ABSTRACT

METHODS TO MONITOR LAKE ERIE’S HARMFUL ALGAL BLOOMS: A FELLOWSHIP WITH THE COOPERATIVE INSTITUTE FOR GREAT LAKES RESEARCH

by Deanna Lynne Fyffe

To fulfill the professional experience requirement for a Master of Environmental Science degree at Miami University, I completed a fellowship with the Cooperative Institute for Great Lakes Research (CIGLR). My work involved investigating data trends of recent Lake Erie harmful algal blooms and potential monitoring methods. Data trends revealed monitoring stations closest to the mouth of the Maumee River had the highest average cyanobacteria concentrations. Bloom biomass distribution tended to favor the surface of the water column but was likely influenced by wind speed in 2016 and 2017. I also compared chlorophyll-a data from a CIGLR-owned bbe FluoroProbe to laboratory extraction data. The bbe FluoroProbe provided consistent results when used in the field and in the laboratory, but generally identified less chlorophyll-a than pigment extraction methods. Additionally, I performed field, laboratory, and analytical work to evaluate commercial in situ fluorometers. Due to proprietary reasons, the individual fluorometer data could not be presented in this report. I provided an example field deployment evaluation for the YSI EXO2 Multiparameter Sonde, a CIGLR-owned fluorometer that was used during the field tests for ancillary data. The EXO2 ultimately had low accuracy when compared to traditional laboratory methods, but both methods produced similar data trends.
METHODS TO MONITOR LAKE ERIE’S HARMFUL ALGAL BLOOMS: A FELLOWSHIP WITH THE COOPERATIVE INSTITUTE FOR GREAT LAKES RESEARCH

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by
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RESEARCH

by

Deanna Lynne Fyffe

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<table>
<thead>
<tr>
<th>Acronym</th>
<th>Description</th>
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<tr>
<td>ACT</td>
<td>Alliance for Coastal Technologies</td>
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<tr>
<td>BB3</td>
<td>Basic Blue 3 Dye</td>
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<tr>
<td>CDOM</td>
<td>Colored Dissolved Organic Matter</td>
</tr>
<tr>
<td>CGLSLGP</td>
<td>Conference of Great Lakes and St. Lawrence Governors and Premiers</td>
</tr>
<tr>
<td>CILER</td>
<td>Cooperative Institute for Limnology and Ecosystems Research</td>
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<tr>
<td>Chl-a</td>
<td>Chlorophyll-a</td>
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<td>CIGLR</td>
<td>Cooperative Institute for Great Lakes Research</td>
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<td>DMF</td>
<td>Dimethylformamide</td>
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<td>EXO2</td>
<td>YSI EXO2 Multiparameter Sonde</td>
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<tr>
<td>fDOM</td>
<td>Fluorescent Dissolved Organic Matter</td>
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<td>GLERL</td>
<td>Great Lakes Environmental Research Laboratory</td>
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<td>GLISA</td>
<td>Great Lakes Integrated Sciences and Assessments</td>
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<td>GLRI</td>
<td>Great Lakes Restoration Initiative</td>
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<td>GLWQA</td>
<td>Great Lakes Water Quality Agreement</td>
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<td>HAB</td>
<td>Harmful Algal Bloom</td>
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<tr>
<td>HABHRCA</td>
<td>Harmful Algal Bloom and Hypoxia Research and Control Act</td>
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<tr>
<td>ISSHA</td>
<td>International Society for the Study of Harmful Algae</td>
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<td>LaMP</td>
<td>Lake Erie Lakewide Management Plan</td>
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<tr>
<td>NOAA</td>
<td>National Oceanic and Atmospheric Administration</td>
</tr>
<tr>
<td>ODH</td>
<td>Ohio Department of Health</td>
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<tr>
<td>Ohio EPA</td>
<td>Ohio Environmental Protection Agency</td>
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<tr>
<td>PC</td>
<td>Phycocyanin</td>
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<tr>
<td>PSP</td>
<td>Paralytic Shellfish Poisoning</td>
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<tr>
<td>SI</td>
<td>Severity Index</td>
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<tr>
<td>WHO</td>
<td>World Health Organization</td>
</tr>
<tr>
<td>WLE</td>
<td>Western Lake Erie</td>
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<tr>
<td>US EPA</td>
<td>United States Environmental Protection Agency</td>
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Section 1: Introduction to Harmful Algal Blooms

Harmful algal blooms (HABs) are blooms of phytoplankton growth that have negative effects on the ecosystem around them (NOAA, 2017a). HABs can occur in both marine and freshwater and are composed of microscopic algae called phytoplankton. Phytoplankton include multiple microbial classes such as dinoflagellates, diatoms, cyanobacteria, cryptophytes, and chlorophytes (Lindsey & Scott, 2010). Phytoplankton are photosynthetic organisms, meaning they use visible light as an energy source (Oliver & Ganf, 2000). They are the foundation of the aquatic food web and are crucial to the ecosystem. However, when they grow uninhibited and form HABs, they negatively impact the environment, economy, and human health (US EPA, 2017a).

Marine HABs are frequently composed of dinoflagellates and are commonly called “red tides” due to their red appearance (Anderson, 1989). Red tides occur in locations all over the world, including New England, Argentina, and Hong Kong. Some dinoflagellates can produce a type of neurotoxin that causes Paralytic Shellfish Poisoning (PSP) (US EPA, 2016a). Red tides cause PSP when shellfish ingest the phytoplankton and their toxins, which accumulate within the shellfish and are passed on to organisms higher in the food chain.

Freshwater HABs, especially in North America, are usually composed of cyanobacteria, commonly known as blue-green algae (Watson, et al., 2016). Cyanobacteria are a type of bacteria that live in aquatic and terrestrial environments all over the world (Whitton & Potts, 2013). Although they are not actually algae, cyanobacteria have pigments necessary to perform photosynthesis, so they are also classified as phytoplankton (Oliver & Ganf, 2000). Cyanobacteria thrive in nutrient-rich waters that supply ample amounts of phosphorus and nitrogen needed for their growth (US EPA, 2017a).

HABs are the result of many forces in a waterbody, including nutrient pollution, food web alterations, introduced species, water conditions, and climate change (NOAA, 2017a). The factors that lead to HAB formation are different depending on geographic location and phytoplankton class. However, nutrient pollution is a common factor that plays into most HAB formations (US EPA, 2017a). Nutrient pollution occurs when a surplus of nutrients, mainly nitrogen and phosphorus, are washed into a water body (US EPA, 2017b). The nutrients commonly come from agricultural and urban water runoff. Phytoplankton use these nutrients to
grow, so when the nutrients are in excess, the phytoplankton can grow uninhibited by food availability. Increased agricultural and industrial land use can also cause nutrient pollution by destructing wetlands, which filter out nutrients (Mitsch & Gosselink, 2007).

HABs are harmful for a variety of reasons. First, they can be toxic (US EPA, 2016a). As mentioned, the dinoflagellates in marine HABs can produce toxins that cause PSP. Likewise, certain cyanobacterial genera can produce toxins that affect the liver (hepatotoxins), brain (neurotoxins), and skin (dermatoxins) (WHO, 1999). These toxins are harmful to organisms living in that aquatic ecosystem and to humans. Humans can be affected by hepatotoxins and neurotoxins through ingestion when using contaminated water as a source of recreation or drinking water. Dermatoxins can cause rashes and blisters on skin from direct contact with the toxins.

Second, HABs can deplete the oxygen in a waterbody. When the phytoplankton die, they are degraded through a process that uses oxygen (Hawley, et al., 2006). When there is a surplus of phytoplankton, the constant degradation can deplete oxygen in the water to extremely low levels, a condition known as hypoxia. Lack of dissolved oxygen alters a normally productive water body to be uninhabitable for many organisms. Fish are stressed in waters with less than 4 mg O_2/l, and all other organisms are stressed at less than 2 mg O_2/l. The area becomes uninhabitable when it has no dissolved oxygen (anoxic) (Hawley, et al., 2006).

Third, HABs can impact the economy through many different industries including public health, fisheries, and tourism (Hoagland & Scatasta, 2006). There are also costs associated with monitoring and managing the HABs. A study in 2005 suggested the European Union experiences economic losses of approximately $813 million (USD) a year between these four sectors (Table 1.1) (Hoagland & Scatasta, 2006). Their highest anticipated effect was due to

<table>
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<tr>
<th>Industry</th>
<th>Annual Economic Impact (Million USD)</th>
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<tr>
<td></td>
<td>European Union</td>
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<tr>
<td>Public Health</td>
<td>11</td>
</tr>
<tr>
<td>Commercial Fisheries</td>
<td>147</td>
</tr>
<tr>
<td>Recreation and Tourism</td>
<td>637</td>
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<tr>
<td>Monitoring and Management</td>
<td>18</td>
</tr>
<tr>
<td><strong>Total</strong></td>
<td><strong>813</strong></td>
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Table 1.1 Estimated Annual Economic Impacts of Harmful Algal Blooms in the European Union and the United States of America (Hoagland & Scatasta, 2006). The European Union is estimated to have an $813 million US dollar impact on their economy with the industry with the largest effect being recreation and tourism. The United States is estimated to have an $82 million economic impact with public health and commercial fisheries experiences the largest effects.
loss of recreation and tourism. The study also estimated an $82 million annual impact in the US, with the highest impact in public health and commercial fishery industries.

Additionally, there are costs to treat drinking water contaminated with toxins. City officials in Auburn, New York claim costs to treat drinking water contaminated with microcystin can cost $1.5 to $6 million each year depending on treatment method (Craig, 2017). An alternative solution for the City of Auburn was to extend the water intake pipe to reach non-HAB infested waters, which would cost $15 million.

Internationally, HABs began getting attention in 1974 with the First International Conference on Toxic Dinoflagellate Blooms (ISSHA, 2017a). The number of countries attending the conference grew from three in 1974 to twenty-two in 1985, demonstrating the increasing concern regarding this issue (LoCicero, 1974; Anderson, White, & Baden, 1985). In 1997, the conference was renamed to the International Conference on Harmful Algae in order to include locations experiencing non-dinoflagellate blooms (ISSHA, 2017b). HABs continue to affect more and more countries around the world (Figure 1.1) (Anderson, 2009). During the 2014 conference, 34 countries attended the five-day conference in New Zealand (ISSHA, 2015).

![Figure 1.1 Global Locations with Paralytic Shellfish Poisoning Toxins in 1970 and 2005 (Anderson, 2009). In 1970, there were only fifteen locations around the world with reported PSP toxins. As of 2005, every continent except Antarctica had at least three locations with PSP toxins. Image courtesy of the United States National Office for Harmful Algal Blooms, Woods Hole Oceanographic Institution.](image-url)
In order to address the growing concerns of marine HABs and hypoxia along the United States coasts, Congress authorized the Harmful Algal Bloom and Hypoxia Research and Control Act (HABHRCA) in 1998 (33 U.S.C § 4001 et seq). HABHRCA created an inter-agency task force to complete thorough assessments of HABs and hypoxia. The assessments addressed economic and ecological impacts as well as potential alternatives for reducing, remediating, and controlling the HABs. The potential alternatives were also assessed for their associated economic and social costs and benefits. HABHRCA also mandated the National Oceanic and Atmospheric Administration (NOAA) to continue researching HABs and hypoxia.

In 2004, the HABHRCA was amended to include freshwater ecosystems, specifically the Great Lakes (Public Law 108-456). Additional amendments in 2014 mandated NOAA to continue researching marine and Great Lakes water systems and enhancing their capacity to observe, monitor, model, and forecast HAB and hypoxia events (Public Law 113-124, 2014). Research conducted under this law indicates that approximately 39% of all United States lakes have detected the cyanotoxin microcystin with a significant increase in the number of lakes affected between 2007 and 2012 (US EPA, 2016b). In 2016, the United States Geological Survey reported that 43 states experienced human or animal illness or death as result of HABs (Figure 1.2) (Graham, Dubrovsky, & Eberts, 2016). In August 2016 alone, at least 19 states had public health advisories due to HABs. These findings indicate that the effects of HABs continue to cause significant harm to ecosystems and human health, which makes the research conducted by NOAA essential for environmental and public safety.

