0.010	-0.138	0.001	0.036	0.055
ABILK09	0.389	0.121	0.732	22.320	0.052	0.000	0.151	0.016	11.200	-0.003	-0.111	0.004	0.060	0.051
ABIS105	0.384	0.061	0.712	21.775	0.023	0.014	0.165	0.013	9.233	-0.003	-0.099	0.003	0.016	0.045
ABIS109	0.366	0.148	0.652	22.225	0.039	0.009	0.193	0.015	9.391	-0.001	-0.106	0.002	0.033	0.057
ABIS205	0.381	0.066	0.696	21.925	0.013	-0.012	0.156	0.012	9.197	-0.008	-0.126	0.002	0.020	0.045
ABIS209	0.342	0.146	1.152	17.330	0.091	0.007	0.134	0.038	7.322	-0.002	-0.114	0.004	0.052	0.036
ADIL105	0.160	0.126	1.094	8.423	0.461	-0.032	0.007	0.081	23.030	-0.014	-0.149	0.009	0.062	0.020
ADIL109	0.217	0.173	0.914	7.918	0.511	0.004	0.022	0.085	20.170	-0.001	-0.118	0.011	0.071	0.022
ADIL205	0.125	0.148	1.062	9.744	0.440	-0.035	0.011	0.107	23.920	-0.014	-0.151	0.010	1.089	0.021
ADIL209	0.144	0.183	0.893	8.827	0.416	0.005	0.022	0.064	22.295	-0.003	-0.123	0.010	0.072	0.025
ADILK05	0.393	0.091	0.695	21.935	0.012	-0.039	0.129	0.016	10.206	-0.016	-0.149	0.000	0.028	0.045
ADILK09	0.366	0.095	0.724	22.215	0.011	-0.010	0.141	0.008	9.671	-0.009	-0.122	0.001	0.031	0.051
ADIS105	0.387	0.159	0.751	13.490	0.307	-0.026	0.257	0.027	11.895	-0.011	-0.144	0.005	0.090	0.043
ADIS109	0.329	0.135	0.578	12.485	0.121	-0.002	0.069	0.015	7.103	-0.005	-0.117	0.005	0.051	0.018
ADIS205	0.261	0.154	0.896	15.985	0.232	-0.031	0.076	0.037	18.690	-0.013	-0.146	0.007	0.031	0.046
ADIS209	0.315	0.133	0.775	19.485	0.124	-0.009	0.117	0.021	12.900	-0.008	-0.122	0.005	0.109	0.049
						ISLE ROYALE NATIONAL PARK (ISRO, I)								
IBLL105	0.548	0.362	0.100	11.250	0.736	0.079	0.065	0.038	5.485	0.024	-0.119	0.003	0.207	0.013
IBLL107	0.722	0.435	0.039	16.575	0.767	0.080	0.051	0.006	1.513	0.022	-0.119	0.003	0.096	0.015
IBLL110	0.553	0.417	0.055	13.185	0.841	0.081	0.074	0.013	5.513	0.009	-0.141	0.003	0.192	0.013
IBLL205	0.635	0.266	0.210	7.154	0.584	0.079	0.080	0.041	2.526	0.016	-0.115	0.005	0.378	0.011
IBLL207	0.809	0.357	0.187	7.390	0.477	0.151	0.120	0.114	1.784	0.083	-0.028	0.095	0.273	0.107
IBLL210	0.601	0.209	0.135	8.329	0.541	0.074	0.029	0.023	2.190	0.006	-0.141	0.003	0.250	0.008
IBLLK05	0.409	0.126	0.746	22.040	0.021	0.045	0.071	0.022	9.535	0.000	-0.124	0.002	0.088	0.042
IBLLK07	0.429	0.167	0.746	22.530	0.016	0.044	0.141	0.013	10.630	0.002	-0.128	0.002	0.064	0.046
IBLLK10	0.428	0.083	0.735	22.160	0.011	0.052	0.139	0.014	9.212	-0.004	-0.140	0.003	0.083	0.043
IBLS105	0.659	0.150	0.626	18.360	0.120	0.052	0.115	0.031	7.688	0.006	-0.120	0.003	0.194	0.031

Table C-7b. Metals (molecular weights 75As – 235U) in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	**75As**	**78Se**	**85Rb**	**88Sr**	**90Zr**	**93Nb**	**95Mo**	**111Cd**	**138Ba**	**181Ta**	**182W**	**205Tl**	**208Pb**	**238U**
UNITS:	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**
SAMPLE NR						***ISLE ROYALE NATIONAL PARK (ISRO, I)***								
IBLS107	0.525	0.150	0.928	24.970	0.025	0.049	0.147	0.017	10.460	0.005	-0.123	0.004	0.046	0.051
IBLS110	0.411	0.091	0.728	22.910	0.033	0.057	0.141	0.025	8.960	-0.003	-0.140	0.003	0.068	0.043
IBLS205	0.520	0.134	0.771	22.480	0.021	0.045	0.139	0.016	9.334	0.000	-0.123	0.002	0.059	0.045
IBLS207	0.744	0.139	0.914	21.010	0.018	0.045	0.131	0.020	10.340	0.002	-0.125	0.003	0.052	0.037
IBLS210	0.423	0.087	0.671	19.545	0.013	0.053	0.129	0.018	7.480	-0.003	-0.140	0.002	0.075	0.041
IDML105	0.241	0.217	0.201	2.369	0.072	0.008	0.049	0.036	5.945	0.005	-0.123	0.002	0.141	0.008
IDML107	0.474	0.446	0.218	3.373	0.062	0.084	0.024	0.029	4.806	0.066	-0.109	0.005	0.260	0.014
IDML110	0.195	0.118	0.095	2.253	0.079	0.082	0.028	0.016	1.015	0.009	-0.125	0.005	0.167	0.006
IDML205	0.293	0.255	0.125	6.788	0.158	0.006	0.268	0.035	26.825	-0.001	0.108	0.003	0.164	0.007
IDML207	0.347	0.319	0.132	15.780	0.169	0.116	0.166	0.043	55.595	0.028	-0.082	0.004	0.139	0.011
IDML210	0.253	0.251	0.112	12.140	0.155	0.075	0.109	0.019	50.895	0.007	-0.132	0.004	0.167	0.007
IDMLK05	0.402	0.128	0.738	22.195	0.025	0.059	0.235	0.026	9.444	0.010	-0.115	0.008	0.089	0.050
IDMLK07	0.422	0.151	0.728	22.525	0.018	0.059	0.143	0.010	9.628	0.003	-0.116	0.002	0.031	0.045
IDMLK10	0.415	0.167	0.718	22.015	0.020	0.065	0.147	0.013	9.381	0.001	-0.135	0.003	0.095	0.044
IDMS105	0.501	0.128	0.771	23.195	0.070	0.137	0.192	0.024	10.685	0.035	-0.030	0.005	0.047	0.052
IDMS107	0.520	0.178	0.944	25.640	0.030	0.087	0.170	0.044	11.350	0.022	-0.104	0.011	0.087	0.051
IDMS110	0.412	0.092	0.742	22.715	0.021	0.077	0.160	0.013	9.956	0.004	-0.132	0.003	0.065	0.046
IDMS205	0.509	0.162	0.761	23.005	0.039	0.107	0.158	0.021	10.365	0.026	-0.085	0.003	0.055	0.054
IDMS207	1.035	0.251	0.802	19.760	0.061	0.070	0.086	0.042	11.235	0.012	-0.110	0.006	0.111	0.026
IDMS210	0.250	0.119	0.217	6.675	0.032	0.061	0.049	0.016	3.831	0.001	-0.136	0.002	0.145	0.009
IPAL105	0.456	0.136	0.117	17.370	0.667	0.022	0.029	0.036	3.729	0.005	-0.117	0.010	0.130	0.013
IPAL107	0.541	0.279	0.193	17.895	0.799	0.068	0.032	0.019	2.403	0.013	-0.122	0.004	0.142	0.008
IPAL110	0.566	0.215	0.130	16.800	0.770	0.034	0.016	0.023	2.418	-0.005	-0.138	0.003	0.164	0.006
IPAL205	0.522	0.179	0.127	16.100	0.702	0.015	0.035	0.026	2.187	0.001	-0.122	0.006	0.172	0.009
IPAL207	0.652	0.321	0.250	18.700	0.806	0.062	0.026	0.028	3.163	0.011	-0.124	0.004	0.250	0.008
IPAL210	0.537	0.168	0.175	13.895	0.635	0.100	0.115	0.050	1.722	0.023	-0.095	0.032	0.213	0.033
IPALK05	0.412	0.101	0.738	22.025	0.019	0.008	0.147	0.017	9.177	-0.002	-0.118	0.009	0.035	0.051
IPALK07	0.426	0.153	0.740	22.500	0.014	0.038	0.143	0.010	9.449	-0.005	-0.130	0.002	0.059	0.043
IPALK10	0.414	0.071	0.725	21.960	0.008	0.044	0.132	0.013	9.072	-0.006	-0.146	0.002	0.053	0.043
IPAS105	0.578	0.064	0.945	24.650	0.020	-0.002	0.152	0.014	9.671	-0.010	-0.124	0.003	0.038	0.051
IPAS107	0.759	0.287	1.169	19.085	0.041	0.047	0.122	0.037	8.035	0.004	-0.125	0.004	0.068	0.015

Table C-7b. Metals (molecular weights 75As – 235U) in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	75As	78Se	85Rb	88Sr	90Zr	93Nb	95Mo	111Cd	138Ba	181Ta	182W	205Tl	208Pb	238U
UNITS:	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb	ppb
SAMPLE NR						***ISLE ROYALE NATIONAL PARK (ISRO, I)***								
IPAS110	0.391	0.106	0.686	20.190	0.019	0.057	0.114	0.013	7.532	-0.003	-0.137	0.002	0.079	0.034
IPAS205	0.604	0.042	0.979	24.310	0.014	-0.002	0.141	0.013	9.818	-0.011	-0.126	0.002	0.044	0.047
IPAS207	0.701	0.182	1.268	24.030	0.032	0.043	0.150	0.017	9.876	-0.002	-0.127	0.003	0.047	0.034
IPAS210	0.413	0.085	0.709	21.295	0.008	0.048	0.123	0.008	8.361	-0.006	-0.146	0.001	0.055	0.038
IRSL105	0.204	0.128	0.155	15.840	0.149	0.110	0.074	0.017	2.598	0.025	-0.049	0.005	0.111	0.012
IRSL107	0.780	0.538	0.431	22.470	0.528	0.531	0.593	0.354	3.083	0.286	0.438	0.120	0.513	0.414
IRSL110	0.223	0.089	0.138	19.790	0.138	0.040	0.025	0.032	2.261	-0.006	-0.144	0.009	1.215	0.014
IRSL205	0.291	0.119	0.230	17.190	0.115	0.057	0.082	0.017	2.272	0.019	-0.091	0.003	0.130	0.014
IRSL207	0.526	0.238	0.259	15.120	0.123	0.125	0.066	0.029	2.527	0.034	-0.062	0.006	0.293	0.015
IRSL210	0.264	0.079	0.172	10.895	0.090	0.041	0.031	0.017	1.352	-0.005	-0.152	0.002	0.284	0.007
IRSLK05	0.411	0.073	0.723	21.835	0.011	0.011	0.156	0.014	9.536	-0.005	-0.118	0.002	0.035	0.043
IRSLK07	0.418	0.252	0.737	22.250	0.023	0.087	0.143	0.017	9.704	0.048	-0.103	0.007	0.058	0.049
IRSLK10	0.417	0.100	0.749	22.000	0.071	0.171	0.189	0.037	9.091	0.024	-0.068	0.009	0.148	0.047
IRSS105	0.617	0.128	0.851	23.435	0.035	0.031	0.149	0.020	9.118	0.006	-0.105	0.006	0.058	0.049
IRSS107	0.416	0.171	0.709	20.800	0.072	0.130	0.188	0.058	9.102	0.046	-0.059	0.033	0.389	0.075
IRSS110	0.427	0.107	0.736	22.145	0.009	0.045	0.140	0.014	9.647	-0.008	-0.150	0.001	0.068	0.039
IRSS205	0.778	0.221	1.051	25.390	0.067	0.057	0.205	0.059	10.068	0.032	-0.072	0.044	0.249	0.091
IRSS207	0.411	0.119	0.722	22.270	0.036	0.099	0.144	0.014	10.095	0.024	-0.095	0.003	0.123	0.051
IRSS210	0.416	0.125	0.749	22.425	0.007	0.043	0.138	0.019	9.098	-0.008	-0.151	0.001	0.091	0.042
						PICTURED ROCKS NATIONAL LAKESHORE (PIRO, P)								
PASL105	0.503	0.120	0.928	13.095	0.390	0.096	0.036	0.138	16.885	0.019	-0.132	0.012	46.510	0.027
PASL108	0.558	0.165	0.871	12.560	0.599	0.093	0.044	0.041	16.710	0.015	-0.138	0.009	0.833	0.035
PASL205	0.416	0.105	1.091	13.375	0.522	0.094	0.056	0.055	16.430	0.011	-0.136	0.011	8.249	0.038
PASL208	0.543	0.246	0.796	10.810	0.913	0.085	0.026	0.007	18.260	0.012	-0.143	0.002	0.249	0.034
PASLK05	0.430	0.144	0.810	22.985	0.093	0.221	0.193	0.023	10.540	0.045	-0.019	0.013	1.062	0.054
PASLK08	0.391	0.117	0.756	23.150	0.014	0.055	0.136	0.009	9.846	-0.007	-0.144	0.001	0.036	0.047
PASS105	0.495	0.234	1.001	17.790	1.613	0.079	0.021	0.026	27.025	0.001	-0.143	0.004	0.148	0.085
PASS205	0.612	0.243	1.321	18.455	1.941	0.090	0.035	0.108	34.045	0.002	-0.139	0.009	0.236	0.088
PMBL105	0.356	0.059	1.044	26.175	0.108	0.090	0.847	0.021	47.495	0.012	-0.124	0.016	0.089	0.366
PMBL108	0.731	0.070	0.989	29.780	0.105	0.068	0.575	0.021	52.810	0.002	-0.136	0.010	0.086	0.180
PMBL208	0.231	0.019	0.939	26.535	0.037	0.060	0.334	0.011	77.060	-0.001	-0.142	0.006	0.054	0.104

Table C-7b. Metals (molecular weights 75As – 235U) in water samples collected from rock pools and Lake Superior at Isle Royale National Park, Apostle Islands National Lakeshore, and Pictured Rocks National Lakeshore, 2010. See Table C-1 for metadata (continued).

PARAMETER:	**75As**	**78Se**	**85Rb**	**88Sr**	**90Zr**	**93Nb**	**95Mo**	**111Cd**	**138Ba**	**181Ta**	**182W**	**205Tl**	**208Pb**	**238U**
UNITS:	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**	**ppb**
SAMPLE NR					***PICTURED ROCKS NATIONAL LAKESHORE (PIRO, P)***									
PMBLK05	0.420	0.067	0.789	23.105	0.042	0.102	0.300	0.042	11.980	0.016	-0.112	0.013	0.093	0.062
PMBLZ05	0.144	0.085	0.398	30.090	0.094	0.118	0.565	0.019	53.665	0.015	-0.109	0.009	0.078	0.153
PMBLZ08	0.196	0.075	0.502	26.155	0.060	0.059	0.482	0.014	42.175	-0.006	-0.143	0.004	0.058	0.152
PMBS205	0.154	0.074	0.356	29.515	0.151	0.208	0.654	0.013	49.990	0.043	-0.040	0.015	0.058	0.160
PMHCA05	0.695	0.132	0.739	24.860	0.033	0.081	0.178	0.012	8.561	0.003	-0.125	0.004	0.228	0.107
PMHCA08	0.649	0.081	1.387	24.225	0.088	0.084	0.173	0.028	8.244	0.005	-0.119	0.008	0.090	0.093
PMHLK05	0.408	0.148	0.857	23.660	0.020	0.074	0.176	0.021	10.390	0.000	-0.132	0.005	0.141	0.052
PMHLK08	0.406	0.055	0.759	23.385	0.020	0.068	0.153	0.011	11.990	-0.002	-0.132	0.003	0.052	0.053
PMHMZ05	0.172	0.112	0.638	13.875	0.159	0.122	0.240	0.018	12.800	0.022	-0.103	0.011	0.142	0.069
PMHMZ08	0.260	0.076	0.833	18.005	0.220	0.214	0.301	0.017	17.965	0.049	-0.018	0.026	0.071	0.160
PMHS105	0.529	0.203	1.201	24.785	0.524	0.103	0.548	0.019	29.415	0.020	-0.119	0.011	2.177	0.462
PMHS108	0.579	0.129	0.851	22.440	0.654	0.077	0.391	0.016	24.720	0.006	-0.131	0.009	0.177	0.409

Appendix D: Comprehensive Taxa List

Zooplankton

	Parks		
CRUSTACEA (20 taxa)	***APIS***	***ISRO***	***PIRO***
Acanthocyclops capillatus	X		
Cyclops sp.	X	X	
Diacyclops albus		X	
Diacyclops langoidus			X
Diacyclops nanus		X	X
Diacyclops thomasi	X	X	
Diacyclops sp.	X	X	X
Eucyclops elegans	X		
Microcyclops rubellus	X	X	X
Microcyclops vericans	X		
Paracyclops (chiltoni)			X
cyclopoid adult, unidentified		X	
Harpacticoid	X	X	X
Epischura lacustris		X	X
Leptodiaptomus sicilis	X	X	X
Leptodiaptomus sp.	X	X	
Limnocalanus macrurus			X
Senecella calanoides	X		
Skistodiaptomus oregonensis	X		
Skistodiaptomus reighardi		X	
CLADOCERA (28 taxa)			
Acroperus harpae		X	
Alona sp.	X	X	
Alona bicolor		X	
Alona circumfimbriata		X	X
Alona costata	X	X	
Alona gutatta	X	X	X
Alona quadrangula		X	
Alona rectangula	X	X	
Alonella nana	X	X	
Biapertura (Alona) affinis	X	X	
Chydorus sp.	X	X	X
Chydorus faviformis		X	
Chydorus sphaericus	X	X	
Eurycercus longirostris	X		
Kurzia (latissima)	X		
(Paralona pigra)		X	

	Parks		
	APIS	***ISRO***	***PIRO***
Ceriodaphnia sp.		X	X
Ceriodaphnia lacustris		X	
Ceriodaphnia quadrangula		X	
Diaphanosoma sp.		X	
Simocephalus sp.		X	
Scapholeberis mucronata		X	
Bosmina longirostrus	X		
Bosmina spp.	X	X	
Daphnia ambigua		X	
Daphnia pulex catawba		X	
Daphnia mendotae dentifera		X	
Holopedium gibberum	X	X	
ROTIFERA (96 taxa)			
Bdelloid rotifer	X	X	X
Adineta sp.	X	X	X
Anuraeopsis fissa	X		
Ascomorpha sp.	X	X	X
Asplanchna herricki	X		
Asplanchna priodonta	X	X	X
Asplanchna sp.	X	X	X
Cephalodella sp.	X		X
Collotheca mutabilis	X		
Collotheca pelagica		X	X
Colurella sp.	X	X	X
Conochilus sp.	X	X	X
Conochilus hippocrepis	X		
Conochilus unicornis	X	X	
Conochiloides dossarius	X	X	X
Dicranophorus sp.	X	X	X
Dissotrocha sp.	X	X	
Encentrum sp.	X	X	
Euchlanis calpidia		X	
Euchlanis dilatata		X	
Euchlanis triquetra		X	
Euchlanis spp.		X	X
Gastropus stylifer	X	X	X
Habrotrocha sp.			X