Figure 1.2 States Affected by Cyanobacterial Harmful Algal Blooms (HABs) (Graham, Dubrovsky, & Eberts, 2016). In August 2016, nineteen states had HABs resulting in beach closures and health advisories. Forty-three states have reported human and animal illness and death as result of HABs.
Section 2: Harmful Algal Blooms in Lake Erie

Lake Erie is the smallest, shallowest, and warmest of the five Laurentian Great Lakes (US EPA, 2016c). The Lake Erie Watershed supports approximately 11.6 million people, one-third of the total population within the Great Lakes basin, making it the most populated of the Great Lakes watersheds (Lake Erie LaMP, 2011). Out of all the Great Lakes, Lake Erie is exposed to the most stress from agriculture, urbanization, and industrialization. In 2017, Lake Erie was found to be the only Great Lake in poor, and deteriorating, condition for nutrients and HABs (US EPA and Canada, 2017). Lake Erie has a long history of HABs, with the worst on record occurring in 2015 (Figure 2.1) (NOAA Great Lakes CoastWatch, 2015).

Prior to European settlement, 1,500 square miles southwest of Lake Erie’s western basin was covered by marshland, which helped filter water before it entered the lake (Allinger & Reavie, 2013). At this time, the area had pristine wildlife and natural forest cover (Mitsch & Gosselink, 2007). After European settlement, the marshland and forestland were converted to agricultural fields (Hartman, 1973). Limnological surveys of Lake Erie in the 1920’s indicated both nutrients and contaminants were being discharged into the Lake (Wright, Tiffany, & Tidd, 1955; Wright & Tidd, 1933). The surveys also indicated that the western basin had more phytoplankton biomass than the central and eastern basins (Wright & Tidd, 1933). The western basin, which is

![Figure 2.1 Satellite Image of the Lake Erie Harmful Algal Bloom on August 22, 2015 (NOAA Great Lakes CoastWatch, 2015). The bloom of 2015 was the most severe on record. It can be seen encompassing the western basin of Lake Erie and extending into the central basin.](image-url)
the shallowest of the three basins, has three major tributaries (Detroit, Maumee, and Sandusky Rivers). Subsequent research found that the amount of nitrogen and phosphorus being loaded into the lake continued to increase, and phytoplankton biomass was twenty-fold higher in 1960 than it was in 1919 (Hartman, 1973).

With Lake Erie’s conditions continuing to deteriorate, the United States of America and Canada signed the Great Lakes Water Quality Agreement (GLWQA) in 1972 (USA and Government of Canada, 1972). This agreement proposed the creation of pollution prevention programs to mitigate pollution from population growth, increased development, and unsustainable use of Great Lakes water (USA and Government of Canada, 1972). In 1978, the GLWQA was amended to limit future phosphorus loading to 11,000 metric tons per year (USA and Government of Canada, 1978). This limit was significantly lower than the 20,000 metric ton load that occurred in 1976. The limit was first met in 1981 through reduction of point sources of phosphorus pollution such as wastewater treatment plants and industries that used phosphorus-containing chemical detergents (Baker, 2007). Monitoring efforts indicated the decrease in phosphorus loading was succeeding in reducing phytoplankton biomass across the lake (Makarewicz, 1993).

The phytoplankton biomass continued to decrease through the 1980’s when quagga and zebra mussels invaded and colonized Lake Erie (Nicholls & Hopkins, 1993). However, starting in the mid-1990’s and continuing through today, Lake Erie’s western basin has been subject to annual blooms from the cyanobacteria *Microcystis aeruginosa* (Allinger & Reavie, 2013). *M. aeruginosa* are colony-forming cyanobacteria that have the capability to produce the toxin microcystin (Figure 2.2) (Rinta-Kanto, et al., 2009).

Microcystin is a hepatotoxin that can cause numbness, motor weakness, incoordination, and respiratory and muscular paralysis (WHO, 1999).

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*Figure 2.2 Microscope Image of Microcystis aeruginosa (US EPA, 2016a).* *M. aeruginosa* are colony-forming cyanobacteria capable of producing microcystin toxins. Photo courtesy of John D. Wehr, Professor at Fordham University. Water sample was collected from Lake Mahopac in New York.
Microcystin can exist both inside and outside the cell, known as particulate and dissolved microcystin respectively. Microcystin is released into the environment when a Microcystis cell dies and lyses (WHO, 1999). Microcystis cells have gas vesicles which allow them to change buoyancy to move around the water column as well as form scums at the water surface (Reynolds & Walsby, 1975; Sejnohova & Blahoslav, 2013).

Efforts to understand the Lake Erie HABs have significantly increased in the twenty-first century. The GLWQA was amended in 2012 to outline the current main concerns regarding the Great Lakes: reducing phosphorus loads and algal blooms, restoring the ecosystems and biological communities, strengthening the delivery of science and information on the Great Lakes, and engaging and educating the public (USA and Government of Canada, 2012). The 2012 amendment also mandated that the phosphorus load limit be reassessed.

Research and remediation efforts in the Great Lakes were strengthened with the creation of the Great Lakes Restoration Initiative (GLRI) in 2010. Through GLRI, a task force was formed comprised of representatives from eleven federal departments, councils, and agencies that are working together to restore the Great Lakes through research and funding (GLRI, 2017). The GLRI aims to clean up areas of concern, control and prevent invasive species, reduce nutrient runoff, and restore habitats for native species protection. As of February 2017, there were 3,455 projects in progress or completed, 224 of which were conducted by NOAA (GLRI, 2017). As of April 2017, 62 of those NOAA GLRI projects were still in progress.

In addition to federal and individual state initiatives, Michigan, Ohio, and Ontario are currently collaborating to reduce phosphorous loading into Western Lake Erie (WLE). In 2015, the Western Basin of Lake Erie Collaborative Agreement set the goal of reducing 20% of phosphorous loading into WLE by 2020 and 40% by 2025 (CGLSLGP, 2015). This agreement serves to advance nutrient reduction efforts in Lake Erie proposed by the GLWQA. Since the agreement was signed, the Ohio EPA published an action plan, including a specific timeline, to address the nutrient pollution concerns (Ohio EPA, 2017).

Of the HABs that have occurred since 2000, the bloom in 2015 was the most severe, receiving a 10.5 on NOAA’s severity index (Figure 2.3) (NOAA, 2017b). The severity index (SI) is based on the HAB’s biomass, or amount of cyanobacteria, present in the lake for a sustained period of
time (NOAA, 2017c). The second most severe HAB occurred in 2011 and the third in 2013. The 2017 HAB was forecast to have an SI of 7.5, falling in line to be the fourth most severe HAB that Lake Erie has experienced since 2002, when the severity calculations began. The 2014 HAB, though only an SI of 6, is notable for causing a water crisis in the City of Toledo. An unpredicted wind event coupled with unusually high concentrations of microcystin caused the Toledo drinking water facilities to be overwhelmed by toxins (Steffen, et al., 2017). The treated water still had concentrations of microcystin above the World Health Organization’s guidelines, so the City of Toledo had a “Do Not Drink” advisory on the drinking water for three days. Though the 2015 HAB was the most severe, it was not as toxic as the 2014 bloom and therefore did not pose as high a threat to the drinking water facilities.

![Figure 2.3 Severity of the Past Fifteen Harmful Algal Blooms of Lake Erie and the 2017 Bloom Prediction (NOAA, 2017). The bloom of 2015 had the highest severity, reaching a 10.5 on NOAA’s severity index. The bloom of 2016 was only approximately a 3 on the severity index. The 2017 bloom was forecast to be a 7.5 but could range from 6 to 9.5 on the index. Figure courtesy of the National Oceanic and Atmospheric Administration.](image)
In addition to nutrient pollution, studies suggest the post-1990 Lake Erie HABs are primarily influenced by invasive mussel species, long-term agricultural practices, and climate change (Allinger & Reavie, 2013; Michalak, et al., 2013; GLISA, 2014). Specifically, it is suggested that the invasive mussels are selectively rejecting *M. aeruginosa* and instead consuming other phytoplankton species (Vanderploeg, et al., 2001). This provides an advantage to *M. aeruginosa*, who therefore have both fewer predators and competition. Additionally, mussels excrete phosphate and ammonia which are available for use by *M. aeruginosa* and other phytoplankton (Zhang, Culver, & Boegman, 2011).

Long-term trends in Ohio’s agricultural practices, specifically fall fertilizer application, surface fertilizer application, and conservation tillage, can increase the amount of phosphorus being loaded into Lake Erie (Michalak, et al., 2013). This is of particular importance in WLE, because land in Lake Erie’s largest subwatershed, the Maumee River Watershed, is predominantly used for agriculture and pasture (Johengen, 2017). Studies in this region found that between 1995 and 2011, a 218% increase in dissolved phosphorus loaded into Lake Erie from the Maumee River (Michalak, et al., 2013).

Another prominent factor affecting Lake Erie is climate change. Evidence for this includes increasing water temperatures, amount of precipitation, and occurrences of extreme weather events. Models predict that Lake Erie will continue to get warmer and as a result, be even more suitable for cyanobacterial growth (Trumpickas, Shuter, & Minns, 2009). Additionally, the amount of precipitation has increased 11% since 1900, and the number of extreme weather events have become more frequent and intense (GLISA, 2014). These events increase nutrient loading by way of agricultural and stormwater runoff, and the events are expected to occur at increasing frequency as climate change escalates.
Section 3: Fellowship Overview

Organization Background

The Great Lakes Environmental Research Laboratory (GLERL) is one of seven laboratories in NOAA’s Office of Oceanic and Atmospheric Research. All NOAA research labs are united by the single purpose of predicting and assessing significant changes in their relative aquatic environment, ensuring the resources of their focus area are managed safely and efficiently, and promoting the development of associated industries (NOAA, 2016). NOAA GLERL was established by the Office of Secretary in 1974 and exists under the United States Department of Commerce. NOAA GLERL’s mission is to conduct research regarding the Great Lakes to better understand the environmental processes at work within the lakes and the corresponding watersheds, as well as to focus on problem solving with resource management and environmental services of the region. NOAA GLERL provides products in the form of datasets, peer-reviewed publications, and annual reports regarding research, publications, and presentations of that year (NOAA, 2017d).

Figure 3.1 National Oceanic and Atmospheric Administration’s (NOAA’s) Great Lakes Environmental Research Laboratory Organizational Structure (NOAA, 2016). A director heads the organization, which separates into teams and research departments, eventually leading down to the NOAA Cooperative Institutes at the bottom. NOAA has 16 Cooperative Institutes, one of which is the Cooperative Institute for Great Lakes Research in Ann Arbor, Michigan.
The NOAA GLERL Director oversees individual research and administrative departments (Figure 3.1) (NOAA, 2016). There are four research departments: observation systems and advanced technology, ecosystem dynamics, integrated physical and ecological monitoring and forecasting, and information services. All four research departments are connected to NOAA Cooperative Institutes, one of which is the Cooperative Institute for Great Lakes Research (CIGLR). CIGLR is a consortium of universities and private sector partners throughout the Great Lakes region (CIGLR, 2017).

CIGLR is primarily sponsored by NOAA GLERL and is hosted by the University of Michigan’s School for Environment and Sustainability. CIGLR is composed of nine research partners, 25 university affiliates, and five private-sector partners, and is supported by nine initiatives and programs (see Appendix A for a complete list of consortium members). The institute prides itself on supporting the research and education of students and postdocs as well as continually publishing new journal articles and public reports. CIGLR’s main mission is to train the next generation of Great Lakes scientists to lead new, exciting research efforts and to transform scientific findings to be usable by society.