Zooplankton

ROTIFERA (continued)	Parks		
	APIS	***ISRO***	***PIRO***
Harringia sp.	X		
Hexarthra mira		X	X
Kellicottia bostoniensis	X	X	X
Kellicottia longispina	X	X	X
Keratella cochlearis cochlearis	X	X	X
Keratella cochlearis robusta		X	
Keratella cochlearis tecta	X	X	X
Keratella crassa		X	
Keratella earlinae	X	X	X
Keratella hiemalis	X		X
Keratella quadrata	X		
Lecane candida		X	
Lecane crepida	X		
Lecane flexilis	X	X	
Lecane inermis	X	X	X
Lecane luna		X	X
Lecane mira	X	X	X
Lecane mucronata	X		
Lecane ovalis		X	
Lecane stenroosi	X		
Lecane (tenuiseta)	X	X	
Lecane tudicola			X
Lepadella patella	X	X	X
Lepadella ovalis	X	X	X
Lepadella triptera	X		
Lophocharis sp.		X	
Monostyla sp.	X		
Monostyla bulla	X	X	X
Monostyla closterocerca	X	X	
Monostyla copeis	X	X	
Monostyla cornuta	X		
Monostyla crenata	X		
Monostyla lunaris	X	X	X
Monostyla obtusa	X	X	
Monostyla quadridentata		X	
Mytilina ventralis		X	
Notholca acuminata	X		X
Notholca caudata	X		

	Parks		
	APIS	***ISRO***	***PIRO***
Notholca labis	X		
Notholca laurentiae	X		
Notholca squamula	X		X
Notholca sp.	X		
Notomata sp.	X		
Philodina sp.	X	X	X
Ploesoma sp.	X		X
Ploesoma hudsoni	X		
Ploesoma truncata	X		
Polyarthra dolichoptera	X	X	
Polyarthra major		X	
Polyarthra remata	X	X	X
Polyarthra vulgaris	X	X	X
Polyarthra spp.	X	X	X
Pompholyx sulcata	X	X	
Proales sp.	X	X	X
Rotaria sp.	X		
Schwabia sp.	X		
Synchaeta sp.	X	X	X
Synchaeta grandis			X
Synchaeta kitina			X
Synchaeta tremula			X
Squatinella sp.		X	
Testudinella sp.	X		
Trichocerca caputina			X
Trichocerca cylindrica	X	X	X
Trichocerca elongata		X	
Trichocerca iernis	X		
Trichocerca porcellus			X
Trichocerca pusilla	X	X	
Trichocerca rousseleti	X		
Trichotria tetractis	X	X	
Wierzyskiella velox			X
unidentified rotifer	X	X	X
TESTATE PROTISTA (23 taxa)			
Arcella gibbosa	X		X
Arcella sp.	X		
Centropyxis constricta aerophila	X	X	X

Zooplankton

	Parks		
TESTATE PROTISTA (continued)	***APIS***	***ISRO***	***PIRO***
Centropyxis constricta spinosa	**X**	**X**	**X**
Codonella sp.	**X**	**X**	**X**
Cucurbitella (tricuspis)			**X**
Cyclopyxis spp.	**X**	**X**	**X**
Difflugia bacillaliarum			**X**
Difflugia (lucida)		**X**	
Difflugia (oblonga)	**X**		
Difflugia urceolata	**X**		
Difflugia sp.	**X**	**X**	
Euglypha sp.	**X**	**X**	**X**
Geopyxella sp.	**X**	**X**	
Hyalosphenia papilio	**X**		
Lesquereusia spiralis		**X**	**X**
Nadinella sp.			**X**
Nebellidae	**X**		**X**
Phryganella sp.	**X**		
Trinema sp.	**X**	**X**	
(Wailesella eboracensis)	**X**		**X**
unidentified testate			**X**
unidentified protist	**X**	**X**	**X**
OSTRACODA (7 taxa)			
Candoninae		**X**	
Cypridopsinae	**X**		
Cypridopsis sp.			**X**
Potamocypris unicaudata	**X**		
Potamocypris sp.	**X**	**X**	
Scottia pseudobrowniana (?)	**X**	**X**	
unidentified ostracod		**X**	
juvenile ostracod		**X**	**X**
OTHER			
Hydrachnidiae	**X**	**X**	**X**
Tardigrada			**X**
Collembola		**X**	

Macroinvertebrates

	Parks		
AMPHIPODA	*APIS*	*ISRO*	*PIRO*
Hyalella azteca		X	
COLLEMBOLA			
PODURIDAE			
Podura aquatica		X	
ISOTOMIDAE			
Semicerura		X	
Genus 1		X	
SMINTHURIDAE			
Sminthurides		X	
Sminthurus		X	
Genus 1		X	
HYPOGASTRURIDAE			
Genus 1		X	
ENTOMOBRYIDAE			
Harlomillsia		X	
Tomocerus		X	
EPHEMEROPTERA			
BAETIDAE			
Baetis		X	
Camelobaetidus?		X	
Genus 1		X	
CAENIDAE			
Amercaenis or Caenis?		X	
HEPTAGENIIDAE			
Heptagenia		X	
Leucrocuta		X	
Genus 1		X	
LEPTOPHLEBIIDAE			
Leptophlebia		X	
Genus 1		X	
ODONATA			
AESHNIDAE			
Aeshna		X	
Triacanthagyna		X	
LIBELLULIDAE			
Erythrodiplax		X	
Libellula		X	
Genus 1		X	

	Parks		
PLECOPTERA	*APIS*	*ISRO*	*PIRO*
CAPNIIDAE			
Paracapnia		X	
Capnia		X	
Allocapnia		X	
PERLODIDAE			
Arcynopteryx		X	
Diura or Isoperla?		X	
Osobenus		X	
Skwala		X	
Genus 1		X	
CHLOROPERLIDAE			
Genus 1		X	
Haploperla		X	
PERLIDAE			
Genus 1		X	
HEMIPTERA			
CORIXIDAE			
Callicorixa		X	
Corisella		X	
Sigara		X	
NOTONECTIDAE			
Buenoa		X	
Notonecta		X	
GERRIDAE			
Aquarius		X	
Gerris		X	
Limnoporus		X	
SALDIDAE			
Rupisalda		X	
Salda		X	
Saldula		X	
COLEOPTERA			
GYRINIDAE			
Gyrinus		X	
SCIRTIDAE			
Ora?		X	
Prionocyphon		X	

Macroinvertebrates

	Parks		
COLEOPTERA (continued)	***APIS***	***ISRO***	***PIRO***
CARABIDAE			
Genus 1		**X**	
DYTISCIDAE			
Acilius		**X**	
Agabus		**X**	
Copelatus?		**X**	
Hydroporus		**X**	
Hydrotrupes?		**X**	
Hygrotus		**X**	
Laccophilus		**X**	
Liodessus		**X**	
Nebrioporus		**X**	
Neoporus		**X**	
Oreodytes		**X**	
Rhantus		**X**	
Stictotarsus		**X**	
Genus 1		**X**	
HYDROPHILIDAE			
Helophorus		**X**	
Helocombus		**X**	
Hydrobius?		**X**	
Hydrochus		**X**	
Paracymus?		**X**	
Genus 1		**X**	
TRICHOPTERA			
LIMNEPHILIDAE			
Frenesia		**X**	
Glyphopsyche		**X**	
Grammotaulis		**X**	
Limnephilus		**X**	
Psycoglypha		**X**	
Hesperophylax designatus		**X**	
GLOSSOSOMATIDAE			
Genus 1		**X**	
HYDROPSYCHIDAE			
Hydropsyche		**X**	
LEPTOCERIDAE			
Oecetis		**X**	
Ceraclea		**X**	

	Parks		
	APIS	***ISRO***	***PIRO***
APATANIIDAE			
Apatania zonella		**X**	
LEPIDOSTOMATIDAE			
Lepidostoma togatum		**X**	
Genus 1		**X**	
PHRYGAENIDAE			
Agrypnia		**X**	
Genus 1		**X**	
HYDROPTILIDAE			
Genus 1		**X**	
Genus 2		**X**	
DIPTERA			
TIPULIDAE			
Antocha		**X**	
Elliptera?		**X**	
Limonia		**X**	
Pedicia		**X**	
Phalacrocera		**X**	
Tipula		**X**	
Genus 1		**X**	
Genus 2		**X**	
SIMULIIDAE			
Helodon		**X**	
Parasimulium		**X**	
Prosimulium		**X**	
Simulium		**X**	
Genus 1		**X**	
CERATOPOGONIDAE			
Echinohelea lanei		**X**	
Stilobezzia elegantula		**X**	
Stilobezzia sp.		**X**	
CULICIDAE			
Aedes		**X**	
Anopheles		**X**	
Psorophora?		**X**	
CHAOBORIDAE			
Chaoborus (Chaoborus)		**X**	

DIPTERA (continued)	Parks APIS	ISRO	PIRO
CHIRONOMIDAE			
PRODIAMESINAE			
Prodiamesa		X	X
Monodiamesa		X	
Odontomesa			X
PODONOMINAE			
Parochlus		X	
TANYPODINAE			
Ablabesmyia	X	X	X
Conchapelopia		X	X
Helopelopia		X	
Labrundinia			X
Procladius		X	X
Thienemannimyia		X	
Zavrelimyia		X	
DIAMESINAE			
Diamesa	X	X	X
Pagastia		X	X
Potthastia		X	
Protanypus		X	
Pseudodiamesa		X	
ORTHOCLADIINAE			
Acricotopus			X
Brillia			X
Corynoneura	X	X	X
Cricotopus	X	X	X
Eukiefferiella	X	X	X
Heterotrissocladius	X	X	X
Limnophyes	X	X	X
Metriocnemus	X	X	X
Nanocladius		X	X
Orthocladius (Eudactylocladius)	X	X	X
Orthocladius (Euorthocladius)		X	X
Orthocladius (Orthocladius)	X	X	X
Orthocladius (Pogonocladius)		X	
Parachaetocladius			X
Paracladius		X	
Parakiefferiella		X	
Parametriocnemus	X		X

	Parks APIS	ISRO	PIRO
Parasmittia		X	
Psectrocadius (Allopsectrocladius)		X	
Psectrocladius (Psectrocladius)	X	X	X
Pseudorthocladius		X	
Pseudosmittia		X	
Rheocricotopus			X
Synorthocladius		X	X
Thienemanniella	X	X	X
Tvetenia	X		X
Orthocladiinae genus		X	
CHIRONOMINAE			
Tribe CHIRONOMINI			
Chironomus	X	X	X
Neozavrelia		X	
Cryptochironomus			X
Dicrotendipes	X	X	X
Endochironomus		X	
Glyptotendipes	X	X	X
Microtendipes			X
Parachironomus		X	X
Paratendipes			X
Polypedilum	X	X	X
Sergentia		X	
Tribe TANYTARSINI			
Micropsectra	X	X	X
Paratanytarsus	X	X	X
Stempellinella			X
Tanytarsus		X	X
PSYCHODIDAE			
Telmatoscopus		X	
STRATIOMYIDAE			
Allognosta?		X	
SCIOMYZIDAE			
Colobaea		X	
DOLICHOPODIDAE			
Campsicnemus		X	
Dolichopus		X	
Diostracus?		X	
Liancalus		X	

Macroinvertebrates

DIPTERA (continued)	Parks *APIS*	*ISRO*	*PIRO*		Parks *APIS*	*ISRO*	*PIRO*
Paraphaenocladius			X	*Paraphrosylus*		X	
DOLICHOPODIDAE (continued)							
Pelastoneurus		X					
Tachytrechus		X					
Telmaturgus parvus		X					
Thinophilus		X					
Xanthochlorus helvinus		X					
Genus 1		X					
EMPIDIDAE							
Hilara		X					
PHORIDAE							
Dohrniphora		X					
Megaselia		X					

Diatoms

	Parks				Parks		
	APIS	***ISRO***	***PIRO***		***APIS***	***ISRO***	***PIRO***
Chrysophyte cysts	X	X	X	*Halamphora*			X
Achnanthidium	X	X	X	*Hannaea*		X	
Adlafia			X	*Hantzschia*		X	
Amphipleura	X		X	*Hippodonta*			X
Amphora	X	X	X	*Karayevia*	X	X	X
Aneumastus	X		X	*Kobayasiella*	X	X	X
Asterionella	X	X		*Krasskella*		X	X
Aulacoseira	X	X	X	*Luticola*	X	X	
Brachysira	X	X	X	*Martyana*	X		X
Caloneis	X	X	X	*Mastogloia*			X
Cavinula	X		X	*Meridion*			X
Chamaepinnularia	X	X	X	*Microcostatus*		X	
Cocconeis	X	X	X	*Navicula*	X	X	X
Cyclotella	X	X	X	*Navicula (small)*	X		X
Cymbella	X	X	X	*Navicula schmassmannii*		X	X
Cymbopleura	X		X	*Neidiopsis*		X	
Decussata			X	*Neidium*	X	X	X
Delicata	X	X	X	*Nitzschia*	X	X	X
Denticula	X	X	X	*Nitzschia (plankton)*	X	X	X
Diadesmis	X	X	X	*Nupela*	X		
Diatoma	X	X	X	pennate GV unid	X	X	X
Diatoma mesodon			X	*Pinnularia*	X	X	X
Diploneis	X	X	X	*Planothidium*		X	X
Discostella	X	X		*Platessa*	X		X
Distrionella	X			*Psammothidium*	X	X	X
Encyonema	X	X	X	*Pseudostaurosira*	X	X	
Encyonopsis	X	X	X	*Puncticulata (large)*	X	X	X
Eolimna		X	X	*Puncticulata (small)*	X		X
Epithemia	X	X	X	*Reimeria*	X	X	X
Eucocconeis	X	X	X	*Rhopalodia*		X	X
Eunotia	X	X	X	*Rossithidium*	X	X	X
Fistulifera			X	*Sellaphora*	X	X	X
Fragilaria (plankton)	X	X	X	*Stauroforma*		X	
Fragilariforma			X	*Stauroneis*		X	
Frustulia	X	X	X	*Staurosira*	X	X	X
Geissleria	X		X	*Staurosirella*	X	X	X
Gomphonema	X	X	X	*Stenopterobia*		X	

Diatoms

	Parks		
	APIS	***ISRO***	***PIRO***
Stephanodiscus (large)	**X**	**X**	**X**
Stephanodiscus (small)	**X**	**X**	**X**
Surirella			**X**
Synedra	**X**	**X**	**X**
Synedra cyclopum	**X**		**X**
Tabellaria (long)	**X**	**X**	**X**
Tabellaria (small)	**X**	**X**	**X**
Ulnaria	**X**	**X**	**X**
Urosolenia	**X**	**X**	**X**

	Parks		
	APIS	***ISRO***	***PIRO***

Appendix E: Key to Larval Amphibians in Rock Pools at Isle Royale

The key developed for this project was primarily made from Altig and Ireland (1984), Altig et al. (2010), Parmelee et al. (2002), and Watermolen and Gilbertson (1996). Variation is common due to plasticity and it is expected that some specimens will not be identifiable to species level. Species in bold are known at Isle Royale; species underlined are expected to occur in rock pools. Specimens diverging strongly from the keys should be photographed from many angles, the pool should be described in notes including lat/long, and further investigation should occur when back in the office. If this keys out to a new species for Isle Royale, contacts with regional experts should be made to determine needs for the possible collection of a specimen.

1	External gills; front legs present as small larvae.	**Salamanders**	**2**
	Internal gills; front legs only when near metamorphosis.	**Frogs and toads**	**6**
2	Dorsal fin extending only to base of paddle-like tail. Gills short. Four toes on hind feet. Dark central dorsum stripe. White/yellow stripe on sides from nose to tail.	**Mudpuppy** *Necturus maculosus*	
	Dorsal fin extending onto body nearly to head. Gills long.	Salamanders/Newts	**3**
3	Dorsal yellow spots. **Dark stripe within eye** (Figure 17). Dark spots on tail. Head somewhat pointed in dorsal view and not large. Body slender. Skin sometimes granular in larger larvae. **Costal grooves absent**.	**Eastern/Central Newt** *Notophthalmus viridescens*	
	Without dorsal yellow spots. **No dark stripe within eye** (Figure 16). Broadly rounded head in dorsal view appears large on chunky body. Skin smooth. **Costal grooves usually distinct** (Figure 2). Chin without markings.	Ambystomatids	**4**
4	Small larvae (<60mm) lack balancers between eyes and gills, and has distinct dorsal and/or lateral black spots. Toes flat and pointed in large larvae. Ground color mottled or uniform gray, costal grooves often pigmented. Reaches very large size (>100mm)	Tiger Salamander *Ambystoma tigrinum*	
	Small larvae with balancers between eyes and gills (Figure 1). Toes rounded. Total length usually between 45–70mm.	Other Ambystomatids	**5**
5	Larvae between 12–50mm. Small larvae yellow-green or brown with dark mottling on body. Larger larvae with a row of small yellow spots and a light lateral stripe. No paired dorsal spots.	Spotted salamander *Ambystoma maculatum*	
	Larvae between 8–30mm. **Dark mottling on tail fins.** Midlateral row of separate or partially fused light spots. **Dark line may extend from snout to fins, but not within eye** (Figures 2 and 16).	**Blue-spotted salamander** *Ambystoma laterale*	

6	**Eyes dorsal** when viewed from above (Figure 15)	*Bufo* and *Rana*	**7**
	Eyes lateral when viewed from above (Hylidae family)	*Pseudacris* and *Hyla*	**12**
7	Vent medial. **Very dark color** often with golden spots. Small <25mm. **Fin clear**, round at end. Tail musculature bicolored (white below, gold speckling on upper edge).	**<u>American Toad</u>** *Bufo americanus* (Figure 3)	
	Vent dextral (exits to side, not middle).	*Rana spp.*	**8**
8	White line from nostril to snout. General color uniform but variable with considerable mottling. Fins variable from clear to boldly marked with fine spots, forming low to medium arch. Iris dark at four compass points (Figure 4). Intestinal coil visible.	Northern leopard frog *Rana pipiens*	
	No white line from nostril to snout.	Other *Rana spp.*	**9**
9	**Tail without spots** or mottling (or only faint small marks). Dorsum uniformly dark and speckled in gold. Tail short, high, and lighter than body. 6-9 weeks from egg to emergence. **Intestinal coil visible**.	**<u>Wood frog</u>** *Rana sylvatica* (Figure 5)	
	Tail with spots or mottling.	Other *Rana spp*.	**10**
10	**Tail with light pinkish buff spots**. Body brown to bright green, belly buff. Small black dorsal spots or mottling. Fins lightly speckled, but no black rectangular markings on distal edge. Large tadpole.	**Mink frog** *Rana septentrionalis* (Figure 6)	
	Tail with dark spots, without pinkish spots.	Other *Rana spp*.	**11**
11	**Intestinal coil not visible.** Tail with fine yellow spots and low arch. Dark or pale brown with **dense fuzzy dots/vermiculations**. Dorsal fin edge has no spots but **distal 1/3 has dark rectangular marks**. Large tadpole.	**<u>Green frog</u>** *Rana clamitans* (Figure 7)	
	Intestinal coil visible. Tail without fine yellow spots. Iris with spots in cardinal directions. Ground color light with purple-black flecks. Tail uniformly speckled. Prefers slow areas of rivers and streams. (Figure 8).	Pickerel frog *Rana palustris*	
12	**Tail often marked with large black blotches, including area near tail musculature. Fins may be tinged red**/orange/yellow. Color and pattern extremely variable. Tail ends in clear flagellum (figure 9). (Not confirmed at ISRO, if this species is observed please photograph!)	**Gray treefrog** *Hyla versicolor*	

Tail generally not strongly marked, no flagellum at tail tip. Tadpole ≤1.2in. *Pseudacris spp.* **13**

13 **Fins clear**, but if spots or blotches present there is a **clear area near tail** (figures 10, 11). Tail dorsum may be crudely banded in younger stages, uniformly dark, or with irregular pale area over surface; often blotchy in large tadpoles. Tail musculature may be bicolored. Throat speckled. Dorsum uniformly medium brown to semi-transparent (depending on turbidity) with copper specks. Egg to adult in two months. **Spring peeper** *Pseudacris crucifer*

Fins clear with dark speckling. **Tail dorsum uniformly dark** above (in clear water) or light (in turbid water), often **distinctly bicolored**. Throat unpigmented. May also appear to have copper speckling. Three months egg to adult. Metamorphs and adults at ISRO often strongly mottled (figures 12, 13 and 14). **Chorus frog** *Pseudacris triseriata*

Figure 1. Balancers and external gills on a spotted salamander (*Ambystoma maculatum*) larva. Photo: Minnesota DNR.

Figure 2. Blue-spotted salamander (*Ambystoma laterale*) larva showing costal grooves and mottled coloration. Photo: Paul Brown.

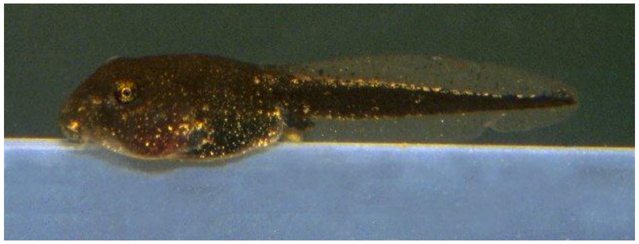


Figure 3. American toad (*Bufo americanus*) larva showing clear tail fin and dark body. Photo: Altig et al. (2010).

Figure 4. Northern leopard frog (*Rana pipiens*) larva. Photo: Altig et al. (2010).

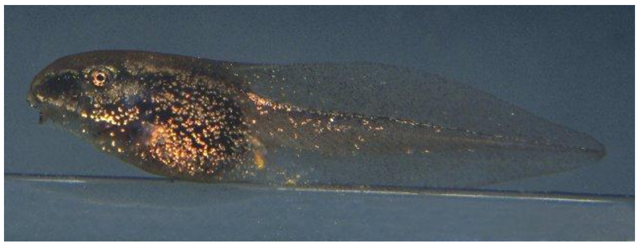


Figure 5. Wood frog (*Rana sylvatica*) larva. Photo: Altig et al. (2010).

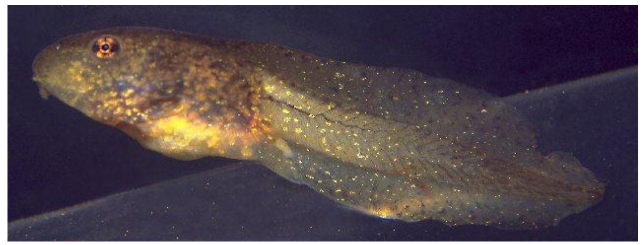


Figure 6. Mink frog (*Rana septentrionalis*) larva. Photo: Altig et al. (2010).

Figure 7. Green frog (*Rana clamitans*) larva. Photo: Altig et al. (2010).

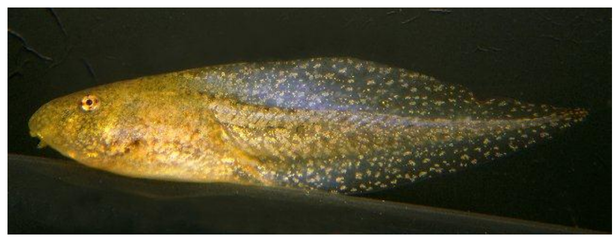


Figure 8. Pickerel frog (*Rana palustris*) larva. Photo: Altig et al. (2010).

Figure 9. Gray treefrog (*Hyla versicolor*) larva. Photo: Altig et al. (2010).

Figure 10. Spring peeper (*Pseudacris crucifer*) larva. Photo: Altig et al. (2010).

Figure 11. Spring peeper larva, showing clear area on tail near musculature, lateral eyes, and bicoloring in tail. Photo: Alex Egan.

Figure 12. Chorus frog (*Pseudacris triseriata*) larva. Photo: Altig et al. (2010).

Figure 13. Chorus frog tadpole showing darkened dorsal tail musculature and lateral eyes. Photo: Paul Brown 2010.

Figure 14. Chorus frog metamporph showing strong mottling. Photo: Alex Egan 2009.

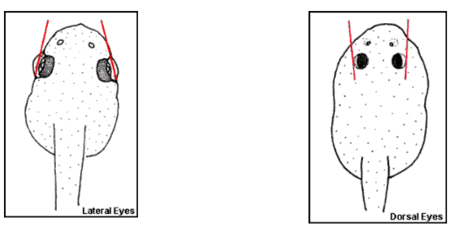


Figure 15. Lateral (left) and dorsal (right) eye configurations. From Altig et al. (2010).

Figure 16. Blue-spotted salamander larva showing mottling on tail fin, dark patches on dorsum, lack of pigmentation on throat, but also showing stripe through eye that does not include the eye. Photo: Alex Egan.

Figure 17. Eastern newt (*Notophthalmus viridescens*) larva showing dark stripe in eye (note: stripe is part of the eye) and black speckles. Photo: Alex Egan.

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Altig, R., and P. H. Ireland. 1984. A key to salamander larvae and larviform adults of the United States and Canada. Herpetologica 40(2): 212-218.

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Parmelee, J. R., M. G. Knutson, and J. E. Lyon. 2002. A field guide to amphibian larvae and eggs of Minnesota, Wisconsin, and Iowa. Information and Technology Report 2002-2004. U.S. Department of Interior, U.S. Geological Survey. Washington, D.C

Watermolen, D. J., and H. Gilbertson. 1996. Keys for the identification of Wisconsin's larval amphibians. Wisconsin Endangered Resources Report #109. Wisconsin Department of Natural Resources, Madison, Wisconsin.

Appendix F: Coastal Rock Pool Monitoring Protocol

Standard Operating Procedures

Prior to designing a project, the goals, objectives, and questions to be answered must be defined based on the needs of managers. Giving guidelines for establishing a long-term monitoring program is the objective of these standard operating procedures (SOPs), with a focus on Chironomidae, diatoms, and water chemistry sampling. Monitoring does not need to occur annually, and could occur as a broader effort between many parks; both strategies will help save limited park resources, and having an established rock pool team that can move from park to park is likely to be more effective than training teams in each park. As summarized in Murray et al. (2002) for marine coastal studies, monitoring should include multiple sites over long time scales, predetermined conditions that signify impact or change, use of ecologically important indicator species to represent a community, and use of statistically valid methods and analyses (including indices) to monitor a system.

Many of the methods for preparing and identifying diatoms, zooplankton, and chironomid exuviae will be challenging for non-experts or organizations without lab space or appropriate equipment. For exuviae, while sorting and subsampling may be accomplished by trained field staff using a dissecting microscope, these tasks may be more appropriate for the crew leader or project manager, who should have knowledge about exuviae traits and differences. Slide mounting and identifications of exuviae are possible with practice and using keys in Wiederholm (1986) and Ferrington et al. (2008) (see SOP 5c for full citations). In-park expertise on these groups would take time to accrue and should be done either by the project manager or samples should be sent to an appropriate expert in chironomid taxonomy. In addition to the benefits from taxonomic expertise, time will likely be saved if experienced personnel accomplish these tasks. For a novice, slide mounting a season's worth of samples may take 7-8 weeks, and identifications may take months with at best a moderate level of certainty for challenging groups. With experience, slide mounting time can be dramatically shortened and an expert can often identify a season's worth of identifications to genus within a week or two.

Index to Standard Operating Procedures

STANDARD OPERATING PROCEDURE 1 – Personnel Requirements, Training, and Sampling Safety Guidelines

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

Within the coastal rock pool project, one or many people will be responsible for study design, data collection, data entry and analysis, and reporting. An outline of the qualifications required, roles and responsibilities of personnel is described, along with annual training and safety guidelines for sampling and specimen processing. Descriptions of these roles are partially informed by NPS Great Lakes Network standards (Hart and Gafvert 2006). Parks often have limited personnel budgets, but it is suggested that the project manager/principal investigator has from one to three assistants depending on the scope of the implemented protocol. Many important roles may be filled by personnel without direct connection to the project, and important working relationships established with additional people: resources specialists, GIS specialists, data managers, curators, statisticians, and ecologists. Training and safety considerations are partially adapted from Long and Mitchell (2010).

Personnel

Project Crew Member: These people will take part in data collection in the field and data input in the office. They may also be part of data verification, laboratory work, data analysis, and report writing. Crew members can be interns, seasonal employees, or local volunteers. All crew members should have some type of biology or ecology background, prior knowledge of the taxa of interest, and must be physically fit for the demands of field work. All personnel must be able to commit the time to learn procedures and accomplish annual tasks. Eager volunteers with no experience can, at a minimum, accompany field staff and record information on datasheets.

Project Crew Leader: This person will have a strong understanding of all aspects of the project and will guide annual training for crew members. The crew leader will generally be present during all aspects of field and lab work, and will have initial responsibility for data analysis, annual reports, and ensuring protocols have been followed. A crew leader should have one year of experience on the project, or experience on similar projects with a relatively strong initial understanding of taxa that will be surveyed. Scientific field skills are preferable and in some parks boat training will be mandatory.

Project Manager: Ultimately, this person is responsible for managing the entire project, ensuring that all needs are met and procedures are followed and completed. The project manager will ensure the crew leader and crew members are adequately trained and informed of changes. The Project Manager will communicate to and between crew personnel, park managers, and collaborating investigators. They will guide report writing and be responsible for synthesis reports and protocol reviews, often in conjunction with specialists such as an ecologist or statistician. They should be familiar with all methods and be able to accomplish all tasks as needed. Managers will ensure data are annually validated and archived. The project manager must be familiar with methods used in the protocols, have appropriate field work experience, know the study area(s), and have some expertise in the taxa of interest.

Workloads

Workload will vary based on the number of sites and samples chosen for annual collections. At a minimum, one sample in spring and one sample in mid-summer are suggested, while monthly sampling is ideal. A minimum of four sample sites is suggested.

Crew members should be expected to work on the field component of a rock pool project for 6-to-12 weeks (0.12-0.25 Full Time Equivalent) during a field season, including training, preparation of gear, site visits, and data entry.

Crew leaders should be expected to work approximately 12 weeks (0.25 FTE), including training, field work, QA/QC, and report writing.

Project manager duties may include those of the crew leader. In addition, 1-2 weeks (0.02-0.04 FTE) may be included for logistical support, hiring, review of reports, and public outreach.

Sample processing of exuviae (picking, sorting, and slide mounting) for a given season may take an additional six or more weeks, though this can be split between two or more people. If other macroinvertebrates are included as targets, picking will occur at the same time as exuviae but sorting may take extra time (up to one week) depending on how many taxa are involved. Identifications of slide-mounted specimens will be very time consuming for non-experts, and some groups can be very challenging to identify to genus, even with good keys like Merritt et al. (2008). For a long-term study, project managers may be interested in training by experts so that in-house identifications can be done with voucher specimens sent to experts for confirmation.

Annual Training

Prior to the field season, the project manager will plan training for all employees or volunteers involved in field work. All personnel must read (or re-read) the protocols and discuss any uncertainties with the project manager. A practice event at a non-study site (to avoid biasing sampling results) should include all employees so everyone receives the same information. While collecting practice data, explanations of how data will be analyzed may help in understanding the purposes behind each step in field work.

All government boat operators must be scheduled to receive the DOI Motorboat Operator Certification Course (MOCC) training and should practice correct beaching techniques with an experienced operator if necessary. Training must encompass enough time that all personnel can observe, practice, and ask questions about procedures. Note-taking during these processes should be encouraged.

Field training for all personnel should include:

- Safety considerations, weather and clothing;
- An overview of the coastal rock pool ecosystem;
- How to limit impacts to the habitat during sampling;
- Explaining maps of landing locations, trails, backcountry routes, and the study sites (this may include a map and compass training/refresher);
- Using field keys to identify organisms of interest and how to describe or photograph unknowns;
- Use of GPS units, range finders, radios, and other equipment;
- Data entry on field forms; and
- Finding permanent study pools, and the procedures for surveying them.

Office training should include how data will be transferred, downloaded, saved, analyzed, and reported. This may include reading software manuals, reviewing previously entered data or reports, and covering QA/QC procedures. During the entire training process, the importance of carefully following SOPs must be noted regularly so that data have a high integrity (Long and Mitchell 2010).

Safety Guidelines

Safety is the first issue that must be considered when sampling – if personnel do not feel conditions are or will be safe, field work should be delayed until conditions improve. While working in the field, safety procedures for respective parks must be followed at all times. Prior to daily field work, on the morning that work is planned, personnel will determine safe weather conditions based on **local** National Weather Service marine forecasts obtained online or via the morning report on park radio.Landing a boat on exposed shorelines and leaving it alone while mapping should only occur when conditions are calm and **forecast to remain calm**. All boat operators should have NPS boat training (MOCC, which is required if operating a park boat), and if sites are unfamiliar to new personnel, an experienced boat operator for the area should be consulted prior to departing for the study sites. All boats must have nautical charts of the study area and safety equipment as designated by the park and U.S. Coast Guard.

For daily field work in which the crew will return to a housing unit or headquarters, at least one park radio with an extra battery and radio call list must be brought to the study site and monitored during work (not left in the boat). For non-park personnel, a cell phone may be an acceptable alternative at some parks, provided it is charged daily and signal strength is good. Most park radios are water resistant, but cell phones should be stored in a waterproof bag. Use of a dispatch service is often required for park personnel, and an expected arrival time should be given to dispatch so a response can be initiated if the time is missed. **For a life-threatening medical emergency call local rangers, dispatch (if available), or 9-1-1.**

For extended, overnight mapping trips, an entire trip plan must be submitted and two radio contacts be identified for a daily check-in. A trip plan must be verbalized to a supervisor, ranger working in the area, or dispatch service, with a daily expected time of return to camp and predefined response in case the check-in or return time is missed. Any changes must be clearly communicated during check-in times or as they occur.

Weather

No sampling or boat travel will happen if thunderstorms or high winds are expected during the day. Weather events can happen much sooner than forecasts predict, so remain constantly aware of changing conditions. Sites are selected with safe collecting in mind and most sampling must take place during relatively calm conditions. However, conditions on Lake Superior can change quickly, which is particularly important when personnel are busy with tasks and may not notice incremental shifts in wind or weather conditions. Weather is a primary issue for safety because Lake Superior is cold, a wind shift can create unsafe collecting or boat beaching conditions, and rain during cool weather can make hypothermia a real threat.

An important part of safety is taking breaks, which reduce both mental and physical fatigue. Personnel can discuss interesting events during sampling, and breaks allow time to stop and take note of weather or other local conditions. Fifteen-minute breaks in the morning and afternoon, along with a half hour lunch break, should always be taken. During hot weather, take extra breaks and/or start early in order to end field work early.

Clothing

In addition to weather, the rocky shores themselves have dangerous conditions. Slipping on wet, gravel-covered, and uneven bedrock can be a source of physical trauma. Pay attention to surroundings and where you are placing your feet. All injuries must be reported to a supervisor as soon as possible, and NPS employees will be required to complete the first step in worker’s compensation paperwork. This documentation is important in case an apparently minor injury becomes major or persists and may be important to guarantee compensation coverage.

Daily weather may not conform to expected weather conditions, even when using a local marine forecast. Personnel should wear or pack appropriate clothes, including rain gear, long pants and long-sleeved shirt for sun and insect protection, a hat with a wide 360° brim, and boots with good tread. Layers are important – a cold, calm morning often becomes a warm, windy afternoon on Lake Superior. Synthetic or wool clothing is preferable to cotton for all field work. Knee pads are important if time is spent looking into pools. Sunscreen and insect repellant should be used **only** if hands and arms will not enter the water. Physical sun protection (large hat, bug net, long-sleeved shirt, and gloves) should be worn if hands will or may enter water, as sunscreen, lotions, and insect repellant can have chemicals that are harmful to aquatic invertebrates and amphibians. Finally, adequate drinking water or a water filter must be carried; windy and sunny conditions are common in these habitats and increase hydration needs.

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STANDARD OPERATING PROCEDURE 2 – Field Season Preparation and Equipment Setup

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

As necessary, scientific research and collection permits for each applicable agency or organization must be obtained prior to the field season. For example, the NPS requires investigators to complete the Research Permit and Reporting System process and receive permission from park managers to collect biological material and remove it from the park.

The project manager must also create the schedule for field and lab time, such as:

Annual Field Schedule

- February – Check equipment and gear lists to ensure all is present and in good condition; purchase new equipment as needed
- April – Field staff training
- April to May – Print field forms; begin field sampling
- June to October – Continue field sampling as frequently as required; use extra time for mapping, data entry, QA/QC
- September to October – Data entry, QA/QC; prepare and ship samples to contracting lab; prepare draft of report including field observations
- November to December – Complete annual report after receiving results from lab

Equipment

The field, laboratory, and mapping SOPs document the equipment needed for completing tasks (Figure F1). Two months before the field season begins, these lists should be checked against gear stored from the previous season to ensure all gear is present and in working condition. Make sure to check battery quality for all equipment that uses them.