CIGLR was previously known as the Cooperative Institute for Limnology and Ecosystems Research (CILER). On May 16, 2017, the University of Michigan was awarded a grant to establish the new cooperative institute, replacing CILER (Cardinale, 2017). With this new cooperative institute, the university consortium can pursue broader, more impactful research projects regarding the Great Lakes. CIGLR aims to take a more interdisciplinary approach than CILER by having data users and research scientists co-design research initiatives. CIGLR will also work with a wider range of stakeholders within two Canadian provinces and eight United States.

At the top of the CIGLR organizational structure is the executive board and the council of fellows (Figure 3.2) (CIGLR, 2017). The executive board includes employees at NOAA and the University of Michigan. The council of fellows is composed of three GLERL branch chiefs, regional consortium principal investigators, and the CIGLR director, associate director, and program manager. Within CIGLR, the research institute, support staff, and regional consortium work together on various research projects.
Figure 3.2 Cooperative Institute for Great Lakes Research (CIGLR) Organizational Structure (CIGLR, 2017). CIGLR is governed by an executive board and council of fellows. The foundation of work performed by CIGLR is conducted by the research institute, support staff, and regional consortium. CIGLR works directly with the National Oceanic and Atmospheric Administration’s Great Lakes Environmental Research Laboratory.

**Fellowship Responsibilities**

During my fellowship with CIGLR, I worked on two main projects. First, I performed data organization and analysis regarding the last three Lake Erie HABs. Second, I helped collect field and laboratory data to evaluate four *in situ* fluorometers with the Alliance for Coastal Technologies. My specific responsibilities included:

1. **Harmful Algal Bloom Data Projects**
   a. Analysis of surface-to-bottom ratios of HAB parameters in Lake Erie
      - Created and organized spreadsheets calculating the surface-to-bottom ratios of particulate microcystin, dissolved microcystin, phycocyanin, and chlorophyll-α concentrations from 2015 - 2017
Investigated the relationship between wind speed and ratios for phycocyanin and chlorophyll-a

bbe FluoroProbe 2016 data interpretation and accuracy analysis

- Created spreadsheets calculating average phytoplankton concentrations and total chlorophyll-a identified by the FluoroProbe for both field and laboratory methods
- Investigated temporal and spatial trends in the data
- Compared FluoroProbe data from field and laboratory methods to investigate consistency of results
- Compared FluoroProbe data to extracted pigment data to investigate accuracy of the FluoroProbe

2. *In Situ* Fluorometer Evaluation

a. Field Work

- Moored Deployment – aided with reference sample collection and processing twice a day while the *in situ* fluorometers were deployed in the Maumee River for fourteen days
- Surface Mapping Deployment – performed the surface mapping and reference sample collections with a team

b. Laboratory work

- Filtered and analyzed reference samples for algal pigments
- Aided laboratory experiments on the *in situ* fluorometers using phytoplankton cultures of known concentrations

Section 4 will explain the data projects in greater detail, including the goal, methods, and results of the data analysis. Section 5 will elaborate on the fluorometer evaluation and will include an example field deployment evaluation of a fluorometer owned by CIGLR.
Section 4: CIGLR Harmful Algal Bloom Data Projects
Since 2009, NOAA GLERL has been collecting weekly water quality data from WLE between March and October every year. The water samples are collected from eight monitoring stations, four of which also have buoys equipped with sensors to provide continuous data (Figure 4.1) (NOAA, 2017e). The weekly samples provide data on nutrients, carbon, toxins, turbidity, and phytoplankton pigments. One of my fellowship projects was to use this data to investigate HAB spatial and temporal trends as well as use the data to assess the accuracy of the bbe FluoroProbe.

Figure 4.1 Weekly and Continuous Water Quality Monitoring Stations in Western Lake Erie (NOAA, 2017e). All stations are monitored weekly through water samples which are processed at the Great Lakes Environmental Research Laboratory. WE08, WE04, WE02, and WE13 provide continuous data via a buoy.

When a HAB is present, many organizations rely on this weekly data for decision making, especially when it comes to public health. Throughout the bloom, CIGLR and NOAA GLERL pay particularly close attention to the phytoplankton pigment and cyanotoxin concentrations, for they are representative of phytoplankton growth and current HAB conditions (Stumpf, et al.,
The pigments allow phytoplankton to use light as an energy source, and different phytoplankton groups often have different pigments for photosynthesis (bbe moldaenke, 2011). Each pigment absorbs certain wavelengths of light, which excites their electrons. When the electrons are excited, the pigments release energy through fluorescence. Chlorophylls and phycobilins are two major pigment classes that help convert light to energy (Millie, et al., 2010). The two pigments classes perform similar functions, but absorb and emit different wavelengths of light.

All phytoplankton have chlorophyll-a (Chl-a) (Whitton & Potts, 2013). Most cyanobacteria and some cryptophytes also have phycocyanin (PC), a type of phycobilin pigment. Chl-a absorbs blue (440 nm) and red (681 nm) light, whereas PC absorbs orange-red light (620 nm) (Stumpf, et al., 2016). Since cyanobacteria have both Chl-a and PC, either one can be used for analytical purposes. Chl-a can be more accurately analyzed in a laboratory setting since it is a standard water quality measurement that has been used and studied longer than PC (Stumpf, et al., 2016). However, Chl-a represents phytoplankton growth as a whole, while PC is more specific to cyanobacteria. Ultimately, I used both Chl-a and PC data in my investigations in Lake Erie HAB trends during the past few years.

**Analysis of Surface-to-Bottom Ratios for Laboratory Data**

For this project, I created a spreadsheet calculating surface-to-bottom ratios of water quality parameters associated with HABs. Specifically, those parameters were particulate microcystin, dissolved microcystin, extracted PC, and extracted Chl-a concentrations. The goal of this project was to investigate trends in HAB vertical movement to better aid development of NOAA’s Experimental Lake Erie HAB Tracker. This forecast model uses remote sensing imagery, weather forecasts, and water current models to estimate the location, intensity, and movement of the HAB throughout Lake Erie (NOAA, 2017f). The HAB tracker presents preliminary data on the vertical distribution of Microcystis, so this project aimed to build upon their current data inputs to make the predictions even more informed.

If the HAB Tracker can accurately predict vertical distribution in their forecasts, drinking water intake facilities could better plan when to intake water based on when the HAB is predicted to be away from the intake. Drinking water facilities generally try to construct water intakes at
maximum depths to minimize harmful pollutants or agents that usually congregate nearer to the water surface (City of Sylvania, 2016). However, *Microcystis* cells have gas vesicles that allow them to move vertically in water column, so they are not always most concentrated at the surface of the lake. Currently, cities often prefer to intake water when wind speeds are high, because the HAB will be more thoroughly mixed throughout the water column. If they intake the water when the HAB is mixed, they reduce the risk of taking in water with highly concentrated cyanobacteria and toxins. However, this method means that the city will intake the water knowing there is cyanobacteria present and significant water treatment will be required. Alternatively, if NOAA can more accurately model the vertical distribution of the HAB, the water intake could potentially choose days or times to intake water when they know the bloom will be concentrated at the water surface – away from the intake valve.

**Methods**

Weekly water samples from the surface and the bottom of the water column were collected at stations WE02, WE08, WE12, and WE13 (Figure 4.1). The surface samples were collected 0.75 m beneath the surface, and the bottom samples were collected 1.0 m from the lake bottom (NOAA, 2017d). Ratios for 2015 and 2016 were already calculated in separate spreadsheets, but the data files needed to be restructured and reorganized. After developing a new spreadsheet with a uniform organizational structure, I double checked the 2015 and 2016 data and calculated ratios for 2017 data as they came in.

I investigated the frequency of ratios greater than, equal to, and less than one for microcystin and pigments of the 2015, 2016 and 2017 HABs. I calculated the ratios by dividing the concentration at the surface by the concentration at the bottom. The PC ratio specifically represents the cyanobacteria distribution in the water column, while the Chl-α represents the distribution of phytoplankton in general. Dissolved microcystin ratios represent the distribution of free microcystin in the water column, while particulate microcystin ratios indicate distribution of cyanobacterial cells with toxins inside the cells.

A surface-to-bottom ratio of greater than one indicates that the concentration of PC, Chl-α, or microcystin was higher at the surface than it was at the bottom of the water column. If the ratio equals one, then the two concentrations are the same at both depths and the variable is evenly
distributed throughout the water column. Lastly, if the ratio is less than one, then the concentration at the bottom of the water column was higher than that at the top. I calculated the percentage of ratios greater than one, equal to one, and less than one for each parameter.

After the ratio distributions were determined, wind speed was added to the investigation. The goal of adding wind to the analysis was to see if PC and Chl-a stopped accumulating at the surface after a certain wind speed. Experimental results suggest that a wind speed of 2-3 m/s are needed to mix surface phytoplankton into the water column (Webster & Hutchinson, 1994). Models and experiments also suggest that Microcystis dominate a body of water in low turbulence conditions, for their buoyancy allows them to move at a higher velocity than the water (Huisman, et al., 2004). However, in more turbulent conditions, diatoms and green algae dominated the environment since Microcystis could no longer control their location within the water column.

Results
PC and Chl-a most often had a greater concentration at the surface of the lake. For both pigments, 70% of the ratios were greater than 1 while only 30% were less than 1 (Table 4.1). This coincides with the expectation that cyanobacteria accumulate near the water surface where the temperature and sunlight intensity are more optimal for their growth (Chorus & Cavalieri, 2000).

<table>
<thead>
<tr>
<th>Ratio</th>
<th>Particulate Microcystin</th>
<th>Dissolved Microcystin</th>
<th>Extracted Phycocyanin</th>
<th>Extracted Chlorophyll-a</th>
</tr>
</thead>
<tbody>
<tr>
<td>&gt; 1</td>
<td>54%</td>
<td>36%</td>
<td>69%</td>
<td>71%</td>
</tr>
<tr>
<td>= 1</td>
<td>2%</td>
<td>16%</td>
<td>0%</td>
<td>0%</td>
</tr>
<tr>
<td>&lt; 1</td>
<td>44%</td>
<td>48%</td>
<td>31%</td>
<td>29%</td>
</tr>
</tbody>
</table>

Ratios for particulate microcystin indicated that on average, it accumulated at the surface and the bottom an equal amount of time. Dissolved microcystin, however, did not appear to accumulate at a specific depth. Of all the ratios for dissolved microcystin, approximately 48% of the ratios indicated greater concentrations at the bottom of the water column, 36% indicated greater concentrations at the surface, and 16% indicated it was the same at both the surface and bottom. This suggests that the cells still producing microcystin toxins are located both at the top and the
bottom of the water column, and once the cells lyse and release the microcystin, it becomes distributed throughout the water column rather than clustering at a specific depth.

After the initial ratios were determined, I examined the relationship between wind speed and pigment ratios. The data from 2015 did not reveal a consistent relationship between biomass distribution (represented by Chl-α and PC ratios) and wind speed (Figures 4.2 and 4.3). Alternatively, in 2016 and 2017, it only took wind speeds of 5 m/s to more evenly distribute the biomass. When interpreting these data, it was important to consider how the HAB intensity varied between the years. The 2015 HAB was a 10.5 on NOAA’s severity index while the 2016 bloom was only a 3 (Figure 2.3). More specifically, the mean PC concentration in 2015 was seven times that of 2016 and twice that of 2017 (Table 4.2). This suggests that cyanobacterial biomass was highest in 2015, which may have played a role in the wind’s ability to mix the water column.

Table 4.2 Western Lake Erie Mean Chlorophyll-a and Phycocyanin Concentrations by Year. Averages were highest in 2015 and lowest in 2016.