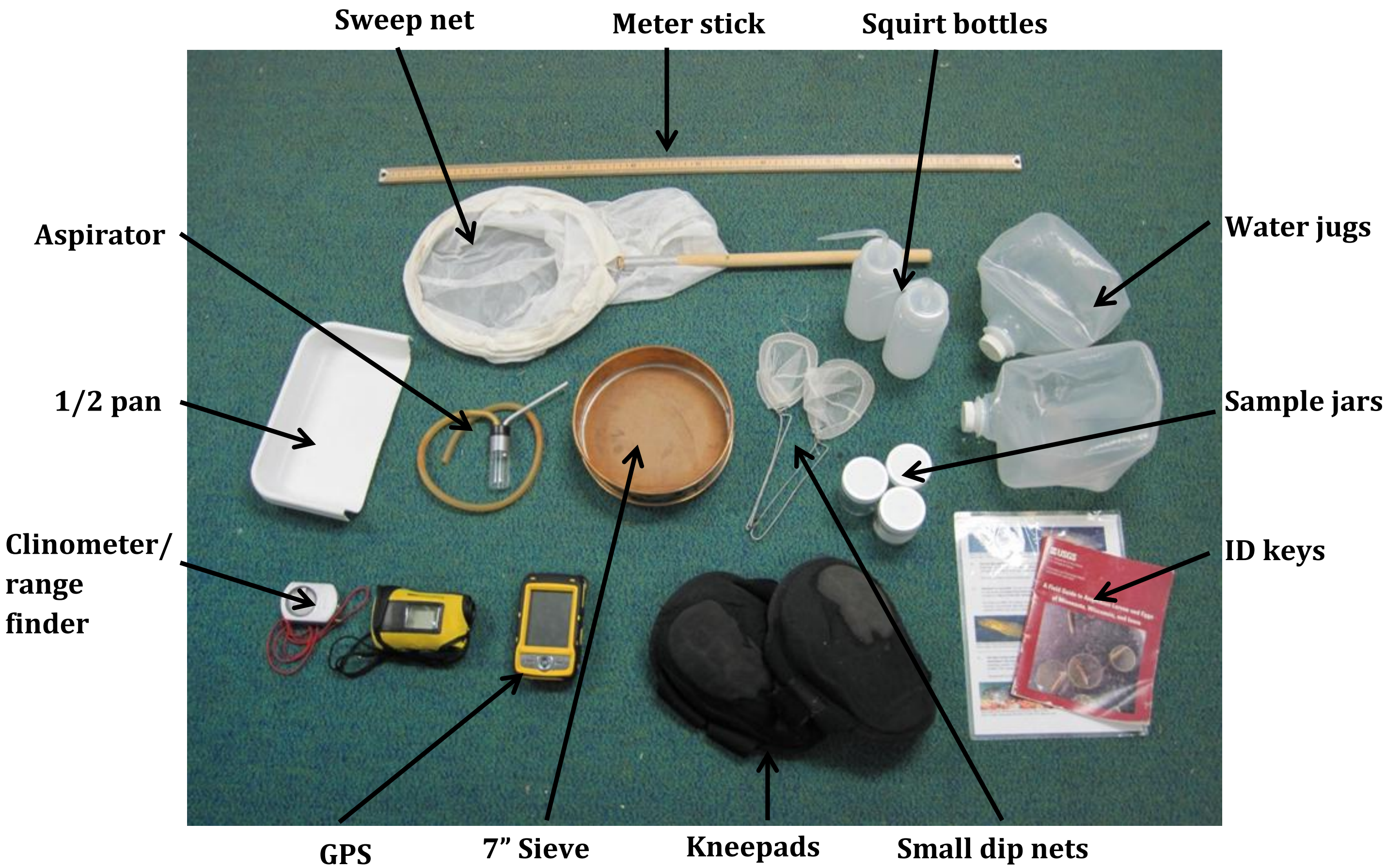

Figure F1. Selected equipment used for macroinvertebrate rock pool sampling and mapping.

STANDARD OPERATING PROCEDURE 3 – Determining and Establishing Sample Sites

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

Planning site selection will help ensure success during long-term studies. Site selection should begin with assessments of high-quality aerial imagery, if available. Large areas can be quickly viewed and general site quality for rock pool habitat can be assessed. If different types of bedrock, exposure to waves, aspect, slope, or other physical conditions exist, a stratified or discriminating site selection can take these into account depending on study objectives. Comparing sites may not be valid if the above conditions are not similar, but dissimilar sites may be of interest for general community assessment across many shoreline types (see Murray et al. 2002 for additional details).

Once a list of potential high-quality sites is made, sites should be randomly selected for visits to assess monitoring potential. Randomization may be stratified to ensure that various ecological conditions are included. Sites may be rejected for many reasons, including lack of adequate habitat for sampling, proximity to other sites, presence of wildlife such as gull colonies that would be disturbed on each visit, and lack of safe access. Project objectives will help determine adequate habitat. If geographic mapping has been done for the areas of interest, sites and pools can be randomly chosen from those data.

Individual pools can be selected by walking the site and randomly choosing pools with dice or a random number list. At locations with dense pools, shorelines may need to be divided into smaller segments. Segments are then chosen randomly before selecting pools within the segments.

Criteria

- Site conditions meet project objectives, such as number of pools of certain types or human visitation/impact patterns
- Site visits will not impact wildlife or sensitive plant communities
- Sites are not too close to other sites and are not pseudoreplicates of each other (e.g., two sites on a small island)
- Conditions are safe for personnel to access and sample at sites during the field season

After pool selection, pools should be marked physically if they will be sampled over time. Markings can be cairns built of stone from the nearby area, if appropriate and if visitors are not likely to disturb them. A waypoint should also be plotted on a map or aerial photo.

Literature Cited

Murray, S., R. Ambrose, and M. Dethier. 2002. Methods for Performing Monitoring, Impact, and Ecological Studies on Rocky Shores. University of California Press, Berkeley, California.

STANDARD OPERATING PROCEDURE 4 – Macroinvertebrate Sample Collections

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

This SOP establishes procedures for collecting macroinvertebrate samples from coastal rock pools. All samples will be collected and treated in the manner described below. The focal group for this SOP is Chironomidae, although dip net and sweep net collections will detect other aquatic groups. Monthly collections between late April and October should be adequate to detect the majority of taxa present. Collections will only occur when there has been no heavy precipitation or strong wave action along shorelines at study sites for ≥24 hours, as pupal exuviae may sink during rainfall and material may be washed out of lower pools by wave action.

Six samples will be collected during each monthly site visit. In the "lichen zone," two larger, permanent pools will be designated for sampling throughout the season, with smaller, ephemeral pools in the area sampled opportunistically and lumped into a single ephemeral zone jar for each site visit. This strategy will be repeated for "splash zone" pools. Opportunistic sampling of ephemeral pools will avoid problems of pool desiccation if rainfall is limited.

Due to apparent community simplicity, only a small number of live specimens should be necessary for collection. Adult chironomids will be collected on a limited basis to compare with surface exuviae, while subsurface taxa (e.g., Trichoptera, Hemiptera, Coleoptera, Culicidae) will be collected only as necessary for identification of a species that appears morphologically or behaviorally different from previously collected taxa.

The number of materials listed below would allow decontamination to occur at a headquarters or other location. Fewer numbers of dip nets, sieves, or trays will require on-site decontamination to avoid moving organisms or pathogens between pools. Decontamination in the field can add an additional hour or more per site visit.

Materials (per site based on 2010 methods)

For collecting

_ Six sample collection jars
_ Six small aquarium dip nets
_ Six 7", 125µm sieves (No. 120)

_ Six pans with at least one shallow edge (8×10 photo developing trays cut in half)
_ Squirt bottle for tap water (mark clearly)
_ Squirt bottle for ethanol (mark clearly)
_ One liter of 80% ethanol
_ Sweep net for aerial capture
_ Aspirator
_ Six pair of tweezers/foreceps
_ Clipboard with datasheets
_ Pre-cut paper squares for site data
_ Watch or timer
_ Pencils

For decontamination, if needed

_ Four 1-gallon jugs with tap water (e.g., clean orange juice or milk jugs)
_ Large pot (bigger than the 7" sieve)
_ Campstove with extra fuel and aluminum wind screen
_ Tongs for handling hot gear

Other

_ Cooler pack for sample jars and liquids
_ Backpack(s) for gear
_ Handheld park radio with extra battery (or cell phone)
_ GPS unit with extra batteries

Methods for Macroinvertebrate Sample Collections

(1) Prior to leaving headquarters, go through materials checklist and make sure all equipment is present and in good condition. Fill all water jugs and ethanol container.

(2) Upon arrival at study site, set up a workstation near pools but where water from washing equipment will not flow into any pools. Windy conditions will often occur and rocks will be needed to prop-up the decontamination pot, so find a good site that can be used during subsequent visits. To help find permanent pools that are sampled during every visit, build a small cairn of local rocks; however, the cairns may be knocked over by waves at splash zone pools. If habitat is dense and a permanent, regularly sampled pool is not unique from nearby pools, a photograph and waypoint should be taken in order to relocate the same pool.

(3) Record sample number (see SOP 5d on labeling), site name, and personnel last names on labels of all six jars (four for permanent pools, two for ephemeral pools). In pencil, which will not wash off in ethanol, record the sample number on a small piece of paper and place in each respective jar.

(4) At each permanent pool, approach slowly and watchfully. With the sweep net and/or aspirator (with a small amount of EtOH in jar to kill insects), collect representative examples of midges above, on, or near the pool. Watch for very small midges on the pool's surface or edge, and for swarms above head level. Larger surface midges or other flies can sometimes be captured with aquarium nets quickly flipped on top of them. Place any captured insects into the appropriate sample jar with ½-inch of 80% ethanol in the bottom. This step should take ≤5 minutes.

(5) Use a pan to collect surface exuviae and dead adult insects by dipping the shallow, cut edge of the pan into the water and allowing surface material to flow into the pan. Pour material into the sieve. Continue this process around the pool for five minutes, sampling from the entire surface if possible.

(6) Use the squirt bottle with tap water to rinse material in the sieve to one side (do not use lake or pool water since small exuviae or other organisms may get into the bottle). Washing material from both the top and bottom of the sieve mesh can help move material, but care should be taken when washing from the bottom side so material isn't ejected from the sieve. Allow water to drain.

(7) Remove cap from the appropriate sample jar and place jar on a flat surface. Tip sieve over the open jar. Use the squirt bottle with 80% ethanol to rinse material from the sieve into the jar. Use tweezers to carefully pull stuck material off sieve mesh and into jar. Be careful to use a minimal amount of ethanol and not overfill the jar.

(8) Carefully observe the pool for several minutes (shorter for small pools, longer for large pools). With a small dip net and tweezers, collect representative examples of all morphologically unique invertebrates in the pool and place in sample jar. (Note: individuals perceived to be of a common and previously collected group may be described in field notes without collecting them.)

(9) Fill the invertebrate sample jar with ethanol to ¼-inch from top. Replace cap. Fill out datasheet.

(10) Repeat steps 3–9 for the other pools. Use a sterilized pan, sieve, dip net, and tweezers for each new pool. If there are fewer than six replicates of collection supplies, use boiling water to sterilize gear for at least 3 minutes before use in new pools.

(11) For ephemeral zone samples, walk the area (either lichen or splash zone) around and between the two permanent pools for ten minutes, looking for small, shallow, ephemeral pools. These pools will vary in surface area but must have a depth between one and four inches. As in step 4 above, collect surface material at each pool for 15–30 seconds, pouring material into the sieve, before looking for another small pool. Standardize effort by panning for 10 total minutes. All material from ephemeral pools will be combined into the same sieve and rinsed into the same jar. If there is uncertainty about which zone a pool belongs to, or whether it is

permanent or ephemeral, skip that pool. Equipment does not need to be sterilized between ephemeral pools at the same site within the same zone.

(12) There should be six jars with invertebrate specimens, one for each of the four permanent pools and one for ephemeral pools in each zone.

(13) Fill out datasheets. Collect gear and go through checklist to account for all gear.

(14) Upon return to headquarters, place all sample jars in storage boxes after checking each lid for a tight fit and that labels are all correct. Sterilize sampling equipment as needed (dip nets, tweezers, sieves, pans). Download all photos or logger data and save backups.

Prior to daily fieldwork: Each morning that mapping is planned, personnel will determine safe weather conditions based on National Weather Service marine forecasts obtained online or via the morning report on park radio.Landing a boat on exposed shorelines and leaving it alone while mapping should only occur when conditions are calm and forecasted to remain calm. For extended mapping trips, an entire trip plan must be submitted and two radio contacts be identified for a daily check-in. A trip plan must be verbalized to a supervisor, ranger working in the area, or dispatch service, with an expected time of return and predefined response in case the check-in or return time is missed.

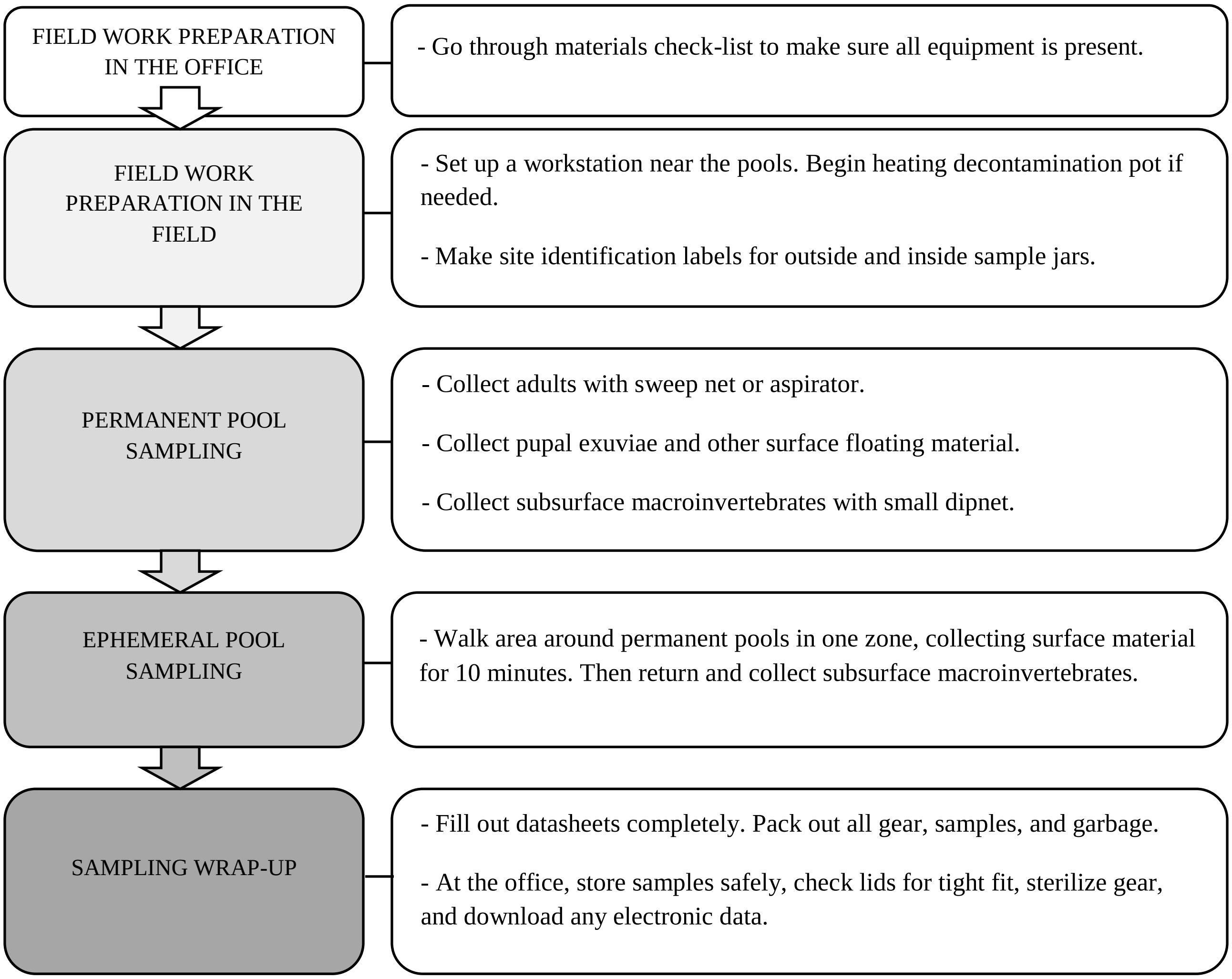

Figure F2. Visual model for rock pool sampling.

Coastal Rock Pool Survey Macroinvertebrate Form (Version 1.00)

Survey site:	GPS Coordinates: N W NAD83 or WGS 84			
Site description:				
Zone (Lichen/Splash/Transition):	Pools fed by rain/splash/runoff/seep:			
Distance to lake:	Distance to soil/vegetation line:			
Multiple pools surveyed?:	Length/width/depth of pool(s):			
Vegetation along pool edges:				
Survey date:	Sample number:			
Personnel: Time:	Weather – Wave height: Wind speed/direction: Cloud cover:			
Recent rain or large waves (include wave direction):				
Air temperature:	Water temperature:			
Taxa observed using pool habitat	Adults	Exuviae	Larvae	Notes (include abundance: rare, common, abundant)
Collembola (springtails)				
Ephemeroptera (mayflies)				
Anisoptera (dragonflies)				
Zygoptera (damselflies)				
Trichoptera (caddisflies)				
Plecoptera (stoneflies)				
Hemiptera – *Gerridae* (water striders)				
Corixidae (water boatmen)				
Notonectidae (back swimmers)				
Coleoptera – *Gyrinidae* (whirligig beetles)				
Dytiscidae or Hydrophilidae (diving beetles)				
Tipulidae (crane flies)				
Culicidae (mosquitoes)				
Simuliidae (black flies)				
Chironomidae (midges)				
Others				
Notes:				

Coastal Rock Pool Survey Macroinvertebrate Form (Version 1.00) *continued*

Site information on front of form				
Survey date:	Sample number:			
Personnel: Time:	Weather – Wave height: Wind speed/direction: Cloud cover:			
Recent rain or large waves (include wave direction):				
Air temperature:	Water temperature:			
Taxa observed using pool habitat	Adults	Exuviae	Larvae	Notes (include abundance: rare, common, abundant)
Collembola (springtails)				
Ephemeroptera (mayflies)				
Anisoptera (dragonflies)				
Zygoptera (damselflies)				
Trichoptera (caddisflies)				
Plecoptera (stoneflies)				
Hemiptera – *Gerridae* (water striders)				
Corixidae (water boatmen)				
Notonectidae (back swimmers)				
Coleoptera – *Gyrinidae* (whirligig beetles)				
Dytiscidae or Hydrophilidae (diving beetles)				
Tipulidae (crane flies)				
Culicidae (mosquitoes)				
Simuliidae (black flies)				
Chironomidae (midges)				
Others				
Notes:				

	Date	Initials		Date	Initials
Field Review			10% QA/QC		
Data Entered + Backed -up			Data Validated		
Data Entry Check					

STANDARD OPERATING PROCEDURE 5a – Picking and Sorting Organisms from Pan and Dip Net Samples

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

This SOP establishes procedures for picking (separating material of interest from algae and detritus) and sorting macroinvertebrates from pan and dip net samples collected from coastal rock pools. Picking and sorting are combined due to the relative lack of algae and detritus in most samples. All macroscopic aquatic insects will be sorted by order or family, while all detritus, partial specimens and non-target taxa will be returned to the original sample jar for potential future use and QA/QC. Prior to sorting, workers must have a basic knowledge of the morphology and traits of the taxa of interest; it will save time to sort material to the finest resolution possible while picking.

Materials

_ Original sample jar
_ White plastic sorting tray (photography tray without ridges)
_ 2 containment trays
_ 2 sieves with 125 micron mesh size (3” sieve usually adequate)
_ Squirt bottle for tap water (mark clearly)
_ Squirt bottle for 80% ethanol (mark clearly)
_ Dissecting microscope with light source
_ One-dram (3.697 mL) snap top vials (best size for most samples)
_ Seven-dram (25.879 mL) snap top vials (for large specimens)
_ Fine-tipped forceps (2 pair) and probe
_ Syracuse dish or petri dish/watch glass
_ Labels
_ Pencils

Methods

(1) Place the first 125 micron sieve into the first containment tray. Remove lid from the original sample jar and check lid for attached organisms. With the squirt bottle containing 80% ethanol, gently rinse any organisms from inside the lid onto the first 125 micron sieve, using

as little EtOH as possible. Locate and remove label from inside the sample jar with forceps. Gently rinse any organisms attached to label onto sieve. Set label aside.

(2) Separate the preservative from the contents of the original sample jar by pouring the sample onto the first 125 micron sieve. With the squirt bottle containing 80% EtOH, gently rinse any organisms and detritus from the inside of the jar and jar threads onto the sieve, using as little EtOH as possible.

(3) Pour preservative from containment tray back into the sample jar. Screw lid onto jar and set aside. If necessary, dispose of excess EtOH properly.

(4) Gently rinse material in sieve with water in a squirt bottle to remove residual EtOH. Then rinse material into one side of the tilted sieve where it can be rinsed into the Syracuse/petri dish, either all at once for a small sample or a little at a time for a large sample. Try to keep only a thin layer of water in the dish and swirl sample to evenly disperse material. Too much water will require continual refocusing through the depth of field and will take more time.

(5) Under the dissecting microscope, scan the contents of the dish and carefully remove all taxa of interest using fine-tipped forceps and a probe. Clean material of detritus and algae while sorting material into one dram (3.697 mL) vials (larger vials may be needed for odonates, belostomatids, and other larger insects). For projects using Chironomidae, different life stages can be separated into vials for later slide mounting and identifications.

(6) After removing all target organisms, swirl the material and re-inspect the contents for any remaining insects or exuviae. If additional insects or exuviae are found, place them into their respective jars. Re-swirl and re-inspect for target organisms. Repeat this step until no additional organisms are found on two consecutive swirl-and-inspection events. During inspections, it may be helpful to establish a standard pattern, such as a back-and-forth grid procedure, to ensure that no area of the Syracuse dish is missed.

(7) Place the sieve back into the containment tray. Rinse residual material (detritus, algae, non-target specimens) from the dish onto the sieve. If the sample is large and only a portion is removed from the sieve at any one time, rinse the residual material onto a second sieve in a containment tray.

(8) Repeat steps 5 through 7 until all material in the sample is picked and sorted (or until four hours have elapsed for very large samples).

(9) After the sample has been entirely processed, rinse residual material into the corner of the sieve with water from the squirt bottle and allow residual water to drip into tray. Using the 80% EtOH squirt bottle, flush residual material back into the original sample jar, using a minimal amount of EtOH. Use forceps to move large material without using EtOH. Fill jar with EtOH until it is 1cm from the top. Place original label back into original sample jar and replace cap. Dispose of excess EtOH properly.

(10) On the outside label of the original sample jar, write “Sample Sorted,” your initials, and date (numeric day, alpha month, numeric year – e.g., “07 Jan 2010”). Place a new label into each of the sorted specimen vials and fill each vial to 80% capacity with EtOH. Fill-out the sample processing log.

STANDARD OPERATING PROCEDURE 5b – Determining if Subsampling is Required, and Subsampling Procedure

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

Taxa of interest, particularly Chironomidae adults or exuviae, may be very abundant in pan or sweep net samples collected from rock pools. This SOP establishes procedures for determining when specimens of these groups are abundant enough to require subsampling. In addition, this SOP establishes procedures for subsampling Chironomidae exuviae when they are abundant. This process will also offer a chance to identify unique specimens that were not detected during the sorting process, which is a primary goal of the rock pool project. (Note: the cut-off of 20 Chironomidae and 16 grid squares can be modified to suit other study needs.) A modification to this subsampling regimen, which may speed this step, would be to establish a minimum number of exuviae for each sample (e.g., ≤100 individuals) and only subsample when this number is exceeded.

Materials

_ One small 3” sieve with 125 micron mesh size
_ Forceps and probe
_ Squirt bottle with 80% ethanol
_ Squirt bottle with tap water
_ Printed locality labels
_ Spoon
_ Syracuse dish or petri dish/watch glass
_ Stopwatch
_ One-dram (3.697 mL) snap top vials
_ Random number table or dice
_ One plain plastic sorting tray (photography tray without ridges)
_ One plastic sorting tray with a numbered 16-grid pattern on the bottom

Methods

(1) Remove a sample vial from the storage box. If there are ≤20 Chironomidae specimens in the vial it does not need to be subsampled. If there appear to be more than 20, continue to step 2.

(2) Place the sieve into the unmarked sorting tray. Take off vial cap and remove label; check label for organisms stuck to it. Strain the preservative from the contents of the sample by pouring though the sieve. Let ethanol drip from the sieve for long enough to remove most ethanol, pour EtOH back into vial, then place the sieve into the numbered sorting tray.

(3) Check vial, vial cap, and label for any attached specimens.

(4) Rinse all of the material from the sieve onto the center of the numbered sorting tray. Add tap water to the tray so that the bottom is covered with about 1 cm of water. Swirl until material is distributed evenly throughout the tray. Wait for material to stop moving.

(5) Randomly select five grid squares from a random number table or using dice. Count the number of Chironomidae in those grids, including all that are completely inside the squares and any that are touching the bottom and right edges (regardless of how much of the body is inside the square). If there are a total of >20 specimens summed across all five grids, then this group should be subsampled; proceed to subsampling in steps 6–10. If subsampling is not needed, return material to its vial.

(6) To subsample, swirl the sample for 10 seconds to redistribute it, then wait for material to stop moving. Beginning with the upper left square, remove organisms from all alternating squares from left to right, then from right to left for the next row down, which will form a checkerboard pattern of target squares (Figure F3). Place organisms into a new 1-dram (3.697 mL) vial. For each grid square, only take organisms that are either completely inside the square or are touching the bottom and right edges (regardless of how much of the body is inside the square). All organisms outside the squares or touching the top and left edges are not removed.

(7) After removing the subsample, review the remaining material and look for general morphological differences from organisms removed. Most or all should appear the same, but representatives of any that appear different (morphological traits, colors, patterns, or size) should be included in the subsample. [Note: this step is used for detection of biodiversity and may not be of interest for other study designs.]

(8) Return the remaining material to the original sample vial, filling 80% full with EtOH. Place the original locality label inside along with a new “subsample remnants” label.

(9) Place a locality label inside the new vial, along with a “subsample for identification” label. Fill vial 80% full with EtOH.

(10) Fill-out the subsampling section of the sample processing log.

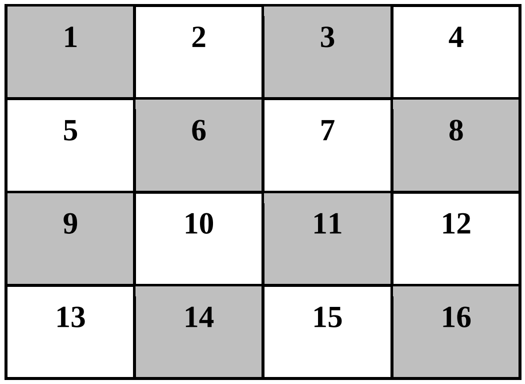

Figure F3. Grid pattern used in sampling and subsampling tray. Samples from every other (gray) square are removed for slide mounting and identification if subsampling is required.

STANDARD OPERATING PROCEDURE 5c – Preparing Slide-mounted Voucher Specimens of Chironomidae Pupal Exuviae

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

This SOP establishes procedures for slide mounting high-quality voucher specimens of Chironomidae pupal exuviae. Chironomidae exuviae are also best identified as slide-mounted specimens under high magnification. ***Slide mounting media such as euparal may cause allergic reactions or respiratory irritation***; we suggest placing the microscope in a fume hood and avoiding excessive contact of media with skin (Figure F4). See material safety data sheets for all mounting media used. Knowledge of chironomid exuviae traits will be important during slide mounting; see Wiederholm (1986) or Ferrington et al. (2008) prior to beginning this process. Mounting multiple specimens per slide can speed the process of identification and save storage space, along with reducing costs of slides, cover slips, euparal, and slide boxes. However, care should be taken to mount specimens in an even horizontal line, and trying to have the same genera on the same slide.

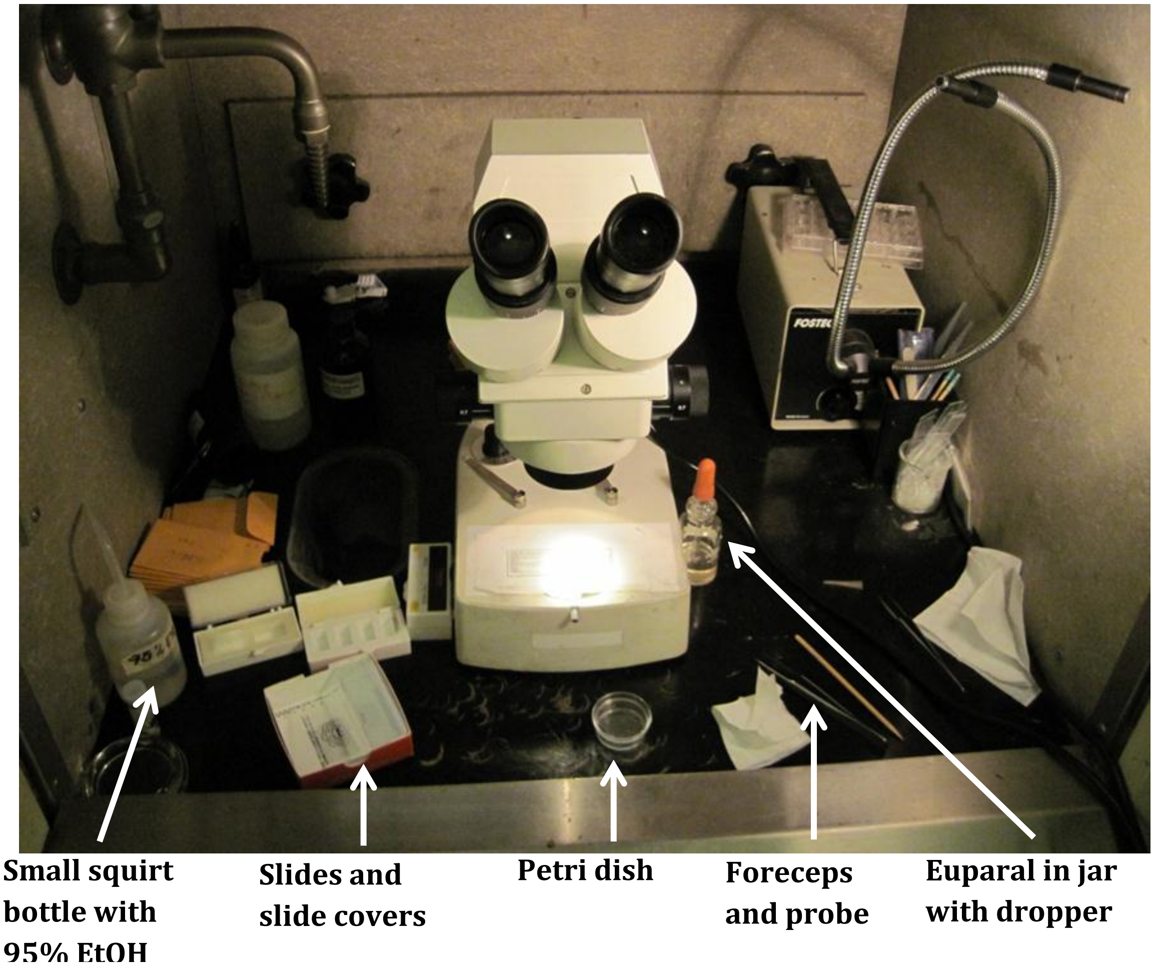

Figure F4. Slide-mounting dissecting scope station set-up inside a fume hood.

Materials

_ Dissecting microscope
_ Archival quality pen
_ 2 pair fine-tipped forceps
_ Needle-tipped probe
_ Small squirt bottle with 95–100% ethanol
_ Syracuse dish or petri dish/watch glass
_ Glass-cleaning towel or Kimwipes
_ Euparal in a glass jar with dropper
_ Slides
_ Cover slips (both round and square)
_ Pre-printed locality labels on sticker paper
_ Scissors

Methods

(1) A template card should be made for slide location, cover slip location, and an X for where exuviae should be mounted. Tape the template to the microscope base so that specimens are mounted in the same location on each slide. This both speeds the process and allows a standardization of exuviae location on the slide, so it can be found easily during identifications.

(2) Remove vial containing exuviae in 80% EtOH. Transfer all specimens from vial to Syracuse or petri dish. Add a thin layer of 95–100% ethanol to the dish to dehydrate exuviae. Use microscope to make sure all specimens are transferred (some are very translucent and small). To avoid rapid evaporation, keep a cap or lid on the dish with 95–100% EtOH when not sorting or pulling specimens from it. Set vial aside. Allow specimens to dehydrate in the ethanol for 5 minutes (cut labels during this time).

(3) Inspect specimens in the dish to see the variability and size of them. Exuviae that are not complete or nearly so (i.e., missing the cephalothorax or entire abdomen) may be returned to the storage vial (unless it appears to be clearly unique from prior specimens). Do not assume that a stray cephalothorax and abdomen belong together.

(4) Clean slide and cover as necessary with Kimwipes. Set a blank slide on the template. Use a dropper to place euparal on the slide in the marked position: the amount will vary depending on the size and number of specimens that will be mounted on the slide, but ensure enough is on the slide to coat the underside of the cover slip without spilling out from under the cover slip.

(5) Select specimen(s) to mount on the slide; try to mount the same taxa (genera are often distinctly different) on each slide. In the Syracuse dish, with forceps and probe, very carefully separate the cephalothorax from the abdomen. Make sure cephalothorax will spread apart when mounted by gently severing structures on the dorsal side. Try to keep the first abdominal segment attached to the abdomen, not to the cephalothorax (this can be important for identifications).

(6) Use forceps to place the specimen in the euparal with the dorsal side up for the abdomen and the ventral side up for the cephalothorax (which should also be spread flat). If mounting several specimens side-by-side, arrange these structures horizontally so that each cephalothorax is directly above the abdomen from the same specimen.

(7) With the second forceps, which should not have euparal on them, gently place a cover slip over the specimens by holding it at a 45-degree angle, then slowly lower the cover. Avoid air bubbles as much as possible. Use a square cover slip if multiple specimens are on the slide and a round cover slip if there is only one (this helps identifiers to look for one or multiple specimens).

(8) Place each completed slide to the side while working through steps 4–7 for the rest of the sample specimens.

(9) If cloudiness develops in the euparal, immediately stop slide mounting and replace 95% ethanol in the dish, taking care to not lose or damage specimens. It is possible that 100% ethanol will be necessary to avoid cloudiness, which can obscure traits during identifications.

(10) When all desired material from a sample has been mounted, return unmounted material to the storage vial.

(11) Affix a locality label to the left side of each slide (Figure F5). Number the slides in this sample sequentially, starting where the last sample left off. Keep slides stored horizontally (flat) in a slide box in numerical order.

(12) Fill out the log-sheet after each sample is completed (Table F1). When finished slide mounting for the day, store slides in slide boxes in the fume hood while euparal dries; all slide boxes should be stored on their sides so slides are face-up. Keep these in the fume hood for several days before working with them. Before euparal dries, which may take several weeks, the cover slips may move if slides are stored vertically. In general, long-term storage of slides should also be horizontal.

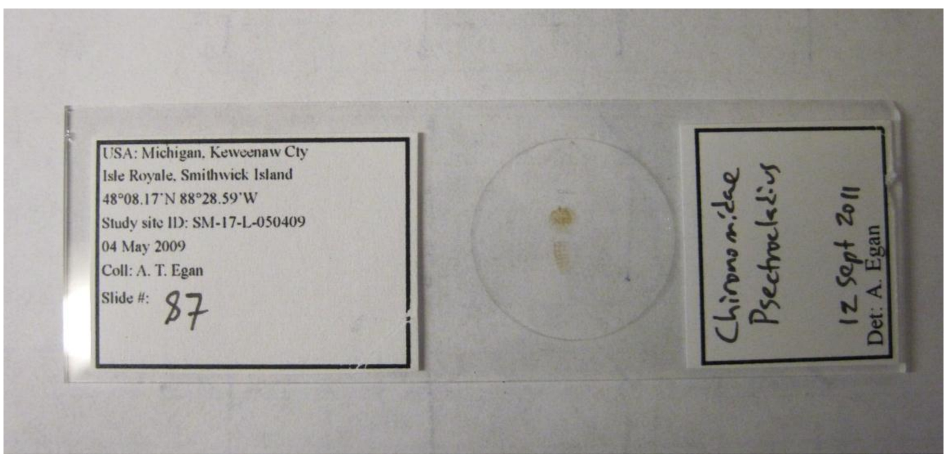

Figure F5. A labeled slide with one mounted exuviae under a round cover slip (Chironomidae: *Psectrocladius* sp.).

Literature Cited

Ferrington, L. C., Jr., W. P. Coffman, and M. B. Berg. 2008. Chironomidae. Pages xx-xx in R. W. Merritt, K. W. Cummins, and M. B. Bert, editors. An Introduction to the Aquatic Insects of North America, 4th ed. Kendall Hunt, Dubuque, Iowa.

Wiederholm, T. (editor) 1986. Chironomidae of the Holarctic region: Keys and diagnoses. Entomologica Scandinavica Supplement Number 28:1-482.

Table F1. Example of a slide-mounting log to document mounting effort.

Rock Pool Project 2009-2010 – Slide Mounting Log Page ___ of ___

Slide #s	**Sample ID**	**Date Mounted**	**Start Time**	**End Time**	**Elapsed Time**	**Slides per Sample**	**Comments**	**Mounted by**

STANDARD OPERATING PROCEDURE 5d – Labeling Macroinvertebrate Specimens

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

Specimens collected in rock pools will be stored in vials or slide-mounted for identification and long-term curation. Labeling is critically important for current and future analyses of samples. All specimen vials or slides will receive labels during this project. Labels for vials will be printed with a laser printer and additional notation to each label will be done in pencil to avoid dissolving ink if the label comes into contact with ethanol. Labels for slides will be printed on sticker paper, which can be cut out and affixed to slides, and information such as slide number will be written with an archival-quality pen.

Methods

(1) Locality labels for pinned and alcohol-preserved specimens will follow the standard of: general location (country, state, county), specific location (park, sample site), latitude/longitude, study site identification code for the project, date (numerical day, 3-4 letter month, full numerical year), and collector name, with text surrounded by a black border (Figure F6).