<table>
<thead>
<tr>
<th>Year</th>
<th>Mean Chlorophyll-α (µg/L)</th>
<th>Mean Phycocyanin (µg/L)</th>
</tr>
</thead>
<tbody>
<tr>
<td>2015</td>
<td>67.81</td>
<td>53.92</td>
</tr>
<tr>
<td>2016</td>
<td>16.30</td>
<td>2.50</td>
</tr>
<tr>
<td>2017</td>
<td>27.16</td>
<td>21.11</td>
</tr>
</tbody>
</table>

Ultimately, during the past three blooms, both Chl-α and PC more often accumulated at the lake surface that at the bottom of the water column. Dissolved microcystin was not found to accumulate at a specific depth, and particulate microcystin was split between surface and bottom accumulation. In 2015, Lake Erie had the worst HAB in its documented history, and wind speeds did not consistently influence the surface-to-bottom ratios of Chl-α and PC (Figures 4.2 and 4.3). In 2016, the HAB was milder, and it took wind speeds of 5 m/s to mix the water enough to prevent the bloom from accumulating at the water surface. In 2017, it also took wind speeds of approximately 5 m/s to maintain Chl-α surface-to-bottom ratios of one.
Figure 4.2 Relationship of Wind Speed to Chlorophyll-\textit{a} Surface-to-Bottom Ratio, 2015-2017. In 2015, the wind speed did not consistently determine whether the chlorophyll-\textit{a} would be at the surface or bottom of the water column. In 2016, wind speeds above 5 m/s seemed to prevent the chlorophyll-\textit{a} from clustering at the surface. In 2017, wind speeds of approximately 5.5 m/s corresponded to a maintained surface-to-bottom ratio near one.

Figure 4.3 Relationship of Wind Speed to Phycocyanin Surface-to-Bottom Ratio, 2015-2017. In 2015, the wind speed did not consistently determine whether the phycocyanin would be at the surface or bottom of the water column. In both 2016 and 2017, wind speeds above 5 m/s seemed to be associated with preventing phycocyanin from clustering at the surface.
**bbe FluoroProbe 2016 Data Analysis**

Fluorometers measure the relative abundance of phytoplankton pigments by emitting various wavelengths of light and measuring any resulting fluorescence. Traditional fluorometers in a laboratory setting indicate the total amount of Chl-a and/or PC present in a sample after the pigments have been extracted from the cells by various methods, which is often a multi-day process. Alternatively, *in situ* fluorometers are designed to determine Chl-a, PC, and/or phytoplankton class abundance instantaneously. “*In situ*” indicates the fluorometers are used directly at the station of investigation (NOAA, 2016). Rather than collect water samples and take them back to a laboratory for pigment extraction, *in situ* fluorometers analyze the phytoplankton in the water immediately surrounding it.

The bbe FluoroProbe is an *in situ* fluorometer owned and used by CIGLR and NOAA GLERL to monitor water quality (Figure 4.4) (bbe moldaenke, 2017). It automatically determines the abundance of each phytoplankton class in the water source (bbe moldaenke, 2011). The fluorometer works by emitting various wavelengths of light and measuring any fluorescence that occurs in response. It has six optical sensors to collect profile data on concentrations of cyanobacteria, diatoms/dinoflagellates, cryptophytes, and chlorophytes. It has separate operation modes for field and laboratory settings. The field methods allow the FluoroProbe to collect data directly in a body of water in the natural environment. The laboratory method employs use of a stand, motorized stirrer, and a large cuvette to take measurements from a water sample brought back to the laboratory from the field.

*Figure 4.4 The bbe FluoroProbe III (bbe moldaenke, 2017). The FluoroProbe has six optical sensors to collect data on concentration of cyanobacteria, dinoflagellates, cryptophytes, and chlorophytes. It automatically corrects data for colored dissolved organic matter and turbidity.*
The goals of this project were to 1) investigate temporal and spatial patterns in the 2016 Lake Erie HAB, and 2) investigate how accurate the FluoroProbe field measurements were compared to both FluoroProbe cuvette and laboratory extraction methods.

**Methods**

During the bloom season of 2016, CIGLR/NOAA GLERL used the bbe FluoroProbe in the field during the weekly monitoring of WLE and also used the instrument in the laboratory to analyze surface water samples collected at the same time. I compiled spreadsheets calculating averages for data collected by the FluoroProbe by both field methods and laboratory cuvette methods. I calculated the average concentrations of chlorophytes, cyanobacteria, diatoms, cryptophytes, and total Chl-a. I also calculated the percentage of cyanobacteria contributing to the total Chl-a and the average concentration of yellow substances, which are dissolved organic matter that also fluoresce light but should not be mistaken for phytoplankton. I calculated averages for these variables for all WLE samples in 2016 and 2017. I then compared the FluoroProbe field data to the laboratory cuvette data to determine how well the results matched. I also compared the FluoroProbe field data to extracted Chl-a and PC concentrations determined by traditional laboratory analysis. The 2017 data was not included in this report because the data collection was still in progress when my fellowship ended.

The FluoroProbe field data was collected by lowering the FluoroProbe at a constant rate down through the water column until reaching the lake bottom. Measurements were recorded by the instrument approximately once per second. Surface water was defined as 0.5 - 1.5 m below the surface, and the water column was anything greater than 1.5 m below the surface. The FluoroProbe laboratory cuvette data was collected by placing the FluoroProbe in a large, 1 liter cuvette for a few minutes to determine algal concentrations. The FluoroProbe was set to collect as many readings as possible within the minute. The water used in the cuvette was collected at the same time that the FluoroProbe was collecting the field data. This same water sample also provided the extracted pigment data.
Results

Overall, average Chl-α concentration at each station reflected the water column depth and proximity to the Maumee River. Stations WE06 and WE09 had the highest average concentrations of Chl-α at approximately 18 – 19 µg/l (Figure 4.5). Both these stations had depths of less than 3 m and were closest to the mouth of the Maumee River, meaning they likely had the warmest water temperatures and highest concentrations of nutrients. Stations WE02, WE08, and WE12 had average Chl-α concentrations between 6 – 10 µg/l. These three stations had depths of 4 – 6 m and were slightly farther from the mouth of the river. Stations WE04, WE13, and WE15 had the lowest average Chl-α concentrations between 4 – 6 µg/l. Both stations WE04 and WE13 had water column depths of at least 8 m, and were in the central area of the western basin rather than being close to the mouth of the Maumee River. Station WE15 only had a depth of approximately 4 m, but was the easternmost of all stations, farthest away from the Maumee River.

Figure 4.5 Average Total Chlorophyll-α Concentration at each Western Lake Erie Monitoring Station in 2016. WE06 and WE09 had the highest concentrations near 18 µg/l, while WE04, WE13, and WE15 had the lowest at less than 5 µg/l.
All stations had their highest cyanobacteria concentration in August except for WE04, which had a slightly higher peak in September (Figure 4.6). This indicates that cyanobacteria dominated the phytoplankton community in August and September. Concentration peaks in late summer are expected since this is when the lake’s water temperature is warmest (Graham, et al., 2016). WE06 had the highest phytoplankton concentrations, reaching a maximum cyanobacterial concentration of 32 µg/l at the water surface on August 22, 2016. WE12 had the lowest concentrations for the summer with a maximum cyanobacterial concentration of 3.49 µg/l at the water surface on August 1, 2016.

The differences in concentration between the monitoring stations are likely due to both their proximity to rivers and their water column depth. The Maumee River provides most of the nutrient pollution in WLE, so one could assume that stations closer to the mouth of the river would have higher concentrations of those nutrients. In addition, shallower areas are warmer than deeper areas, and warmer water is more sustainable for cyanobacterial growth. Therefore, it
is understandable that monitoring stations in shallower water would have higher concentrations of cyanobacteria, as was the case for stations WE06 and WE09. These stations which typically see the highest concentrations of Chl-α and PC are closest to the mouth of the Maumee River and have depths of less than 3 m.

The FluoroProbe total Chl-α concentration determined in the field was consistent with the cuvette data. The relationship and similarities between the two methods were determined by calculating the linear equation and coefficient of determination for total Chl-α. Linear relationships were determined for each of the eighteen monitoring trips in 2016. Additionally, an overall equation and coefficient of determination was calculated for all data points from 2016 (Figure 4.7). The overall equation for all data from the 2016 season had a slope of 0.9233 and a y-intercept of 0.4513. A slope near 1 and a y-intercept near 0 indicate the two methods provide very similar data. The coefficient of determination was 0.895, meaning the regression accounted for 89.6% of the data’s variability. Ultimately, the field and laboratory operational settings for the bbe FluoroProbe provided consistent results.

![Graph showing linear relationship between FluoroProbe Cuvette and Field Measurements](image)

*Figure 4.7 Relationship of Total Surface Chlorophyll-α Concentrations Measured by the bbe FluoroProbe Field and Cuvette Methods. The slope is approximately 1, the y-intercept is 0.4513, and the coefficient of determination is 0.896. This indicates the two methods provide consistent results.*
Results from the surface water sample Chl-a extractions differed from the FluoroProbe data. When all points from the 2016 monitoring stations were plotted, the slope was 0.3922, the y-intercept was 1.7827, and the coefficient of determination was 0.8163 (Figure 4.8). This suggests that for every 1 µg/l of Chl-a detected by the laboratory extraction methods, the FluoroProbe detected approximately 0.4 µg/l of Chl-a, signifying that the FluoroProbe usually detected less than half of the Chl-a that was actually present. This could result in an understatement of the severity of phytoplankton growth in a water body if simply relying on in situ estimates.

Additionally, as the extracted Chl-a concentration increased, the data’s variability also increased. This is likely due to the colonial nature of the Lake Erie *Microcystis*. As the bloom intensifies, the *Microcystis* form large, flaky colonies. In these conditions, the bbe FluoroProbe could take a measurement of a large flake, or it could miss a flake entirely, resulting in highly variable data. Additionally, the bbe FluoroProbe is calibrated using laboratory-grown cultures which do not necessarily function or grow the same as the natural *Microcystis* colonies in Lake Erie.

![Figure 4.8 Relationship of Total Surface Chlorophyll-a Concentrations Measured by Laboratory Extraction and In Situ by the bbe FluoroProbe. The laboratory methods consistently identified more chlorophyll-a than the in situ method, as indicated by the low slope. As the extracted concentration increases, so does the data’s variability.](image-url)
**Data Project Summaries**

The two data projects I worked on gave some insight into the characteristics of the HABs and the accuracy of our methods. The surface-to-bottom ratio analysis of HAB data indicated that both PC and Chl-a had higher concentrations at the surface of the water column 70% of the time. Particulate microcystin distribution favored the surface and bottom of the water column an equal amount of time, and dissolved microcystin was distributed throughout the water column. When PC and Chl-a ratios were plotted against wind speed, it became apparent that biomass distribution is very likely dependent on wind speed. In 2015, wind speed did not appear to affect biomass distribution. In 2016 and 2017, wind speeds of 5 m/s or greater had biomass distributed throughout the water column rather than clustering at the surface. The HAB of 2015 was the most severe on record, so perhaps the intensity of the bloom is also a factor in the vertical distribution of the phytoplankton and toxins. These data will be used by NOAA to further inform the Experimental Lake Erie HAB Tracker forecasts on vertical distribution of cyanobacteria.

The bbe FluoroProbe 2016 data analysis provided insights into HAB characteristics and also revealed how similar or different results were for field, cuvette, and laboratory extraction methods. First, the data revealed which weekly monitoring stations were most impacted by the bloom and when each station had the highest concentration of phytoplankton. Ultimately, the stations closest to the mouth of the Maumee River, WE06 and WE09, had the highest Chl-a concentrations. Most stations had the highest cyanobacteria concentrations at the water surface during August, except WE04 which peaked in September. Second, the analysis suggested that the FluoroProbe data collected by the field methods accurately matched the data from the laboratory cuvette methods, indicating the FluoroProbe provides consistent results whether in the field or in the laboratory. However, the FluoroProbe’s *in situ* Chl-a measurements were consistently lower than traditional extraction methods. Moreover, as the Chl-a concentration increased, so did the FluoroProbe data’s variability. The difference in results for *in situ* and extraction measurements of Chl-a will be considered and investigated further by CIGLR and NOAA GLERL during future blooms.
Section 5: *In Situ* Fluorometer Evaluation Project

**Background and Objectives**

The second major project I worked on during my fellowship was part of the Alliance for Coastal Technologies (ACT) evaluation of *in situ* fluorometers. ACT is a NOAA and EPA funded component of the US Integrated Ocean Observing System that fosters partnerships between research institutions, private companies, and regional resource managers (Johengen, 2017). The goal of ACT is to provide third-party testing and validation of instruments and technologies in both laboratory and field settings. ACT focuses on sensor technologies that are used for monitoring coastal and freshwater environments. The sensor evaluations performed during the summer of 2017 focused on *in situ* fluorometers.