- Study site identification codes are structured as follows: Four-letter park code–two-letter site code–alphanumeric pool code–six-digit date (ddmmyy) (Figure F6).

(2) Specimen labels for pinned and alcohol-preserved specimens will follow the standard of: order (in capital letters [optional]), family, genus and species, date (numerical day, 3–4 letter month, full numerical year), and determiner name, with text surrounded by a black border.

(3) Slide-mount labels will follow the same standard as the locality labels, including a slide number at the bottom, for the label that will be affixed to the left side of the slide (see Figure F5). The right-hand slide label will include order name (in capital letters [optional]), family, genus and species (if multiple specimens are on a single slide they will be identified based on their mounted position), date and determiner name as with specimen labels. Both will have text surrounded by a black border.

(1) Locality label example	(2) Specimen label example
USA, Michigan, Keweenaw Cty Isle Royale, Blueberry Cove 48° 00.04 N, 088° 40.59 W Study site ID: BL-L1-040610 4 June 2010 Coll. A.T. Egan	**Chironomidae Xenochironomus xenolabis (Kieffer) 9 Dec 2010 Det. A.T. Egan**

(3) Slide label examples

USA, Michigan, Keweenaw Cty Isle Royale, Blueberry Cove 48° 00.04 N, 088° 40.59 W Study site ID: BL-L1-040610 4 June 2010 Coll. A.T. Egan Slide #	**Chironomidae Xenochironomus xenolabis (Kieffer), left Xenochironomus xenolabis (Kieffer), center Xenochironomus xenolabis (Kieffer), right 18 Sept 2011 Det. A.T. Egan**

Figure F6. Examples of labels for locality (1), specimen identification (2), and slides (3).

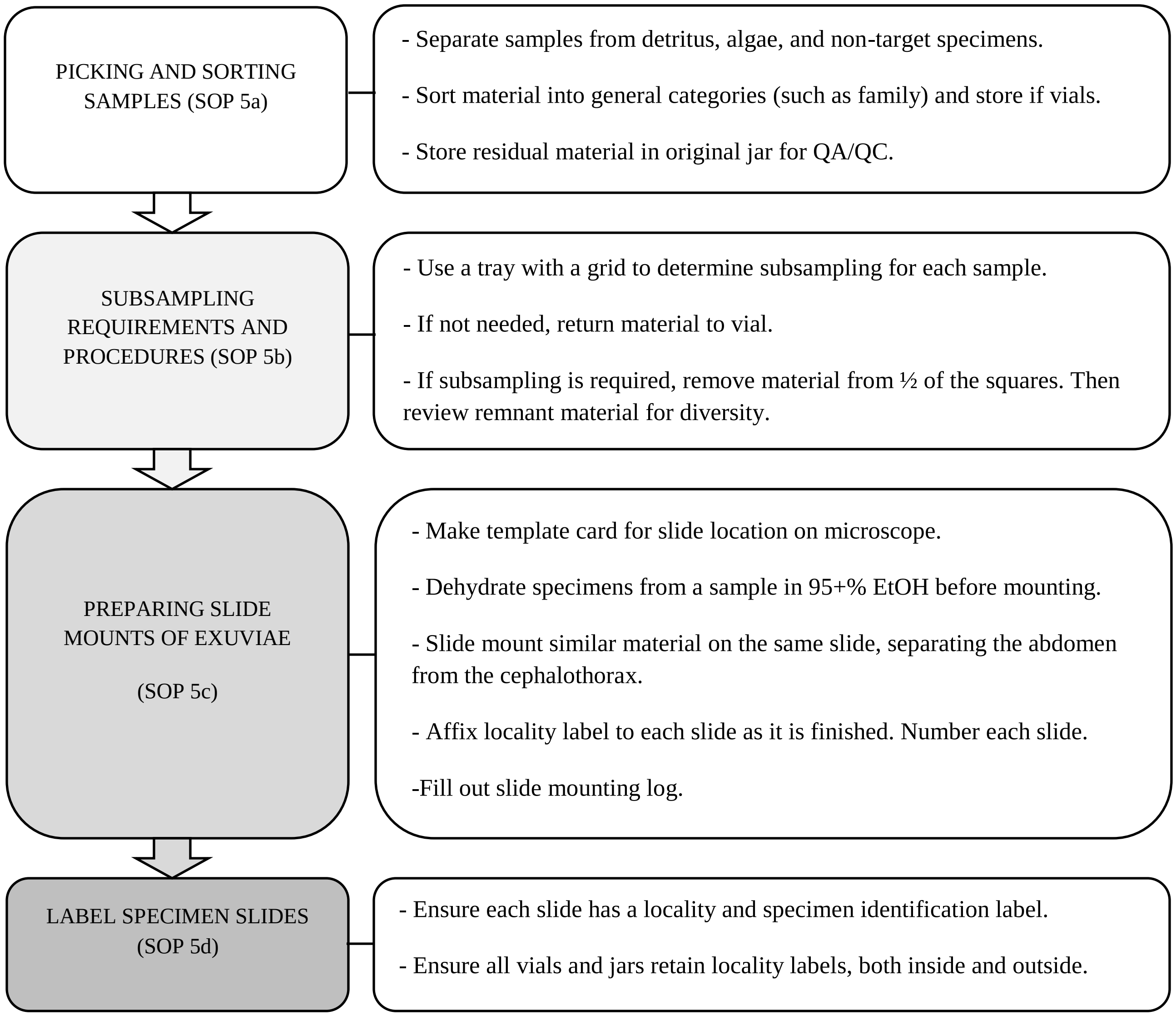

Figure F7. Visual model for rock pool Chironomidae exuviae lab work.

STANDARD OPERATING PROCEDURE 6 – Diatom and Water Quality Sampling and Processing

Version 1.00
Prepared by Mark Edlund

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

Biological and water quality sampling of splash pools takes place in remote locations and thus requires modifications to typical sampling protocols. This SOP outlines field sampling and preservation of splash pool diatom communities, and field sampling and processing methods for water quality. Diatom sampling targets a similar community among pools (the epipelon, or community living within the detrital material sedimented in the pool). Water quality measures include Total Phosphorous/Total Nitrogen (TP/TN), chlorophyll, Dissolved Organic Carbon/Dissolved Inorganic Carbon (DOC/DIC), anions, cations, metals, soluble reactive phosphorous (SRP), NH_4, NO_3/NO_2. On-site measures include pH, temperature, conductivity, and dissolved oxygen (DO) as measured with a sampling probe (YSI or Hydrolab).

Field Gear Checklist

_ Diatom sampling kits (six or seven 10-mL prelabelled plastic Evergreen bottles per shoreline site, 6–7 disposable plastic pipettes in ziploc)
_ Large cooler with ice if possible
_ Fifteen-to-eighteen 1-L amber bottles for bulk water collection (need 3 1-L bottles per pool)
_ YSI or Hydrolab multiprobe, pre-calibrated
_ field notebook and pens
_ GPS, compass
_ measuring tape (10–25 m)
_ personal gear (hipboots, raingear, hat, first aid, sunscreen, weather-appropriate clothing)
_ field sheets
_ watch/timer
_ camera

Processing Gear (can be done in field if necessary)

Diatoms: 100 mL 37–40% formaldehyde solution and pipette

Water Quality:

1. Hand vacuum pump

2. Plastic 1 L sidearm Erlenmeyer flask for filtering

3. Large glass Millipore filtering apparatus (2 pc glass with blue clamp)

4. Filter forceps

5. Two types of filters, 47 mm diameter, GF/C and PCTE

6. Graduated cylinders, 50 mL, 1 L, 25 mL

7. Water quality sampling kit (5 bottles, 1 foil envelope, all labelled)—60 mL clear HDPE, 15 mL clear LDPE, 15 mL amber PP, 60 mL amber HDPE, 60 mL amber glass with Polyseal cap

8. Processing solutions: 100 mL saturated $MgCO_3$ solution, 30 mL concentrated HNO_3, 30 mL concentrated H_2SO_4, 10 mL $CuSO_4$ solution (40 mg $CuSO_4$ + 10 mL dH_2O)

9. Jug of distilled water and small squirt bottle

10. 60 mL plastic syringe with Luhr-lok filter apparatus

11. Electrical tape, disposable pipettes

Sampling Sequence at each pool (6 "pools" total—2 in lichen zone, 2 in splash zone, one composite ephemeral pool in lichen zone, one composite ephemeral in splash zone). Steps 1–5 at each regular pool, Step 7 at the composite ephemeral pools.

1. Water quality and site description (GPS; pool depth, length, width; distance to lake; distance to treeline; site description; Multiprobe for pH, temperature, DO, and conductivity; collect 3 L whole water from each of two lichen pools, two splash pools, and from Lake Superior.)

2. Diatom (collect 9 mL of material from pool bottom using pipette)

3. Pupal exuviae (chironomids) using pan and sieve technique (see SOP 4)

4. Zooplankton (35 µm nets)

5. Large insects/adults/larvae—sweep nets, aspirator, then sweep nets at subsurface (observe for 5 min, collect representatives and preserve with skins)

6. Amphibians (identify eggs, tadpoles, and adults—*no collections*)

7. Ephemeral pools (one set in lichen zone, one set in splash zone, 10 minute sampling at each zone, 15–30 secs per pool)

    a. diatoms, composite sample (approx. 1 mL from each pool until vial is full)

    b. pupal skins (tray and sieve)

c. sweep, aspirate

d. composite zooplankton

e. no water quality measurements

Water Quality Field Sampling

1. Record GPS location; measure depth, length, and breadth of pool, and distance to lake and to treeline. Using calibrated Multiprobe or YSI meter, take measurements of temperature, DO, pH, and conductivity at mid-pool. Record on field sheets/book.

2. To sample water, open three 1-L amber bottles and fill 1/3-full of water from pool, cap, rinse, discard, and repeat. For water collection, fill all three 1-L bottles from mid-pool or as far as you can reach from edge without disturbing pool.

3. Cap and label (S1, S2, L1, L2, LkSup), or record colored tape sequence for each pool; place all water samples in cooler. Should have fifteen 1-L samples in cooler from each site.

4. Collect a second (duplicate) sample at 10% of the pools (two duplicates during each water quality sampling event at ISRO; one duplicate during each water quality sampling event at APIS and PIRO)

Diatom Sampling and Preservation

General Description: At each splash pool, 9 mL of material will be collected with a disposable pipette from the bottom gunk of the pool. For the ephemeral pools, a composite sample is made using approximately 1 mL of sedimented material from each ephemeral pool until 9 mL are in vial. All diatom material will be placed in individual 10 mL plastic screw top vials that have been prelabelled. Small kits of 6–7 vials and pipettes are premade.

1. LABEL THE SAMPLE VIALS WITH YOUR POOL ID. Using disposable pipette as many times as necessary in a pool, collect 9 mL of the biological goo that accumulates in the bottom of each index pool. If there is not much goo in the bottom of the pool, use the pipette to scrape the rocky surfaces as you release the bulb to try and scrape some material from rocks. You should be able to see some material in the vial when you are done. Make sure to label vial and store in cooler.

2. From each set of ephemeral pools, LABEL THE SAMPLE VIAL, and collect samples in 1 mL increments using a pipette until a total of 9 mL of sample is in vial. Label vial and store in cooler.

3. Thus, each shoreline site should end up with 6 diatom samples (2 from the two lichen pools, 2 from the two splash zone pools, 2 from each of the ephemeral pool types—lichen and splash.

4. DUPLICATE samples. In about every third diatom sampling kit bag is an extra sample vial. Please use this to collect a duplicate sample from one of the index (lichen or splash zone) pools. Please label as duplicate and store in cooler.

5. Diatom Preservation: BE CAREFUL…Put on gloves and eye protection and do preserving of diatoms outside. Using the bottle of 37% formaldehyde solution and a disposable pipette, add 10 drops (about 0.5 mL) of formaldehyde solution to each sample vial. Cap vial, agitate for 3–5 secs to mix, and store in ziploc baggie. Preserved diatoms can be stored at room temperature, but store in the dark if possible. FIRST AID—for formalin on skin, rinse off and wash site; for formalin in eyes, rinse, rinse, and rinse some more with clean water. Then go to the doctor if any irritation or vision problems develop.

Water Quality Processing

1. Assemble parts for large glass filter apparatus (stored in the box) using a GF/C filter (rough side up, not patterned side) and attach to 1 L Erlenmeyer flask. Connect the hand vacuum pump to flask.

2. Locate 60 mL syringe and the luhr-lok filter holder (the clear plastic one) and load with a PCTE filter, shiny side up, and make sure both diffuser discs have the side with circular ribs against the filter.

3. You need three disposable or glass pipettes with bulbs and the four water quality chemicals: (1) the 30 mL vial of concentrated H_2SO_4, (2) the 30 mL vial of concentrated HNO_3, (3) bottle of saturated magnesium carbonate, and (4) for each park's water quality sampling event also make up 10 mL of copper sulfate solution by adding 10 mL of distilled water to the small glass vial that holds 40 mg of $CuSO_4$; agitate to dissolve (this solution is used drop-wise and will be used for several days of sampling—DO NOT THROW AWAY after each day you process water quality samples). SAFETY: THE ACIDS ARE NASTY. USE GLOVES AND EYE PROTECTION, RINSE WELL IF YOU GET ON SKIN, NEUTRALIZE ANY SPILLS WITH BAKING SODA.

4. Fill squirt bottle with distilled water.

5. You will generate six water quality end-products consisting of five bottles and one filter packet by the end of this procedure. The order is: TP/TN, chlorophyll-*a*, cations/metals, anions, SRP/NO_3/NH4, DOC/DIC. Locate and unpack one of the water sampling kits that are prepacked in ziplocs—this kit will be used for one pool. There are five bottles and a foil filter packet. LABEL ALL BOTTLES AND FILTER PACKET WITH SAMPLE SITE NUMBER AND DATE (e.g., ISRO-PI-L2-13MY2010)

   For each sampled pool, you need gloves and eye protection and the following equipment:

   (i) TP/TN—60 mL clear HDPE, 25 mL graduated cylinder

   (ii) Chlorophyll—foil packet, saturated $MgCO_3$ solution and plastic pipette, big glass filter apparatus with GF/C filter

(iii) Cations/metals—15 mL clear LDPE, syringe and PCTE filter, concentrated HNO_3

(iv) Anions—15 mL amber polypropalene, syringe and PCTE filter

(v) SRP/NO_3/NH_4—60 mL amber HDPE, syringe and PCTE filter, concentrated H_2SO_4

(vi) DIC/DOC—60 mL amber glass with polyseal, $CuSO_4$ solution, electrical tape. Stretch the tape tight as you wind it around the cover to seal out any air!!

6. TP/TN: Using the first 1-L of water you collected, mix the bottle gently and pour approx. 10 mL into a 60 mL clear HDPE sample vial, rinse, and discard water. Using the 25 mL graduated cylinder, rinse with a little sample water, then fill to 20 mL and pour into the 60 mL bottle, and cap.

7. Chlorophyll: From your other two liters of water, mix and pour 1 L of water into the 1L graduated cylinder. Rinse the glass filter apparatus with a little distilled water from your squirt bottle, then start pouring your 1L of water from the graduated cylinder into the filter apparatus. Start pulling a vacuum to assist the filtering, trying to not go above 30 on the gauge. Slowly add water to the apparatus. As you near completion of filtering the first 1L, is the filter pretty green or brown yet? If so, add 0.5 mL of saturated $MgCO_4$ (shake the bottle of $MgCO_4$ to suspend) to the last bowlful of water you filter. If the filter is not so green yet, release vacuum, pour off the waste in the big Erlenmeyer flask, and then measure and filter another 500 mL to 1 L of water. As the last bowlful of water is being filtered, add 0.5 mL of saturated $MgCO_4$ using a plastic pipette and finish the filtering. Disassemble the apparatus and fold the filter with the forceps into quarters. Do not exactly align edges of filter when folding; this makes it difficult to unfold for analysis. Place filter in labelled foil envelope, finish folding the edges of the envelope, and LABEL WITH THE VOLUME OF SAMPLE FILTERED.

8. Cations/metals: Assemble syringe and filter appartus with the PCTE filters. Use your first 1 L of water you took. YOU WILL SIMPLY USE THE SAME FILTER TO FILL THE NEXT FOUR BOTTLES unless it gets too hard to push water throught—then just put a new one in. DO NOT PULL THE PLUNGER BACKWARD OR YOU WILL TEAR YOUR FILTERS—UNSCREW SYRINGE FROM FILTER HOLDER FIRST. For cations/metals, add 5 mL to the 15 mL clear LDPE, rinse the bottle and discard, then fill to top with filtered sample water. Add 2 drops of concentrated HNO_3 (nitric acid), and cap with no head space. For acid spills neutralize with baking soda provided.

9. Anions: Pour out water in the 15 mL polypropalene bottle (this is just distilled water for storage purposes), then, using syringe and filter apparatus with the PCTE filters, add 5 mL to 15 mL amber polypropalene bottle, rinse and discard, then fill to top and cap with no head space. NO PRESERVATIVE.

10. SRP/NO_3/NH_4: Use syringe and filter appartus with the PCTE filters, add 5–10 mL to the 60 mL amber HDPE bottle, rinse and discard, then fill to top with 60 mL of filtered sample water,

add 6 drops of H_2SO_4 (sulfuric acid), and cap with no head space. For acid spills neutralize with baking soda provided.

11. DIC/DOC: Syringe and filter appartus with the PCTE filters, add 5–10 mL to the 60 mL amber glass bottle, rinse and discard, then fill to top with 60 mL of filtered water, add 5 drops of the $CuSO_4$ (copper sulfate) solution, cap without any head space, and wrap top with electrical tape—stretch that tape as you wrap to completely seal the bottle from outside air!

12. Sample storage

    TP/TN—frozen

    Chlorophyll, foil packet—frozen (make sure volume filtered is on label)

    Cations/metals—refrigerate

    Anions—refrigerate

    SRP/NO_3/NH_4—refrigerate

    DIC/DOC—refrigerate after wrapping top with electrical tape

13. Fill out chain of custody form with information for sample transfer.

14. Samples do NOT need to be mailed or shipped immediately. It is probably best to hold May and July samples from ISRO until return trip in July. May and September trips to APIS and PIRO should be returned ASAP to St. Croix Watershed Research Station.

15. BLANKS AND DUPLICATES. About 10% duplicate samples and blanks are needed for water quality. For each of the water quality sampling weeks at ISRO, please take two duplicate samples from any of the index pools and process as you would a normal index pool sample. Make sure to LABEL as DUPLICATE. We also need to do two blank samples for each sampling week at ISRO. A blank is done by simply processing distilled water as you do a normal sample (it will be very easy to filter!!). Make sure to LABEL as BLANK along with park and date.

16. Make arrangements before the field season with qualified analytical laboratories capable of analyzing the following samples (example analytical setup):

    (a) Chlorophyll: APHA Standard Method 10200 H. (Chlorophyll) and EPA Methods 445.0 (Chlorophyll and Pheophytin in Algae by Fluorescence) and 446.0 (Chlorophylls and Pheopigments in Phytoplankton by Spectrophotometry).