In June 2017, ACT developed protocols to evaluate multiple commercial *in situ* fluorometers on their ability to correctly and consistently identify the presence and abundance of phytoplankton classes within a natural environment. The evaluation parameters included accuracy, precision, range, response linearity, deployment length, and reliability in both field applications and against laboratory standards (see Appendix B for more details on each parameter) (Johengen, 2017). If a fluorometer receives a positive evaluation, this means ACT determined that the device is able to collect accurate and reliable data. This would greatly decrease the need for laboratories to use extraction methods to determine the true concentration of phytoplankton pigments. The current extraction methods have a time lag of multiple days between sample collection and release of data. If a fluorometer could accurately determine the same information instantaneously, it could provide immediate or continuous data for use by the scientific community and the public alike.

It is important to note that the fluorometers were not compared to each other but instead were evaluated on an individual basis. Throughout the summer of 2017, I worked with a team of ACT personnel to collect data for the fluorometer performance evaluation in freshwater environments. Additional data were collected by ACT personnel in estuarine and marine environments. This report focuses solely on the freshwater data which were collected through three tests:

1) Field deployment at a moored (stationary) location on the Maumee River
2) Laboratory experiments at GLERL
3) Field deployment using surface mapping on Lake Erie
The full evaluation is scheduled to be completed by April 2018, with most of the data analysis being performed in November 2017. Due to proprietary reasons, data from the four in situ fluorometers being evaluated cannot be presented in this report. The analysis presented only includes data collected by NOAA GLERL or CIGLR owned instruments including desktop fluorometers in the laboratory and the YSI EXO2 Multiparameter Sonde used during the field deployments.

**YSI EXO2 Multiparameter Sonde**

NOAA GLERL owns a YSI EXO2 Multiparameter Sonde (EXO2) that collects real-time data on temperature, dissolved oxygen, Chl-\(\alpha\), PC, and more. The EXO2 has seven sensor ports, one of which is occupied by a central wiper (Figure 5.1) (YSI Inc., 2017a). The wiper prevents sensors from becoming covered with sediment, microbial growth, or other substances that could accumulate during a long-term deployment and prevent them from recording accurate measurements. The other six sensor ports can be filled with any of the thirteen sensor options, allowing up to 23 parameters to be measured (see Appendix C for details on the parameters that can be measured).

The EXO2 owned by NOAA GLERL measures depth, temperature, conductivity, pH, turbidity, dissolved oxygen, Chl-\(\alpha\), PC, and fluorescent dissolved organic matter (fDOM) (see textbox). It logs all data internally and can reach depths of 250 m. It requires four alkaline batteries and can operate at temperatures from -5 to 50 °C. The accuracy of EXO2 Chl-\(\alpha\) measurements are sometimes diminished by the

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**Conductivity** — ability of the water to conduct an electrical current (YSI Inc., 2017b)

**Turbidity** — clarity of water; as clarity decreases, turbidity increases (YSI Inc., 2017c)

**Colored Dissolved Organic Matter (CDOM)** — organic matter dissolved in water that absorbs ultra-violet light (YSI Inc., 2017d)

**Fluorescent Dissolved Organic Matter (fDOM)** — fraction of CDOM that can fluoresce (YSI Inc., 2017d)
presence of colored dissolved organic matter (CDOM) and high turbidity. Both CDOM and turbidity can cause a false positive reading of Chl-α (Turner Designs, 2017).

Methods

Moored Deployment

During the moored deployment with CIGLR, the in situ fluorometers were secured in a flow-through tank at the Bowling Green Water Treatment Plant off the Maumee River in Waterville, Ohio (Figure 5.2). The flow-through tank used was constructed by the United States Geological Survey in 2016 for a different ACT project studying nutrient sensors. The deployment for the current ACT project occurred over 14 consecutive days from July 25 – August 7, 2017 when the water was expected to have variable concentrations of cyanobacteria and high concentrations of Chl-α (100 µg/L) and turbidity (100 NTU) (Johengen, 2017). I helped set up and clean the tank as well as collect, process, and analyze the reference samples.

The day prior to deployment, each in situ fluorometer was calibrated by our team according to manufacturer manuals (Johengen, 2017). They were then programmed to record data every 30 minutes for the duration of the deployment. The internal clocks were checked against www.time.gov and then placed in baths of deionized water and two concentrations of Basic Blue 3 (BB3). The water bath allowed

Figure 5.2 Flow-Through Tank Setup for the Moored Deployment. The black tube in the rear of the tank is where the water comes in from the Maumee River. The PVC tubes in the front are where the water leaves the tank from openings at the top and the bottom. The metal frame above the tank is where the motor, EXO2, PAR sensor, and three in situ fluorometers were hung. Photo by Deanna Fyffe.
the fluorometers to take a blank reading of ultrapure water, and the BB3 ensured the fluorescence measurements were calibrated properly prior to deployment.

On the day of deployment, all fluorometers were arranged next to each other in a deployment rack so that they were all at the same depth and all received uniform light intensity (Figure 5.3). A space of one fluorometer diameter was left between each fluorometer to minimize interference in readings. The fluorometers were deployed 20 cm above the bottom of the flow-through tank, which was 1 m deep. In addition to the in situ fluorometers being tested, the EXO2 was deployed to independently measure temperature, conductivity, CDOM, turbidity, Chl-a, and PC every 15 minutes. A LI-COR LI-193 Underwater Spherical PAR Sensor was also deployed to determine water clarity and light intensity. A motorized fan was situated beneath the fluorometers to ensure ample mixing of the water within the flow-through tank prior to reference sample collection.

![Figure 5.3 Topical Diagram of the Moored Deployment Flow-Through Tank. The three in situ fluorometers are represented by dark grey boxes. The EXO2 was situated in the top right corner, the motor in the middle, and the PAR sensor in the bottom left corner. Diagram by Deanna Fyffe.](image)

During the deployment, at least one ACT team member traveled to the Bowling Green Water Treatment Plant every weekday to collect two to four reference samples. I traveled with at least one other team member for all but two days of the deployment to help with reference sample collection and analysis.

Ten minutes prior to reference sample collection, we would isolate the tank by closing the inflow and outflow valves and turning on the motor. This ensured the water was homogeneous, and the reference samples were representative of the water the fluorometers were measuring. At the same time the fluorometers took a reading, we collected reference water samples in two 2-liter Van
Dorn bottles. Reference samples were collected at least twice each weekday approximately an hour apart (Johengen, 2017). These reference samples were processed on site and analyzed back at GLERL or external laboratories for Chl-\(\alpha\), PC, turbidity, CDOM, and phytoplankton taxonomic groupings and abundances (see Appendix D.1 through D.4 for details on methods used in processing these samples).

Measures were taken for both quality assurance and quality control for laboratory analysis, including having each entity collect field trip blanks and field duplicates (see Appendix D.5 and D.6 for details on the quality assurance and quality control measures respectively).

**Laboratory Experiments**

The laboratory experiments occurred at GLERL on August 10, 2017. The purpose of the experiments was to see how well the fluorometers detected different phytoplankton of known concentrations. The experiment used one cyanobacteria genera (*Aphanizomenon* sp.), one dinoflagellate (*Peridinium inconspicuum*), one cryptophyte (*Cryptomonas* sp.), and one chlorophyte (*Chlorella* sp.) (Figure 5.4). All cultures were grown and maintained by NOAA GLERL researcher Dave Fanslow. They were grown in WC media, commonly used for freshwater cyanobacteria growth (Kurmayer, et al., 2017).

![Figure 5.4 Phytoplankton Cultures for the Laboratory Experiment. The cultures were grown and maintained by Dave Fanslow of NOAA GLERL. The cultures from left to right are *Chlorella* sp., *Cryptomonas* sp., *Aphanizomenon* sp., and *Peridinium inconspicuum*. Photo by Deanna Fyffe.](image-url)
The experiment had nine trials, each with a different combination of phytoplankton or water quality parameters (see Appendix E for a tabular breakdown of the trials). The fluorometers sat in each solution for 5 minutes taking readings every 10 seconds. The first four trials consisted of the individual phytoplankton cultures to see if the fluorometers could correctly identify the phytoplankton class and abundance with no other species possibly interfering. The remaining five trials contained different mixtures of the four individual phytoplankton cultures. Trial five had concentrations of 10 µg/L *Chlorella* sp. and 2 µg/L each of the other three phytoplankton. Trial six had 10 µg/L *Chlorella* sp., 10 µg/L *Cryptomonas* sp., and 2 µg/L each of *Aphanizomenon* sp. and *Peridinium inconspicuum*. This continued until all four phytoplankton were at 10 µg/L. Trial nine consisted of the mixture from the eighth trial and added CDOM and turbidity, each at a concentration of 30 µg/L. After all the fluorometers recorded measurements from each trial, reference samples were collected and analyzed for the same parameters as explained in the moored deployment.

Ultimately, this experiment allowed our team to assess accuracy and precision the fluorometers by comparing their measurements to known phytoplankton concentrations. By testing each phytoplankton class individually, we first determined if the fluorometers could correctly identify the class without any other potential interference. Then, we combined all four cultures at various concentrations to observe how the fluorometers performed when there was a mixture of phytoplankton classes. Lastly, the addition of CDOM and turbidity tested the ability of the fluorometers to correct for these interfering characteristics and still give accurate results.

**Surface Mapping Deployment**

Part of the ACT evaluation included surface mapping, which took place on a boat in WLE on August 16, 2017. The fluorometers were secured in a flow through tank which took in water from just below the lake surface. The boat cruised to various locations while the fluorometers continually collected data. By having the fluorometers continually collect data during the entire excursion, the HAB conditions across the surface of the lake could be mapped out. Reference samples were collected periodically at specific locations and were analyzed for the same characteristics as the previous two tests: Chl-α, PC, turbidity, CDOM, and phytoplankton taxonomic groupings and abundances.
The day before deployment, each fluorometer was programmed to collect data as quickly as possible, about once every 10 seconds (Johengen, 2017). The internal clocks were checked against www.time.gov and then the fluorometers were placed in baths of deionized water and two concentrations of BB3, just as was done for the moored deployment. The day of the deployment, the fluorometers were placed in a flow-through tank constructed by CIGLR engineer Russ Miller which had a water exchange rate of approximately 5 minutes. The EXO2 was also deployed within the tank to independently measure temperature, conductivity, CDOM, turbidity, Chl-a, and PC every minute. One of the team members was constantly churning the water in the flow through tank to ensure it was homogeneous.

During the deployment, the boat cruised to seven of the eight NOAA monitoring stations in WLE (Figure 4.1). Station WLE15 was left out since it was a significant distance from all other stations and was far from the HAB, which extended along the westernmost coast and northeast toward Canada (Figure 5.5) (NOAA, 2017g). The boat cruised into and out of the HAB multiple times so that the fluorometers could operate in a wide range of phytoplankton taxonomy and concentrations (Johengen, 2017). When the boat reached a monitoring station, the flow-through tank was isolated and thoroughly mixed for 20 minutes. Reference samples were collected 5 and 20 minutes into each isolation period.

Figure 5.5 Extent of the Harmful Algal Bloom on August 16, 2017 (NOAA, 2017g). The day of the surface mapping deployment, the bloom mostly manifested along the westernmost coast but also extended east and north near stations WE08 and WE04. The image was produced from Copernicus Sentinel 3 data. The grey areas indicate missing data as result of cloud coverage. Image courtesy of the National Oceanic and Atmospheric Administration and the National Aeronautics and Space Administration.
Reference Sample Analysis

Pigments

Pigment concentrations, such as Chl-a and PC, represent phytoplankton and cyanobacteria abundance respectively. All reference samples were analyzed for both of these pigments. I filtered majority of the reference samples and performed all extractions for both PC and Chl-a.

The entire process to analyze pigments was performed in minimal light because ultraviolet rays can break down pigments once they are extracted from the cells (Yentsch & Menzel, 1963). Each reference sample provided six filter replicates: two for Chl-a, three for PC, and one reserve. The filters used were 25 mm Whatman GF/F filters and the vacuum was kept low (<5 in Hg) during filtration. The volumes filtered varied depending on coloration of the filter. All six filters were placed in 15 ml opaque, poly-carbonate centrifuge tubes and stored in a cryofreezer at -80°C until the date of analysis (see Appendix D.1 for more detailed methods).

On the day of Chl-a analysis, two tubes were removed and allowed to thaw to room temperature. Once thawed, dimethylformamide (DMF) was added and the tubes were placed in a water bath for 25 minutes. The samples were read in a Turner Designs 10 AU fluorometer, which was calibrated using standard Chl-a concentrations and a blank of DMF (see Appendix D.2 for a more detailed procedure for Chl-a analysis).

For PC analysis, a phosphate buffer was added to the PC tubes before they went through a 3-day freeze-thaw process which included a shaker table, two sonications, and centrifugation. When ready, the PC tubes were analyzed using a Turner Designs AquaFluor fluorometer calibrated with authentic PC standards (see Appendix D.3 for a more detailed procedure for PC analysis).

Turbidity

The turbidity of each reference sample was determined by a Hach 2100AN Turbidimeter at GLERL. The turbidimeter was calibrated with certified turbidimetric standards prior to reference sample readings. The turbidity readings were performed the day of reference sample collection immediately upon return to the laboratory.
Colored Dissolved Organic Matter
All CDOM samples were collected by filtering 40-45 mL reference sample into a 50 mL BD Falcon Tube. The filters used were 25 mm Millex syringe filters with a pore size of 0.2 µm. The samples were stored in a freezer until shipment to Moss Landing Marine Laboratory where the analysis was performed (see Appendix D.4 for a more detailed procedure on CDOM analysis).

Phytoplankton
To analyze phytoplankton community composition, 500 mL of each reference sample were set aside and preserved with acidic Lugol’s. The phytoplankton analyses for this project were performed by contracted laboratories that had previously worked with ACT and had undergone technical audits by ACT’s Quality Assurance Manager. For the WLE samples, Euan D. Reavie of the National Resources Research Institute performed the analysis, following protocols outlined by the United States Environmental Protection Agency’s Great Lakes National Program Office Biological Surveillance Program.

Example Evaluation - Field Deployment Chlorophyll-α Measurements
Since the fluorometer evaluation is not being released until April 2018, data from the in situ fluorometers could not be used in this report. Therefore, the only data presented here were collected by CIGLR and NOAA GLERL owned instruments that are not being evaluated in the ACT project. The EXO2 was deployed alongside the in situ fluorometers in both the Maumee River and Lake Erie in order to provide ancillary data such as temperature, turbidity, and Chl-α concentration. It was not used during the laboratory experiments.

This example evaluation using the EXO2 demonstrates how the individual fluorometers will be evaluated for the field deployments. In the actual in situ fluorometer evaluations, there will be additional assessments on precision, range, response linearity, deployment length, and reliability. Based on my access to data and the fact that majority of the data analysis was performed after my fellowship ended, I used the protocols as a guide to assess the EXO2 results from the moored and surface mapping deployments. The full evaluation will include additional parameters, but only Chl-α, data were available at the time of this report. Additionally, the full evaluation will include results from the laboratory experiments, but the EXO2 was not used for this test so it could not be demonstrated in this report.
Moored Deployment

Generally, the EXO2 Chl-α measurements and the reference sample Chl-α measurements followed the same trend throughout the moored deployment with the EXO2 usually reporting lower concentrations (Figure 5.6) (see Appendix F for the moored deployment data). During the final day of the deployment, the EXO2 reported more than triple the amount of Chl-α identified through laboratory extraction. At this point, the water was a dark brown color, suggesting the CDOM was very high (Fondriest Environmental, Inc., 2014). Since the EXO2 does not have a system in place to correct for fluorescence by CDOM, it is possible that the fluorescence from CDOM was recorded as Chl-α. However, this is only speculation since the CDOM sample analysis was not complete at the time this report was written.

Figure 5.6 EXO2 and Reference Sample Chlorophyll-α Measurements throughout the Moored Deployment. Top: EXO2 measurements (blue) and reference sample measurements (orange) throughout the deployment. The first two reference samples were collected on July 25th at 1 pm and 3 pm, but the EXO2 failed to begin recording until 7:30 p.m. on July 25th. Bottom: the absolute difference between EXO2 and reference sample chlorophyll-α measurements throughout the duration of the deployment.
The linear regression of reference Chl-a concentration versus EXO2 Chl-a concentration during the moored deployment had a slope of approximately 0.3 and a y-intercept of 13.5 (Figure 5.7). The coefficient of determination was 0.271, indicating the equation accounted for 27% of the data’s variability. The linear regression was significant with a p-value of 0.013. This indicates that there is evidence of a linear relationship between reference sample and EXO2 measured Chl-a from the moored deployment.

Figure 5.7 Linear Regression of Reference Chlorophyll-a versus EXO2 Chlorophyll-a for the Moored Deployment. The equation has a slope of 0.3 and a y-intercept of 13.5. The coefficient of determination is 0.271 and the p-value is 0.013.

**Surface Mapping Deployment**

The EXO2 provided very different results during the surface mapping deployment. During the moored deployment, the mean difference between the EXO2 and laboratory measurements (delta) was approximately 35 µg/L. During the surface mapping deployment, Chl-a existed at much higher concentrations, and delta increased to a mean of 139 µg/L. The largest deltas occurred at station WE06, where the Chl-a concentration reached 805 µg/L (Figure 5.8) (see Appendix G for the surface mapping deployment data). This is the location where the bloom was most severe and there was significantly more Chl-a than any other station. Although the values were incorrect, the EXO2 measurements did follow the same trend as the reference sample measurements throughout the deployment.
Figure 5.8 EXO2 and Reference Sample Chlorophyll-a Measurements throughout the Surface Mapping Deployment. Top: EXO2 measurements (blue) and reference sample measurements (orange) throughout the time of the deployment. The EXO2 values are plotted by the primary axis on the left, and the laboratory values are plotted on the secondary axis on the right. Bottom: the absolute difference between EXO2 and reference sample measurements throughout the duration of the deployment.
The linear regression of reference Chl-a versus EXO2 Chl-a had a slope of 0.04 and a y-intercept of 2.4 (Figure 5.9). The figure does not include the two extreme data points obtained at station WE06. The coefficient of determination was 0.3266, indicating the equation only accounted for 33% of the data’s variability. Moreover, the linear regression had a p-value of 0.052, meaning it was not significant. This indicates there is not a strong linear relationship between reference sample and EXO2 measured Chl-a during the surface mapping deployment.

Figure 5.9 Linear Regression of Reference Chlorophyll-a versus EXO2 Chlorophyll-a for the Surface Mapping Deployment. The equation has a slope of 0.04 and a y-intercept of 2.4. The coefficient of determination is 0.905 and the p-value is 0.052.

Summary

The in situ fluorometer evaluation project conducted by ACT aimed to determine how well four commercially available fluorometers could correctly and consistently identify the presence and abundance of phytoplankton classes within a natural environment. To do this, ACT coordinated with laboratories on the east coast, west coast, and Great Lakes to conduct testing. CIGLR and NOAA GLERL performed fluorometer testing in freshwater environments through three major tests: a moored deployment, laboratory experiments, and a surface mapping deployment. The data from the four commercial fluorometers could not be used in this report due to proprietary reasons, so data from the EXO2, a CIGLR/NOAA GLERL owned instrument, was used instead to demonstrate how the fluorometers will be evaluated for the field deployments. With the exception of two reference samples, the EXO2 consistently measured less Chl-a than the traditional extraction method used on the reference samples. This could be due to a calibration error, because the EXO2 correctly identified trends in the changing Chl-a concentration.
Section 6: Current Status of Lake Erie

HABs continue to be a global problem, affecting waters in Vietnam, Chile, Portugal, and larger areas such as the Gulf of Guinea and the Southwest Pacific (ISSHA, 2017c). In the United States alone, HABs have been anecdotally reported in 43 states (Graham, Dubrovsky, & Eberts, 2016). Some states, such as Ohio, experience annual HABs that are closely monitored by NOAA (NOAA, 2017h). In Lake Erie, the 2017 HAB proved to be much more intense than the previous year. Although 2015 is still the worst HAB on record for Lake Erie, the bloom of 2017 provided major concerns. The Maumee Bay State Park in Oregon, Ohio had four recreational advisories due to the HAB or algal toxins throughout the summer of 2017, including an elevated advisory that lasted for 24 days (ODH, 2017). Additionally, the HAB grew in the Maumee River in September due to low flow, high temperatures, and high nutrients (Figure 6.1) (NOAA GLERL, 2017).

Figure 6.1 The Maumee River Harmful Algal Bloom on September 22, 2017 (NOAA GLERL, 2017). Photo credit: Zachary Haslick, Aerial Associates Photography Inc.

Mayor Hicks-Hudson of Toledo joined many concerned citizens in appealing to the federal government to declare Lake Erie as impaired (Patel & Parshina-Kottas, 2017). If determined at the federal level to be impaired, the nutrient loadings into Lake Erie can be regulated under the Clean Water Act, which requires a plan to reduce pollution sources in all impaired waters (US EPA, 2017c). However, deterring nutrient pollution can be very difficult since nonpoint sources...
are often main contributors (US EPA, 2017d). Also, although this could help decrease the HABs intensity, nutrient loading is not the sole cause of the blooms.

This summer demonstrated to me that HABs are much more complex than conveyed to the public. There is no single solution or preventative measure that can take place to eliminate them, and any measures we do take to improve the water quality of Lake Erie will likely take many years to have an observable impact. It will take significant research and stakeholder cooperation to address the Lake Erie HABs.

The research conducted by CIGLR and NOAA GLERL this summer will continue being applied to future HABs. The weekly sample surveys are providing long-term data that demonstrate how the ecosystem in WLE has changed throughout the past decade. Within the next few years, their newly developed environmental sample processor will be implemented on a larger scale within the lake, allowing continuous data collection on algal toxins in multiple locations. As the fluorometers evaluated this summer continue to be adjusted and improved, continuous data collection will become increasingly accurate. These possibilities of accurate real-time data could allow government and research organizations around the world to have easier and faster access to data regarding HABs. This could greatly improve monitoring efforts and help the scientific community better understand HABs, their driving forces, and their effects.