    (b) TP: Standard Method 4500-P H. (Manual Digestion and Flow Injection Analysis for Total Phosphorus) and Lachat QuikChem Method 10-115-01-1F.

(c) TN: Standard Method 4500-N C. (Persulfate Method), 4500-NO_3 I. (Cadmium Reduction Flow Injection Method), and Lachat QuikChem Method 10-107-04-1-A.

(d) SRP: Standard Method 4500-P G. (Flow Injection Analysis for Orthophosphate), and Lachat QuikChem Methods 10-115-01-1-A (high range) or 10-115-01-1-A (low range).

(e) NH_4: Standard Method 4500-NH_3 F. (Phenate Method), 4500-NH_3 I. (Flow Injection Analysis), and Lachat QuikChem Method 10-107-06-1-B.

(f) NO_3-NO_2: Standard Method 4500-NO_3 I. (Cadmium Reduction Flow Injection Method), and Lachat QuikChem Method 10-107-04-1-A.

(g) DIC/DOC: Standard Method 5310 C.—Persulfate-Ultraviolet or Heated-Persulfate Oxidation Method, Tekmar-Dohrmann Phoenix 8000 carbon analyzer.

(h) Anions: Ion chromatography system, Dionex ICS 2000 - AS19 (4 mm) column - ASRS 300 (4 mm) suppressor - NaOH eluent generator.

(i) Cations: Inductively Coupled Plasma - Optical Emission Spectrometry, Thermo Scientific iCAP 6500 dual view ICP-OES.

(j) Trace metals: Inductively Coupled Plasma – Mass spectrometry, Thermo Scientific XSERIES 2 ICP-MS w/ ESI PC3 Peltier cooled spray chamber, SC-FAST injection loop, and SC-4 autosampler.

STANDARD OPERATING PROCEDURE 7 – Diatom Sample and Slide Preparation

Version 1.00
Prepared by Mark Edlund

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

Analysis of diatoms requires removal of organic material from field samples using oxidative methods (cleaning) and preparation of permanent microscope mounts of cleaned diatom material. This SOP is modified from Ramstack et al. (2008a, 2008b).

Equipment needed for cleaning rock pool diatom samples

_ 50 mL NUNC polypropylene fliptop conical centrifuge tubes (NUNC Catalog #362697; available through Fisher Scientific, www.fishersci.com/)
_ 10% (by volume) reagent grade hydrochloric acid (HCl)
_ 30% (by volume) reagent grade hydrogen peroxide (H_2O_2)
_ De-ionized (DI) water
_ Spatula
_ Permanent marker to label centrifuge tubes
_ Water bath set at 85 °C
_ Centrifuge capable of holding 50 mL NUNC centrifuge tubes and spinning at 3500 rpm
_ Positive-draw fume hood

Equipment needed for preparing diatom slides

_ Disposable plastic pipettes (approx. 7 mL)
_ Kimwipe® tissues
_ De-ionized (DI) water
_ Glass coverslips (22 mm × 22 mm) – No. 1 thickness, two per sample
_ Aluminum drying plate (squares are etched onto the surface, each square is etched with an identifying number)
_ Hotplate with temperature control
_ Positive-draw fume hood
_ Glass microscope slides (1 × 3 inches; 2.5 × 7.5 cm), two per sample
_ Permanent marker

_ Zrax mounting medium, a toluene-based mounting medium (information on Zrax can be found at http://www.sas.upenn.edu/~dailey/zrax.pdf)
_ Fine-tipped forceps
_ Rounded toothpicks, disposable wooden stirring sticks, or pencil with eraser
_ Single-edged razor blade
_ Diamond or carbide-tipped marking pencil
_ Paper labels for slides

Personal protective equipment (PPE) needed for cleaning and mounting rock pool diatom samples

_ Acid-impervious gloves
_ Safety glasses and face shield
_ Laboratory coat or apron, acid resistant

Precautions

Hydrogen peroxide (H_2O_2) is used to digest organic materials; it is a corrosive material and should be used under a fume hood while wearing a face shield, safety glasses, gloves, and a lab coat. H_2O_2 is corrosive and a strong oxidizer; store in a cool, dry, well-ventilated area away from incompatible substances, and keep away from heat, sparks, and flame. Refer to the MSDS in your laboratory for complete storage and handling instructions.

Diatom Sample Cleaning

1. Sediment samples should be stored in a dark, 4 °C, relatively air-tight environment until processed. Process samples within two weeks of collection.

2. Mix each rock pool diatom sample well with a spatula or gentle mixing to ensure an accurate representation throughout the whole sample.

3. Place approximately 0.5-to-1.0 mL of homogenized sample in a labeled, 50 mL polypropylene centrifuge tube with a snap cap (tubes should be labeled with the date, unique sample ID, and your initials). The samples will not be analyzed quantitatively for diatom concentration, so it is not necessary to record the volume of sample used.

4. Add 10 mL 30% peroxide (H_2O_2), and leave sample in fume hood overnight with top cap open to intitiate oxidation of organics. In the morning, place sample and tube in an 85 °C water bath (with the centrifuge tube flip-top open) for 3 hours. Monitor the samples closely for the first 20 minutes to make sure the reaction is not too violent. If the samples come close to boiling over, remove them from the water bath and let the reaction proceed at room temperature under a positive-draw fume hood (the samples can be put back in the water bath after the reaction has slowed down, but watch them closely to make sure they do not boil over). Dense samples with lots of silts may need periodic homogenization with a glass stirring rod to ensure that buried organic material is adequately exposed to the peroxide. Samples are fully digested when the

solution is clear and the reaction has stopped; if samples are not fully digested after 3 hours, they can be left in the water bath for additional time, or left at room temperature under a positive-draw fume hood until the digestion is complete.

5. When cool, fill each centrifuge tube to even levels (to approximately the 50 mL mark on the tube) with de-ionized (DI) water.

6. Centrifuge each sample for 6 minutes at 3500 rpm. Repeat five times for each sample, decanting supernatant each time and re-filling with DI water. When decanting, pour off as much water as possible without losing any of the sediment.

7. After the last centrifuge run, decant but do not re-fill the centrifuge tubes.

8. The samples can now be used to prepare diatom slides.

Diatom Slide Preparation

1. Use a Kimwipe® to wipe each coverslip as you remove it from the box. Place each coverslip on a marked space of the aluminum drying plate. Be sure the aluminum drying plate is clean and dry to avoid cross-contamination. NOTE: All slides will be made in duplicate, so prepare two coverslips for each sample.

2. Under the fume hood, add one small drop of 10% HCl to each centrifuge tube to create a better distribution of diatoms.

3. Add de-ionized (DI) water to each centrifuge tube until the liquid is a slightly translucent grey color. This means that more water will be added for samples with a high diatom abundance and less water for samples that have a lower density.

4. If the solution is still overly saturated with diatoms (either the solution is still fairly dark grey or you have previously tried to make a slide from a similar sample that was too dense to count), put a large drop of distilled water on the coverslip before transferring the diatom suspension.

5. Agitate the sample to a uniform dispersion and use a plastic pipette to add the diatom solution to each coverslip. Fill each slip corner-to-corner and to the maximum surface tension of the coverslip. If the coverslip overflows, discard it and repeat the procedure with a freshly cleaned coverslip. Use a new pipette for each sample.

6. Once the aluminum drying plate is loaded with coverslip preparations, allow coverslips to dry undisturbed overnight at room temperature. The drying plate should be on a surface where vibrations from surrounding equipment are minimal and free of drafts so that the distribution of diatoms on the coverslip is even. It is highly recommended to draw a map of the sample locations on the drying plate to ensure that samples do not get mixed up.

7. Once coverslips have dried, a diatom density check can be performed on unmounted coverslips. Coverslips can be placed, diatom side up, on a slide and observed at 400X under a microscope. If

diatoms are too dense the coverslip can be discarded and remade—when remaking, dilute the sample either by adding more DI water to the centrifuge tube, or adding more DI water to the coverslip before transferring the diatom suspension. If the diatoms are not dense enough, more of the suspension can be added to the same coverslip and dried again. A final density check will be made after the coverslips are mounted, but performing this preliminary check before making permanent slides will save time and materials.

8. After the coverslips have dried, place the aluminum drying plate full of coverslips on a hotplate to drive off hydroscopic water (for 1 hour at 250 °C). The hotplate should be under a fume hood in preparation for the following steps.

9. Label microscope slides and clean with a Kimwipe®. Slides can be labeled with a Sharpie marker at this stage, a paper label will be added at the end of the process.

10. Turn on the fan of the fume hood. Once coverslips have been dried, add a small drop of mounting medium to a slide by placing the slide over the top of the bottle and inverting, or by using a disposable pipette (the volume of mounting medium on the slide should be equivalent to approximately 2 to 4 drops of water). Place the drop of mounting medium in the center of the slide to leave room for a label on the side of the slide. Remove the appropriate coverslip from the aluminum plate with forceps, being careful to handle the coverslip only at the extreme corners. Invert the coverslip and place it gently on the portion of the slide that is covered with mounting medium.

11. Place the slide (keep the coverslip-side up) on the hotplate (which is still set at 250 °C within the fume hood, with the fan turned on). Bubbles will soon result from the evaporation of the toluene; keep the slide on the hotplate until the bubbles significantly diminish.

12. Remove the slide from the hotplate. Using a rounded toothpick, wooden stirring stick, or eraser end of a pencil, gently position the coverslip and press down on it to form a thin layer of mounting medium beneath the entire coverslip. Press down firmly, but not so hard as to damage the coverslip. The mounting medium will harden quickly; if it is necessary to reposition the coverslip after the medium hardens, the slide will have to be put back on the hotplate for a few seconds to soften the mounting medium.

13. If there are no bubbles under the coverslip and the mounting medium sufficiently covers the area of the coverslip (i.e., no edges of the coverslip are free of mounting medium), then set the slide aside to cool. If you are unable to remove all of the bubbles, put the slide back on the hotplate for a few seconds and repeat step 12. If there is not enough mounting medium on the slide, put the slide back on the hotplate and add a small amount of mounting medium to the edge of the coverslip with a disposable pipette (it will be pulled underneath as it heats) and repeat steps 11 through 12.

14. Once the slide has cooled, scrape the excess mounting medium from the edges of the coverslip with a razor blade. Always start in the center of the coverslip and work outward to avoid popping the coverslip loose from the slide; be sure that the blade is aimed away from your fingers.

15. Look at the completed slides on the microscope under low to medium magnification (dry 100X to 400X) to confirm that there is an even distribution of diatoms and that most the diatom frustules do not overlap, are not too dense, or too dilute. Remake any slides that have problems with the diatom dispersion or density that would interfere with the quality and accuracy of the analysis.

16. Label all completed slides with a printed paper label that contains the name of the state, the county, the name of the park, the GPS coordinates of the sample location, the rock pool ID, the collector's name, and the date (designate the duplicates of each sample as 'a' and 'b'). Use a dating convention in which the day and month are readily distinguishable (e.g., 21 Sept 2008 or 21 IX 2008) to avoid confusion between European and North American abbreviation styles. A diamond etching pen can also be used to label slides with the sample ID and date in case the paper labels ever fall off.

Literature Cited

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STANDARD OPERATING PROCEDURE 8 – Mapping Coastal Pools

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

Mapping may be designed to enumerate many details of rock pools, including locations of all available habitat, or only basic data of interest. This SOP suggests basic ecological data to be collected, but it can be modified to suit management needs. Mapping acts as a snapshot of conditions and should be repeated occasionally to determine trends or changes to habitat or communities. Timing should coincide with activity of organisms that will be mapped.

For safety, it is advised that a two-person team is involved in mapping, particularly for remote locations or overnight trips. Mapping should not occur if conditions are dangerous (such as wet bedrock or large waves), or if recent rain or wave activity has filled depressions that are not ecologically functional pools. Steep cliffs should be avoided, and safe working conditions must always come before data collection.

Equipment For Mapping Field Work

_ Two-way radio with extra battery, or cell phone
_ Digital camera with extra battery
_ Juno GPS with 2+ extra batteries and extra stylus for touch-screen
_ Laser rangefinder
_ Clinometer (unless rangefinder has one)
_ Measuring tape or meter stick
_ Field microscope or jeweler's lense
_ Quad maps or printed GIS maps of area
_ Keys for identification of invertebrates, amphibians, or other taxa of interest
_ Rite-in-the-Rain® project notebook
_ One dram (3.697 mL) vials with 80% EtOH for preserving unique specimens (3–4 per day of mapping)
_ 2+ 60-mL jars (for viewing/identifying specimens for capture and release)
_ 4+ foreceps
_ 4+ aquarium dip nets
_ Kneepads

_ Long-sleeved shirt and windbreaker/raingear
_ 360° sun hat
_ Bug headnet/suit
_ Sunglasses (polarized)
_ Camp stove, fuel, and large pot (for overnight trips)
_ Appropriate camping gear, if necessary

Prior to Daily Fieldwork

Each morning that mapping is planned, personnel will determine safe weather conditions based on National Weather Service marine forecasts obtained online or via the morning report on park radio. Do not land a boat on exposed shorelines and leave it alone while mapping unless conditions are calm and forecasted to remain calm. For extended mapping trips, an entire trip plan must be submitted and two radio contacts be identified for a daily check-in. A trip plan must be verbalized to a supervisor, ranger working in the area, or dispatch service, with an expected time of return and predefined response in case the check-in or return time is missed.

Methods for Habitat Mapping

1. The Trimble unit should remain on with Terrasync software open during shoreline hikes, unless long periods of hiking or canoeing are needed between points. Datum should be automatically set to NAD83. In the "Data" menu, choose the "Rock pool mapping DD [data dictionary]" and select the file name for the general location of mapping (e.g. Todd Harbor west, Edwards Island). Input a new file name if necessary, or open an existing file ("New" menu) to add more data.

2. Press "Create" for a new point (each pool), input antenna height as 0.000 m, highlight "ISRO Rock Pool" as the feature, then press "Create" button. A list of drop-down menus will appear and logging will automatically be paused. Stand or kneel next to the pool and press "Log"; a beeping sound begins as waypoints are collected, which is indicated in the top right corner. From 10 to 30+ waypoints should be collected, which the unit will average into a final point for the pool. While waiting, work through the drop-down menus to describe the pool.

3. Drop-down lists will be used to identify the following for each pool:

    Ecological Zone – Splash, Lichen, or Forest transition (NOTE: Some pools have lichen on the upslope side and no lichen on the down-slope side; these pools are included in the splash zone. See Definitions section at the beginning of this report.)

    Pool Permanence – Permanent pool or Ephemeral pool

    Recharge Source – Rain, Splash, Seep, or a combination of these

    Number of meters for maximum depth**, length, and width of the pool (**Depth measurement should be done using a clean local stick. Depth sticks should not be used on different islands; a new stick should be used for each ½-mile of shoreline to avoid moving taxa long distances. Mark each stick with a 2 mm and 10 mm mark to quickly assess depth.)

Distance to lake – using laser rangefinder or measuring stick, in meters

Angle to lake – degree of slope to lake edge

Topography – level, sloping, or cliff

Amphibian taxa observed – not detected (default), adults, larvae, eggs, or a combination

Amphibian taxa abundance – rare, occasional, common, abundant

Notes or other observations

Date of visit (automatic)

Time of visit (automatic)

Observer initials

4. Identifications will be done with the assistance of an amphibian key that includes eggs and tadpoles, including photos and descriptions. If species-level identification is not possible, the closest group will be identified (e.g., salamander, *Rana* sp.). This may require use of binoculars or an aquarium net and jar to capture, view, and release the amphibians. Do notdirectly handle amphibians, and do not place hands in pools if sunscreen, lotions, or insect repellant is on hands. If amphibians are captured with aquarium nets for identification they must be handled gently and returned to the original pool. Take photographs of confusing individuals (place a pool identifier in each picture) and send the photo(s) to a taxonomic expert for later identification. Change nets whenever a new depth stick is employed. Sterilize nets, forceps, and jars via boiling for several minutes as needed.

5. Each observer or team of observers must ensure that pools are not counted twice or missed. A standard pattern of walking shorelines should be used by each observer or agreed upon by each team. A distinct landmark (e.g. a waterfall, rocky point, or beach) should be used as a daily stopping point (also for longer breaks). In the project notebook at the end of the day, record the date, observer name(s), area worked, weather encountered, important or interesting notes from the day, and coordinates of beginning and end points of daily mapping.

6. If a section of shoreline has a cliff or dangerous conditions, as determined by each observer, that area can be skipped. Avoid the area by paddling or boating around it, or by hiking inland. Lake Superior is frigid year-round, and no risks should be taken in order to map an area. Note the area skipped in the daily log and estimate if any pools were likely at the location.

Recommended Methods for Biota Mapping

1. Biota sites will be chosen in a stratified random manner prior to field work.

2. Biota mapping should follow a 50- or 100-meter transect parallel to the shoreline, depending on density of pools. Determine beginning and ending points along the shoreline, then estimate a perpendicular line to the vegetation/forest line in order to give the transect boundaries. For pools that are less than halfway inside the transect edges, map only those that fall on the eastern/southern perpendicular line. Sample those pools that are more than half-way inside the area.

3. When approaching a pool, some taxa may be disturbed and leave the area (midges might fly to other pools, frogs or odonate larvae may hide under rocks), so watch for these species. Upon reaching a pool, the observer should kneel or lay on the rocks to closely observe the pool surface and subsurface for active taxa, dead insects, or exuviae. This step may be very short for small pools, but may take 5–10 minutes for larger, complex pools. The observer should get a sense of what taxa are present and general abundance of each.

4. During observation of a pool, check margins where semi-aquatic species may be, and use the depth stick to disturb areas to see if new taxa flush from hiding. Some species (e.g. beetles) will need to access the surface to collect air, so watch for this behavior. Use sweep net to collect aerial adults as necessary. Identify aquatic macroinvertebrates to the most refined level possible, using keys and taking photos as necessary. Taxa such as coleopterans, odonates, and corixids may be captured, viewed in a 60 mL jar, and returned to the pool. Refer to step 4 in "Methods for Habitat Mapping" above for details on capturing specimens for identification.

5. Follow steps 1–3 of "Methods for Habitat Mapping," using the "Rock Pool Biota DD," after adding more fields as needed to the data dictionary.

Post-Processing of Field Data

Transferring and correcting data

1. One of the priorities after a day or more of mapping is to transfer the data from the field PC to ArcView shape files. Base station correction data are sometimes not available for over 24 hours; if so, wait for a day or two before completing this process.

2. Plug Trimble into computer via USB cable. Ensure ActiveSync synchronizes the connection. Open GPS Pathfinder software.

3. Click Data Transfer, Receive, Add, and DataFile. Choose the files that need to be imported and click Open, then Transfer All. Files should then be in a Trimble Field Data file in the IsleRoyaleRockPoolMapping folder.