Additionally, many organizations and institutions are already working together to improve Lake Erie’s water quality conditions through preventative measures. The Ohio EPA has published its action plan to reduce phosphorous loading into Lake Erie, and Michigan and Ontario will have action plans completed by 2018 (Ohio EPA, 2017). The Ohio plan introduced specific actions to be taken by individual stakeholders including federal and state agencies as well as universities. The Ohio EPA used input from both agricultural and environmental associations to develop the action plan. Problems like HABs have many branches of stakeholders that often have competing interests and priorities, so it is promising that the Ohio stakeholders are working together to determine what can be done to improve the conditions of Lake Erie.
Works Cited


National Oceanic and Atmospheric Administration Great Lakes Environmental Research


Appendices

Appendix A: List of CIGLR Consortium Categories and Members (CIGLR, 2017)

1. University Partners
   a. Central Michigan University
   b. Cornell University
   c. Grand Valley State University
   d. University of Michigan
   e. Michigan State University
   f. University of Minnesota-Duluth
   g. The Ohio State University
   h. University of Windsor
   i. University of Wisconsin-Milwaukee

2. University Affiliates
   a. Bowling Green State University
   b. Eastern Michigan University
   c. Purdue University
   d. State University of New York – ESF 18 Member Consortium
   e. University of Minnesota
   f. University of Toledo
   g. University of Wisconsin-Madison
   h. Wayne State University

3. Private Sector Partners
   a. Fondriest Environmental
   b. Great Lakes Environmental Center
   c. LimnoTech
   d. The Nature Conservancy, Great Lakes
   e. National Wildlife Federation, Great Lakes Regional Center

4. Supporting Initiatives and NOAA Programs
   a. Council of Great Lakes Industries
   b. Great Lakes Commission
   c. Great Lakes Observing System
   d. USGS Great Lakes Science Center
   e. International Joint Commission
   f. Lake Superior National Estuarine Research Reserve
   g. Old Woman Creek National Estuarine Research Reserve
   h. Thunder Bay National Marine Sanctuary
   i. Wisconsin-Lake Michigan National Marine Sanctuary
Appendix B: ACT Parameter Definitions and Details (Johengen, 2017)

Table 0.1. Alliance of Coastal Technologies’ Evaluation Parameters for In Situ Fluorometers. ACT is evaluating fluorometers based on accuracy, precision, range, response linearity, deployment length, and reliability (Johengen, 2017).

<table>
<thead>
<tr>
<th>Parameter</th>
<th>Definition</th>
<th>Details</th>
<th>Location</th>
</tr>
</thead>
<tbody>
<tr>
<td>Accuracy</td>
<td>Closeness of agreement between the result of a measurement and reference,</td>
<td>Repeated comparisons between instrument measurements and reference water samples. Reported as the absolute and relative difference between reference and measured values</td>
<td>Laboratory</td>
</tr>
<tr>
<td></td>
<td>targets (dye proxies, live cells)</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Precision</td>
<td>A measure of the repeatability of a measurement obtained under stipulated</td>
<td>Calculating the coefficient of variation (100 x Standard Deviation/Mean) of 5 replicate fluorometer measurements at 3 different reference cell concentrations</td>
<td>Laboratory</td>
</tr>
<tr>
<td></td>
<td>controlled conditions</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Range</td>
<td>Upper and lower limits of detection and quantification</td>
<td>Measurement of the minimum and maximum concentration of specific phytoplankton taxa that the instrument can distinguish from background and previous reference levels, respectively</td>
<td>Laboratory</td>
</tr>
<tr>
<td>Response</td>
<td>Stability of a predetermined response or calibration factor, computed as:</td>
<td>Pigment concentration and cell counts of known additions of cultured algae into filtered natural water. The influence of turbidity (light scattering particles) and colored dissolved organic matter on fluorometric response will also be assessed through addition experiments</td>
<td>Laboratory</td>
</tr>
<tr>
<td>Linearity</td>
<td>(sample solution measurement – blank solution measurement) / [reference</td>
<td></td>
<td></td>
</tr>
<tr>
<td></td>
<td>standard] over a range of reference standard concentrations</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Deployment</td>
<td>Amount of time the instrument can operate in a submerged deployment setting</td>
<td>Successful deployment requires the sensor to perform within the targeted ranges of accuracy throughout the expected deployment application and duration. Also, comparisons will be made of the percent of data recovered as a proportion of the data that an instrument was designed to collect during its deployment period</td>
<td>Field</td>
</tr>
<tr>
<td>length</td>
<td>at a depth of one meter below the surface without needed maintenance or</td>
<td></td>
<td></td>
</tr>
<tr>
<td></td>
<td>recalibration</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Reliability</td>
<td>The ability to maintain integrity or stability of the instrument and data</td>
<td>Pre- and post-measures of blanks and reference dyes to quantify drift during deployment periods. Comments on the physical condition of the instruments (e.g., physical damage, flooding, corrosion, battery failure, etc.) will also be recorded.</td>
<td>Laboratory,</td>
</tr>
<tr>
<td></td>
<td>collections over time</td>
<td></td>
<td>Field</td>
</tr>
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</table>
## Appendix C: YSI EXO2 Multiparameter Sonde Sensor Information (YSI Inc., 2017)

<table>
<thead>
<tr>
<th>EXO Parameter Measured</th>
<th>Sensor</th>
<th>Range</th>
<th>Accuracy</th>
<th>Response</th>
<th>Resolution</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Ammonium</strong> <em>(freshwater only)</em></td>
<td>Ammonium Sensor&lt;br&gt; Ammonia with pH sensor</td>
<td>0 to 200 mg/L (0 to 30˚C)</td>
<td>±10% of reading or 2 mg/L-N, w.i.g.</td>
<td>T63&lt;30 sec</td>
<td>0.01 mg/L</td>
</tr>
<tr>
<td><strong>Barometric Pressure</strong></td>
<td>Integral Barometer</td>
<td>375 to 825 mmHg</td>
<td>±1.5 mmHg from 0 to 50˚C</td>
<td>-</td>
<td>0.1 mmHg</td>
</tr>
<tr>
<td><strong>Blue-Green Algae, Phycocyanin</strong></td>
<td>Total Algae Sensor</td>
<td>0 to 280 μg/L; 0 to 100 RFU</td>
<td><strong>Linearity:</strong> R² &gt; 0.999 for serial dilution of Rhodamine WT solution from 0 to 100 μg/mL BGA-PC equivalents</td>
<td>T63&lt;2 sec</td>
<td>0.01 μg/L; 0.01 RFU</td>
</tr>
<tr>
<td><strong>Blue-Green Algae, Phycoerythrin</strong></td>
<td>Total Algae Sensor</td>
<td>0 to 280 μg/L; 0 to 100 RFU</td>
<td><strong>Linearity:</strong> R² &gt; 0.999 for serial dilution of Rhodamine WT solution from 0 to 280 μg/mL BGA-PE equivalents</td>
<td>T63&lt;2 sec</td>
<td>0.01 μg/L; 0.01 RFU</td>
</tr>
<tr>
<td><strong>Chloride</strong> <em>(freshwater only)</em></td>
<td>Chloride Sensor</td>
<td>0 to 18000 mg/L (0 to 30˚C)</td>
<td>±15% of reading or 5 mg/L-Cl, w.i.g.</td>
<td>T63&lt;30 sec</td>
<td>0.01 mg/L</td>
</tr>
<tr>
<td><strong>Chlorophyll</strong></td>
<td>Total Algae Sensor</td>
<td>0 to 400 μg/L; 0 to 100 RFU</td>
<td><strong>Linearity:</strong> R² &gt; 0.999 for serial dilution of Rhodamine WT solution from 0 to 400 μg/L Chl a equivalents</td>
<td>T63&lt;2 sec</td>
<td>0.01 μg/L Chl; 0.01 RFU</td>
</tr>
<tr>
<td><strong>Conductivity</strong></td>
<td>Conductivity / Temperature Sensor</td>
<td>0 to 200 mS/cm</td>
<td><strong>0 to 100:</strong> ±0.5% of reading or 0.001 mS/cm, w.i.g.; <strong>100 to 200:</strong> ±1% of reading</td>
<td>T63&lt;2 sec</td>
<td>0.0001 to 0.01 mS/cm (range dependent)</td>
</tr>
<tr>
<td><strong>Depth - 10 m</strong></td>
<td>Integral, Non-vented Depth Sensor</td>
<td>0 to 10 m (0 to 33 ft)</td>
<td>±0.04% FS (±0.004 m or ±0.013 ft)</td>
<td>T63&lt;2 sec</td>
<td>0.001 m (0.001 ft) (auto-ranging)</td>
</tr>
<tr>
<td><strong>Depth - 100 m</strong></td>
<td>Integral, Non-vented Depth Sensor</td>
<td>0 to 100 m (0 to 328 ft)</td>
<td>±0.04% FS (±0.04 m or ±0.13 ft)</td>
<td>T63&lt;2 sec</td>
<td>0.001 m (0.001 ft) (auto-ranging)</td>
</tr>
<tr>
<td><strong>Depth - 250 m</strong></td>
<td>Integral, Non-vented Depth Sensor</td>
<td>0 to 250 m (0 to 820 ft)</td>
<td>±0.04% FS (±0.10 m or ±0.33 ft)</td>
<td>T63&lt;2 sec</td>
<td>0.001 m (0.001 ft) (auto-ranging)</td>
</tr>
<tr>
<td><strong>Dissolved Oxygen, % air saturation</strong></td>
<td>Optical Dissolved Oxygen Sensor</td>
<td>0 to 500% air saturation</td>
<td><strong>0 to 200%:</strong> ±1% of reading or 1% saturation, w.i.g.; <strong>200 to 500%:</strong> ±5% of reading</td>
<td>T63&lt;5 sec</td>
<td>0.1% air saturation</td>
</tr>
<tr>
<td>Measurement</td>
<td>Type</td>
<td>Range</td>
<td>Accuracy and Response Time</td>
<td>Detection Limit</td>
<td></td>
</tr>
<tr>
<td>------------------------------</td>
<td>--------------------------</td>
<td>-------------------</td>
<td>-----------------------------</td>
<td>-----------------</td>
<td></td>
</tr>
<tr>
<td>Dissolved Oxygen, mg/L</td>
<td>Optical Dissolved Oxygen Sensor</td>
<td>0 to 50 mg/L</td>
<td>0 to 20 mg/L: ±0.1 mg/L or 1% of reading, w.i.g.; 20 to 50 mg/L: ±5% of reading</td>
<td>T63&lt;5 sec 0.01 mg/L</td>
<td></td>
</tr>
<tr>
<td>fDOM (CDOM)</td>
<td>fDOM Sensor</td>
<td>0 to 300 ppb Quinine Sulfate equivalents (QSU)</td>
<td>Linearity: R2 &gt; 0.999 for serial dilution of 300 ppb QS solution Detection Limit: 0.07 ppb QSU</td>
<td>T63&lt;2 sec 0.01 ppb QSU</td>
<td></td>
</tr>
<tr>
<td>Level, Vented - 10 m</td>
<td>Integral Vented Level Sensor</td>
<td>0 to 10 m (0 to 33 ft)</td>
<td>±0.03% FS (±0.003 m or ±0.010 ft)</td>
<td>T63&lt;2 sec 0.001 m (0.001 ft)</td>
<td></td>
</tr>
<tr>
<td>Nitrate (freshwater only)</td>
<td>Nitrate Sensor</td>
<td>0 to 200 mg/L-N (0 to 30˚C)</td>
<td>±10% of reading or 2 mg/L-N, w.i.g.</td>
<td>T63&lt;30 sec 0.01 mg/L</td>
<td></td>
</tr>
<tr>
<td>pH</td>
<td>pH Sensor</td>
<td>0 to 14 units</td>
<td>±0.1 pH units within ±10˚C of calibration temp; ±0.2 pH units for entire temp range</td>
<td>T63&lt;3 sec 0.01 units</td>
<td></td>
</tr>
<tr>
<td>ORP</td>
<td>pH/ORP Sensor</td>
<td>-999 to 999 mV</td>
<td>±20 mV in Redox standard solution</td>
<td>T63&lt;5 sec 0.1 mV</td>
<td></td>
</tr>
<tr>
<td>Temperature</td>
<td>Conductivity / Temperature Sensor</td>
<td>-5 to 35˚C 35 to 50˚C</td>
<td>±0.01˚C 0.05˚C</td>
<td>T63&lt;1 sec 0.001 °C</td>
<td></td>
</tr>
<tr>
<td>Turbidity</td>
<td>Turbidity Sensor</td>
<td>0 to 4000 FNU</td>
<td>0 to 999 FNU: 0.3 FNU or ±2% of reading, w.i.g.; 1000 to 4000 FNU: ±5% of reading</td>
<td>T63&lt;2 sec 0 to 999 FNU = 0.01 FNU; 1000 to 4000 FNU = 0.1 FNU</td>
<td></td>
</tr>
<tr>
<td>Salinity</td>
<td>Calculated from Conductivity and Temperature</td>
<td>0 to 70 ppt</td>
<td>±1.0% of reading or 0.1 ppt, w.i.g.</td>
<td>T63&lt;2 sec 0.01 ppt</td>
<td></td>
</tr>
<tr>
<td>Specific Conductance</td>
<td>Calculated from Conductivity and Temperature</td>
<td>0 to 200 mS/cm</td>
<td>±0.5% of reading or 0.001 mS/cm, w.i.g.</td>
<td>- 0.001, 0.01, 0.1 mS/cm (auto-scaling)</td>
<td></td>
</tr>
<tr>
<td>Total Dissolved Solids (TDS)</td>
<td>Calculated from Conductivity and Temperature</td>
<td>0 to 100,000 mg/L Cal constant range 0.30 to 1.00 (0.64 default)</td>
<td>Not Specified</td>
<td>- Variable</td>
<td></td>
</tr>
<tr>
<td>Total Suspended Solids (TSS)</td>
<td>Calculated from Turbidity and user reference samples</td>
<td>0 to 1500 mg/L</td>
<td>Not specified</td>
<td>T63&lt;2 sec Variable</td>
<td></td>
</tr>
</tbody>
</table>
Appendix D: Detailed Methods