4. Unplug the field PC from the desktop computer and charge batteries as needed.

5. In GPS Pathfinder Office, click the Differential Correction tool (under Utilities). This will open a wizard process. The recently transferred file(s) should automatically appear on the list. Click the Add button if files do not appear. Click Next. Continue clicking Next for the Processing Type,

Correct Settings, Base Data, and Output Folder. Click Start to begin differential correction. **NOTE**: For the Base Data page, the default base provider for Isle Royale should be "CORS, Upper Keweenaw 5 (KEW5), MICHIGAN" (or KEW6; or Ontario Ministry of Natural Resources, Thunder Bay, if KEW5 will not work). If KEW5 is not selected as base provider, click the Select button to select it. For the Output Folder page, the "use project folder" and "create a unique filename based on the input filename" buttons should be the default; if not, choose the one that is closest with the highest integrity rating. Other parks should use closer and more appropriate base stations.

6. Read through the details of the processing page. Most of the positions should be corrected, although some may fail to correct and will be filtered out. There should be a short chart for the file showing proportional estimates for accuracies of the points. Few points will be better than 0.5–1 m accuracy, and some may be >5 m. If the correction appears to fail, try running through the wizard process again; if it still fails, try using another base station (KEW6 or Thunder Bay). Using another base station is important if many waypoints appear unprocessed. This may mean a base station was not operating during the time these points were collected.

Converting to .shp files for use in ArcView

1. In GPS Pathfinder Office, click Export (also under Utilites). Recently corrected (.cor) files should automatically be selected. If not, Browse to the files. Under the Choose an Export Setup, make sure the "ESRI Shapefile – UTM16" is chosen (for other parks, ensure which UTM band is appropriate); this is an export file made for rock pool work and can be used for any Isle Royale data. This file should be a UTM projection in Zone 16 North, with a Datum of NAD1983 (Conus) CORS96. Also make sure the Output Folder is "IsleRoyaleRockPoolMapping/Export." Click OK. **NOTE**: if "ESRI Shapefile – UTM16" is not available, a new export setup should be made to ensure the correct projection between rock pool datapoints and other shape files such as maps. To make a new export projection, click New from the export setup section, then New ESRI Shapefile; Browse in the Coordinate System tab to Projected, UTM, NAD1983 UTM Zone 16N.prj file. Name the file something like "ESRI shapefile – 16N NAD83 ISRO." In the position filter tab select "include non-GPS positions" and "include positions that are uncorrected." Once this is set up you should not need to do it again unless parameters change or you're on a new computer.

2. Go to the export folder (the default folder for new files in this system) and transfer the new files into the Rock Pool GIS Files folder. Pathfinder may rename the files to something that does not resemble the original file name; if so, look for the date the file was created.

3. Open ArcMap. Click Add Data and add the necessary RGB.sid aerial photos of Isle Royale (C:\ISRO_GIS\AerialPhotos2009ISRO20090517\MRSIDS\); then navigate to the Rock Pool GIS Files folder and add the new .shp file(s). Zoom to Full Extent (the globe icon) and make sure the datapoints are overlaid with the Isle Royale image in the appropriate location. Incorrect projection can make datapoints show up in very far-away places; if this is the case, the Isle Royale map will be shown in one part of the extent, while the datapoints will be shown in another. To find a feature on a large, blank map, click Find (black binoculars icon), type the

number “1” into the Find category, and click Find. The value list should show the number 1 (corresponding to the automatically generated FID number assigned to a datapoint in the new shape file); right-click on the 1 and “flash” the object for a brief crosshair to show its location, or zoom to the object to find where it is on the map. If the new datapoints align correctly (there will be some error, such as points in the lake or forest) then the process is complete. Back-up all data files (shp, ssf, cor, and txt) on different computers and/or a flash drive.

Additional Notes and Suggestions

If mapping other taxa is of interest to a monitoring program, mapping may be divided into two parts: habitat and biota. Separating these tasks will speed data collection, as the biotic inventory of each pool is time-intensive and requires a detailed knowledge of invertebrate life cycles. Habitat mapping can give a general idea of the spatial components of habitat densities, along with knowledge about pools such as size, distance to important features (like other habitats), recharge sources, or other stratifications deemed important. Biota mapping can use a subset of transects that measure the same metrics from habitat mapping, but will also determine presence and abundance of taxa in each pool. Taxonomic refinement to genus level will not generally be possible without collections and keying specimens under a microscope, but family or subfamily identifications may be possible in the field. The goals of the particular program, including focal taxa, will determine what taxonomic resolution is needed and possible. Biota mapping can be included within habitat mapping or may be done separately. Either way, biota mapping will identify habitat metrics favored by different taxa or communities and can then be extrapolated to the entire habitat dataset. This will generate baseline community data, estimates of relative abundance, species/community use of habitat types, and expected distribution of taxa.

The creation and completion of a database for all coastal rock pools is likely to take several months, depending on the length of shoreline, density of pools, equipment available, and employees available. At Isle Royale, pools in some locations were very dense. Passage Island required 2+ employees 11 days to map over 45,000 pools. Alternatives to this time-intensive (yet highly accurate) method may include breaking shorelines into segments (e.g., 1/5 km or 1/8 mi) and having a tally sheet for pool metrics of interest. For example, each sheet may have tallies for total number of pools, number of permanent or ephemeral pools, number of splash zone or lichen zone pools, number of pools with particular amphibians present, or other metrics. When pools have taxa of interest present, those pools may be measured in detail, including spatially. Each shoreline segment can then be geospatially referenced with data linked to the polygon, line, or point that represents the segment. As a result, coastal sensitivity or change in rock pool habitat and communities, based on predefined metrics of interest, can be assessed more quickly and on a regular basis.

STANDARD OPERATING PROCEDURE 9 – Data Management

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

Data management guidelines assure that acquisition, storage, management and archiving of data will be standardized, useful, and accessible into the future (Hart and Gafvert 2006). Also included is a plan to ensure the data quality. A project manager or principle investigator must have responsibility for ensuring that the following guidelines and procedures are followed during an inventory and monitoring project. For national park units in the Great Lakes Network (GLKN) region, data management should closely follow GLKN guidelines for consistency and compatibility, which these guidelines and procedures do. Refer to Hart and Gafvert (2006) for further details on the GLKN data management plan.

Basic components of the rock pool project (adapted from Hart and Gafvert 2006):

- Collect and archive raw data
- Enter/import data
- Validate data and archive validated data
- Analyze data and produce a report
- Archive data products (report, database)

Standard operating procedures for data collection, both in the field and in the laboratory, can be found in SOPs 4 and 5. Details and variations from the SOP may exist, such as use of different field computers or recording different metrics of interest, but all staff or cooperators collecting or analyzing data must read and follow the SOPs to ensure quality and consistency of data. All staff must be trained by the project leader or principal investigator. Quality assurance and quality control (SOP 8) includes using standard data sheets (for hardcopies) or data dictionaries (for electronic data input), with as much information pre-written as possible so as to reduce the likelihood ofmistakes during data input. Field leaders must double-check data entry before leaving a site, while events are easy to remember and missing data are still possible to collect. Upon returning to the office, electronic data should be downloaded and reviewed, and hardcopy data entered to a spreadsheet or database. Lab work also requires QA/QC procedures, such as reprocessing 10% of samples to check for missed specimens. All data should be validated by the project leader or principal investigator before data analysis. During all stages of the research process, all files must be backed-up in multiple locations, ideally including a park or Network server. If personal computers are used for data entry and analysis, all versions of all files must be transferred to a park computer or server; this is particularly important at the end of a season and during staff turnover.

Data File Standards

Data files should follow standards set by the GLKN office (Hart and Gafvert 2006). Unless a specific need exists, word processing should be done in Microsoft Word or Adobe Acrobat, while databases should be created in Microsoft Access. To keep track of file versions, use the following conventions:

- No spaces or special characters in the file name (instead, separate elements with underscores)
- Include a date in the file name (yyyymmdd)
- Include the status of the file in the name (such as "draft" or "final")
- Include notes on peer review personnel in the name (e.g., "AEcomments")

Examples of the above conventions:

"ISRO_Rock_Pool_Protocols_draft_20120323" indicates an Isle Royale rock pool protocol draft that was updated on 23 March 2012, or

"ISRO_Rock_Pool_Protocols_draft_20120323_TLcomments" indicates the same document with review comments by Toben Lafrancois.

Database Design

Design databases in Microsoft Access following criteria in Hart and Gafvert (2006). The structure will be based on field forms and the GIS data dictionary developed for the project. Core fields include general data that may be useful to other projects or databases, while project-specific fields are those that will be primarily useful to rock pool or coastal habitat investigators. The project-specific fields can be modified to suit different variations of rock pool studies, but the core fields should always be part of a database.

Core database fields:

Site name (general)	Date
Park code	Time
Observer (primary)	Geographic coordinates
Observer/s (secondary)	

Project-specific database fields (examples):

Project name	Pool length
Site description	Pool width
Project sample number	Order observed
Sky conditions	Family observed
Wind conditions	Order/family observed notes
Air temperature	Order collected
Water temperature	Family collected
Recent rain or waves	Family collected notes

Ecological zone (splash, lichen, or transition)
Pool Permanence (permanent, ephemeral)
Distance to lake
Distance to forest
Pool water source/s
Pool depth

Genus collected
Genus collected notes
Species collected
Species collected notes
Other taxon collected

Literature Cited

Hart, M., and U. Gafvert. 2006. Data management plan: Great Lakes Inventory and Monitoring Network. National Park Service Great Lakes Inventory and Monitoring Network Report. GLKN/2006/20. National Park Service, Ashland, Wisconsin.

STANDARD OPERATING PROCEDURE 10 – Quality Assurance/Quality Control for Rock Pool Data

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

This SOP closely follows Great Lakes I&M Network procedures for data management, which is summarized and modified below; for additional information on GLKN procedures refer to Hart and Gafvert (2006). Quality assurance and quality control (QA/QC) procedures will be used to identify and correct errors of data entry, data collection, and data transmission. This process includes using standardized data sheets to ensure that all data of interest are collected while in the field, since returning to collect data can be expensive, time consuming, or not possible if too much time has passed. Double-checking data inputs is important to catch simple errors such as a misplaced decimal point or incorrect site label, which can compromise a dataset if not noticed early. Finally, reprocessing a subset of samples in the lab ensures that the primary observer adequately processed the original samples. Field staff must read protocols and follow them for the process to maintain integrity.

Data Collection – Field Work

Field forms can generally be printed on regular paper since sample collection should not be done in rainy conditions, but using waterproof paper may be useful both because it is thicker and so unexpected contact with water will not damage it. Data entry in the field should always be done in pencil or waterproof ink so data are not lost from water damage. Data entry should generally avoid codes or truncating words since individuals may interpret these differently, especially after employee turnover or when read by people who are not trained in the same field. If codes are needed, a standardized list should be created and included in final reports. If two or more people are working in the field, data entry errors can be avoided by having one person make measurements and vocalize the measurements to a second person who records the data while repeating the measurements back to the observer.

During field work, the project/field crew leader must review the transcribed data prior to leaving the work site, ensuring that all necessary fields have been completed and that values, text, or codes all make sense. Data sheets should have a box for initials to keep track of quality control throughout the processes of collecting and managing data (see Coastal Rock Pool Survey Macroinvertebrate Form in SOP 4). To change field data, draw two lines through the original data and enter the new data next to it; do not erase original data.

Field data should be copied into a database (Microsoft Access, unless there are justifications for another program), and all data from the field sheets and database files should be compared to each other in batches (weekly or after a full sampling round) to ensure correct transcription. Data from field forms should be copied exactly into electronic files. These procedures should be done by personnel familiar with the project and the database, and ideally should be done with one person reading data while another inputs the data. After entering data, everything should be verified to catch mistakes such as misspellings, incorrect entries, or values that do not make sense.

Hard-copy files should be archived (in binders or scanned as pdf files) and multiple electronic files should be saved in different locations: one file on a work computer, one on a supervisor's computer, and a backup on a computer or server not at the same site such as at the Great Lakes Network office. A separate folder within an appropriate subfile system should be created and clearly named for the project (e.g., C:\Documents\Inventory and Monitoring\Rock Pool Project).

Data Collection – Laboratory Work

Laboratory work also requires quality control procedures. After general processing, such as picking or sorting, every tenth sample (10% of total samples) should be double-checked by an observer other than the person who initially processed the sample, using the same procedures. For example, after samples collected from the field are picked, where all specimens of interest are removed from the sample jar, cleaned of detritus and algae, and placed in new jars/vials, a second person will re-process every tenth original sample jar starting with the first. If during re-processing more than five specimens of interest are found, then that sample, along with the next nine (up to but not including the next re-processed sample) will be processed again to look for missed specimens.

Validation of Data

Validation of all data is a review process that is done by someone with strong knowledge of the project and dataset. QA/QC review may be challenging for large databases, such as those created during mapping with field PCs where huge amounts of data may be created weekly; in these cases, minor mistakes may not be obvious. Instead, it may be necessary to look for obvious inconsistencies such as a pool that is 11 cm wide by 25 cm long but the depth cell shows 155 cm, which likely means an extra "5" was entered. Another mistake might include American toad larvae being listed for small pools, yet no toad larvae were encountered during the week or the habitat is suspect. Many mapping mistakes may be found by projecting data onto a basemap, modifying outputs, and looking at the results to see if any points appear to be outliers or don't look as expected. The database can then be consulted for these points, and revisiting the site or removing the datapoint may be necessary.

Changes made during validation will be included on original hardcopy files (two lines striking out original data, with changes entered next to it) and on electronic files. For electronic files, changing data without removing original data may not be possible or easy in some programs (e.g., ArcView), so a field should be included in Access that denotes a change was made in a record, and the original data should be entered into a comments/notes section. During validation, ensure that all field sheets are accounted for in the dataset. A primary validation technique used by GLKN includes predefined look-up tables or data ranges, which should be included for many data dictionary entries on Trimble Juno field PCs used during mapping (see SOP 8). This both speeds data collection and reduces error.

Another validation technique includes searching for outliers using a graphic or statistical tool. Since extreme values may actually be correct, detected outliers should be checked against datasheet entries or some other source (e.g., odd weather entries can be checked against National Weather Service data or park weather records, and extreme pool sizes can be checked by placing the point on a high-resolution aerial image to see if it reasonably matches data). If data appear correct, noting it in a comments box with initials and the date will keep others from having to check it again. All the above procedures must be done prior to data analysis and reporting.

Literature Cited

Hart, M., and U. Gafvert. 2006. Data management plan: Great Lakes Inventory and Monitoring Network. National Park Service Great Lakes Inventory and Monitoring Network Report. GLKN/2006/20. National Park Service, Ashland, Wisconsin.

STANDARD OPERATING PROCEDURE 11 – Reporting

Version 1.00
Prepared by Alex Egan

Previous Version	Version Date	Author	Changes Made	Reason for Change	New Version

Reporting results of projects is important for several reasons: informing managers of the most recent knowledge of resource conditions, creating a referral document summarizing and/or analyzing knowledge to date, and guaranteeing results that are meaningful. Reporting standards below largely conform to GLKN reporting standards (Hart and Gafvert 2006, Route and Elias 2007), which are similar to other NPS Inventory and Monitoring Network standards (e.g., Long and Mitchell 2010).

Reporting is primarily the responsibility of the project manager, though other staff associated with the project can be included when drafting reports. A report should be prepared annually, even if it is only a summary of findings and notes/observations from the season. At least limited analysis, both from annual results and including prior data if applicable, should be included in annual reports to inform park managers. As the individual parks will be the main audience for these reports, review by and submission to the parks is appropriate. Ideally, the report will include:

- an overview and justification of the project, including goals and objectives,
- methods used both for field work and analyses,
- changes or problems that may have occurred during the current sampling and suggestions for improvements,
- results and summaries of data (including appendices with raw data tables, if useful),
- discussion and interpretation of findings to date, and
- acknowledgements that list all personnel who contributed to the annual effort.

On a longer time scale, synthesis reports should be done approximately every 5 years. These will include in-depth analysis of data and may act as a basis for publication in a scientific journal. These reports, along with journal articles, will be important as the foundation of informing both park managers and the public regarding changing resource conditions. Following synthesis reports or publications, data should be presentated to the scientific community in symposia. Presentations to the public, as part of an interpretive program in the parks where the research is occurring, should also be considered. Although the audience for synthesis reports is the broader scientific community, presentations and journal articles should be reviewed by park managers so sensitive information on species or landscapes will not become public knowledge. If peer reviewing is not available within the park system, synthesis reports should be submitted as one or more manuscripts to appropriate scientific journals.

Another type of reporting includes reviewing the protocol itself, which should occur every 3–5 years if a sustained program is in operation. Modifications for improvement must be evaluated for consistency to the goals and objectives of the protocol. In addition, the review will ensure that data collection and handling procedures, as implemented by staff, continue to be in compliance with accepted protocols. These reviews should evaluate not only the data collection and handling, but also how data are reported and where improvements can be made. Revision of the protocol should not compromise the original goals and objectives. Revision should only occur following a review by park managers. Clear documentation of changes, including date, approving personnel, details of the change and justification, must be listed at the beginning each standard operating procedure.

Literature Cited

Hart, M., and U. Gafvert. 2006. Data management plan: Great Lakes Inventory and Monitoring Network. National Park Service Great Lakes Inventory and Monitoring Network Report. GLKN/2006/20. National Park Service, Ashland, Wisconsin.

Long, J. D., and B. R. Mitchell. 2010. Northeast Temperate Network long-term rocky intertidal monitoring protocol. Natural Resource Report NPS/NETN/NRR—2010/280. National Park Service, Fort Collins, Colorado.

Route, B., and J. Elias (editors). 2007. Long-term ecological monitoring plan: Great Lakes Inventory and Monitoring Network. Natural Resource Report NPS/GLKN/NRR—2007/001. National Park Service, Fort Collins, Colorado.

STANDARD OPERATING PROCEDURE 12 – Revising the Protocol

Version 1.00
Prepared by Alex Egan

Previous Version	Revision Date	Author	Changes Made	Reason for Change	New Version

Making changes to the SOPs requires following procedures and documenting changes so that institutional memory is not required to understand the reasoning behind changes. Changes may be required if better techniques are established or methods need modification to effectively survey different sites. Keeping track of versions is important to link annual surveys with the correct procedures. The following procedures are modified from Ammann and Raimondi (2008).

1. Minor edits do not require a full review, but the project manager should consult with the crew, crew leader, and other natural resource managers before making the changes.

2. Major changes should be enacted only after a full review of the protocol by experts in the field.

3. Changes will be documented fully in the annual report and in summary at the beginning of each protocol that is changed. Minor changes are noted in hundredths (e.g., 1.01 will become 1.02). Major changes are noted in whole numbers (e.g., 1.02 will become 2.00). Note the previous version, revision date, author(s), changes made, reasons for change, and the new version number.

4. Inform appropriate personnel, such as data managers or resource managers, of the changes so databases and metadata can remain up-to-date.

Literature Cited

Ammann, K. N., and P. T. Raimondi. 2008. Long-term monitoring protocol for rocky intertidal communities of Redwood National and State Parks, California. Natural Resource Report NPS/KLMN/NRR—2008/034. National Park Service, Fort Collins, Colorado.

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