D.1 Water Sample Filtering Protocol (NOAA, 2010)

a. Scope and Application - This method provides a procedure for filtering water from the western basin of Lake Erie for quantification of nutrients, algae, and related parameters in the western basin of Lake Erie.

b. Summary of Method - Filtering is completed as soon samples are returned from the field. Filtering and storage methods are unique to each parameter and must not be confused. All filtering must be done at < 200 mm Hg.

c. Sample Handling - Samples should return from the field in coolers with ice packs as needed. Filtering should be done immediately or samples must be stored in incubator. Gloves should be worn at all times and special care should be given to avoiding contamination.

d. Chlorophyll-α

i. Subsampling: Rinse graduated cylinder 3 times with ultrapure water. Invert /mix sample jug well and rinse graduated cylinder with sample. Filter 50mLs of water onto a 25 mm GF/F. Do not allow air to be sucked through the filter. Place filter in 15 mL amber vial. Filter 2 replicates

two.

ii. Storage: Store in cryofreezer.

e. Phycocyanin

i. Subsampling: For master stations only, rinse graduated cylinder 3 times with ultrapure water. Shake sample jug well and rinse graduated cylinder with sample. Filter as much water as possible onto a nucleopore 1.0 μm filter. Rinse any sample left on cylinder and filter column onto filter with ultrapure water. Store filter in a screw cap microcentrifuge tube. Collect 3 replicates.

ii. Storage: Store in cryofreezer.

D.2 Determining Chlorophyll-α Concentrations

a. Fill water bath with deionized water and turn on about 45 minutes before use. The water bath is already set to the proper operating temperature (65°C).

b. Test the automatic dispenser attached to the DMF jug by dispensing into a 10 ml graduated cylinder. Should result in 6.5 ml.

c. Remove GF/F-filtered samples from freezer and add 6.5 ml of DMF to plastic centrifuge tubes/amber vials containing filter.

d. Record sample ID and volume of water filtered for each sample

e. Turn on fluorometer to allow time to warm up.

f. Place tubes (with caps loosened) in water bath at 65°C for 25 minutes. Remove tubes from water bath and allow them to cool to room temperature.

g. Agitate each tube with vortex shaker for about 15-20 seconds.

h. Take a BLANK reading (DMF only) and also read the 2007 and 2009 solid standards.

i. Pour liquid into clean cuvette (fill to at least half) without disturbing filter.

j. Place cuvette in fluorometer and record chlorophyll reading when stable.

k. Repeat steps I & J for each sample tube.

l. Pour waste DMF into DMF waste bottle in the hood, discard plastic centrifuge tubes and cuvettes in blue drum.
D.3 Determining Phycocyanin Concentrations

a. Depending on water clarity, filter between 50 to 150mL of collected water, with a Nucleopore 47mm diameter, 0.4µm thickness filter (cat. no. 111107). Graduated 15mL conical polypropylene tubes can be used to store the filters. Once filtration is complete, store containers in the -80°C cryo-freezer until future use.

b. All procedures from here on need to be competed in low level light. Remove the test tubes from the cryo-freezer. Transfer 9mL of Phosphate Buffer, pH=6.8 APHA- for phenos (Fisher cat. No 5807-32). Make sure that the filters are submerged in the solution. Place tubes with filters and buffer on shaker table in a dark incubator at 5°C for 15 minutes. Vortex solution at medium speed by hand for 10 seconds. Place tubes in standard freezer (-20 to -10°C) to freeze for 2 hours until frozen.

c. Remove test tubes from the freezer and place on shaker table in a dark incubator set to 5°C to thaw for 2 hours. The goal is to remove the maximum amount of substrate from the filter.

d. Freeze overnight and thaw the next day as above.

e. After thaw is complete, samples need to be sonicated. Utilize the Fisher FS110 H sonicator and sonicate samples in ice water for 20 minutes.

f. When sonication is complete, allow samples to extract in a dark incubator on a shaker table overnight at 5°C

g. The next day, use a probe sonicator to sonicate each sample individually for one minute while sitting in an ice bath.

h. Centrifuge samples at 4700 xG for 20 minutes at 7°C.

i. Remove samples from centrifuge. Pipette 3.1 mL buffer into a cuvette.

j. Transfer the cuvette to the AquaFluor and get a reading for phycocyanin and phycoerythrin and record. Repeat for each sample.

k. Input relative fluorescence readings into a spreadsheet. Use the calibration curve for each AquaFluor unit to determine the actual concentration of extracted Phycocyanin in the sample. (concentration PC (ug/L) = ((slope of calibration curve * relative fluorescence)*(volume of Phosphate Buffer/1000)) / (volume filtered/1000).

D.4 Colored Dissolved Organic Matter Analysis (Johengen, 2017)

Sample Preparation – Approximately 40 ml of sample water will be filtered and used to rinse the sides of the flask and the 50 ml BD Falcon storage centrifuge tubes, and then discarded. Following the rinse, an additional 45 ml of the CDOM designated sample will be subsequently filtered using 47 mm GF/F filters (0.7 µm pore size) with low vacuum pressure (<5 in Hg). The filtrate will be place in the centrifuge tube, capped, wrapped with Parafilm, labeled, and stored in a refrigerator (4° C) until analysis. All samples will be shipped to MLML on dry ice for analysis using a calibrated laboratory-grade spectrophotometer.

Sample Spectrophotometric Analysis – The sample and 200-300 ml of MilliQ will be equilibrated to room temperature (failure to match the blank and sample temperature results in an artifactual feature in the spectrum at long wavelengths). The spectrophotometer will be allowed to warm up (30-40 min) before scanning and the scanning set for 1-2 nm intervals, with a 4-6 nm slit width.
The blank will be set with MilliQ water in cuvettes in both the sample and reference positions (dual-beam instrument) or by setting 0 and 100% transmission with a MilliQ blank (single-beam instrument). Matched 10 cm quartz or optical glass cells will be used for a dualbeam spectrophotometer and a single cell in a single-beam spectrophotometer. A rescan of the blank will be run to verify that the instrument has not drifted and to define the amplitude of noise.

The sample will be mixed by rocking the centrifuge tube before decanting the sample. The sample cuvette will be emptied and rinsed with 10-15 ml of sample prior to filling with sample and checked to ensure that there are no air bubbles in the light path when filled. Scans will be run between 250 and 800 nm (quartz) and electronic files will be saved for each sample. The cuvette will then be rinsed with MilliQ between samples. A MilliQ blank will be run after every 5th sample. For dilute samples (those that are not significantly yellow in color when viewed down the 10 cm pathlength of the cuvette or have OD < 0.2 at 400 nm), triplicate scans of each sample will be run and averaged to reduce the effect of noise.

Parameterizing Absorption – The absorption spectrum of CDOM follows an approximately exponential decline in the visible (400-700) wavelengths. Absorption from optical density will be calculated by subtracting the optical density at 750 nm to correct for residual scattering and converted as:

\[ a[\lambda] = \frac{2.3 \times (\text{OD}[\lambda] - \text{OD}[750])}{d} \]  

(1)

where \(a[\lambda]\) is absorption (m\(^{-1}\)) at wavelength \(\lambda\) (nm), OD is optical density (dimensionless), and \(d\) is pathlength (0.1 m).

Because the magnitude of the parameters that describe the CDOM fit are dependent on the range and means of the curve fitting, it is critical to employ the same protocol. A non-linear fit over the range 400 – 700 nm (Equation 2) will be used. This equation minimizes the effect of the unfavorable signal:noise ratio at long wavelengths on the goodness-of-fit at the wavelengths of most interests (i.e. the excitation wavelengths in the 400 – 500 nm range); is less sensitive to thermal artifacts at long wavelength; and is less sensitive to the range of wavelengths used than the linear fit (Equation 3).

\[ a[\lambda] = a[400]e^{-S\lambda} \]  

(2)

\(a[\lambda]\) is absorption (m\(^{-1}\)) at wavelength \(\lambda\), \(a[400]\) is absorption (m\(^{-1}\)) at the anchor wavelength of 400 nm, and \(S\) is the spectral slope (nm\(^{-1}\)). Note that wavelength must be expressed as \(\lambda - 400\) before fitting for the anchor value to be at 400 nm.

An alternative for those without a non-linear fitting package is to log-transform the absorption data and fit to the linear equivalent.

\[ \ln(a[\lambda]) = \ln(a[400]) - S\lambda \]  

(3)

Note that the anchor wavelength must be expressed as the anti-log for reporting. Because the linearized fit is sensitive to the dispersion due to noise at long wavelengths, this is best fit only for the interval 400-500 nm.

The parameter estimates and standard errors for \(a[\lambda]\), \(S\) will be reported along with the \(R^2\).
D.5 Quality Assurance Methods (Johengen, 2017)

ACT assessments include technical audits and data quality assessments. Fundamental principles of the ACT assessment process include:

a. Assessments are performed by the ACT QA Manager, who is independent of direct responsibility for performance of the Verification.

b. Each assessment is fully documented.

c. Each assessment must be responded to by the appropriate level of the ACT team. ACT quality assessment reports require a written response by the person performing the inspected activity, and acknowledgment of the assessment by the ACT Director.

d. Corrective action must be documented and approved on the original assessment report, with detailed narrative in response to the assessor’s finding. Initials and date are required for each corrective action response. Acknowledgment of the response will be provided by the ACT Director.

Technical Audits – Technical audits are systematic and objective examinations of the verification test implementation to determine whether data collection activities and related results comply with the Test Protocols, are implemented effectively, and are suitable to achieve its data quality goals. Audits for the HABS Fluorometry Verification will include: (1) technical system audits (TSAs) and audits of data quality (ADQs)

A TSA is a thorough, systematic, and qualitative evaluation of the sampling and measurement systems associated with a Verification test. The objective of the TSA is to assess and document the conformance of on-site testing procedures with the requirements of the Test Protocols, published reference methods, and associated SOPs. The TSA assesses test facilities, ACT Fluorometer II Protocols, PV17-01 16 equipment maintenance and calibration procedures, reporting requirements, sample collection, analytical activities, and QC procedures. Both laboratory and field TSAs are performed. The QA Manager will conduct a TSA of the laboratory component and at least one field test during the verification. The TSA is performed following the EPA document Guidance on Technical Audits and Related Assessments for Environmental Data Operations, EPA QA/G-7, January; 2000.A TSA checklist based on the Test Protocol is prepared by the QA Manager prior to the TSA and reviewed by the ACT Chief Scientist. At the close of the TSA, an immediate informal debriefing will be conducted. Non-conformances are addressed through corrective action. The QA Manager will document the results of TSAs and any corrective actions in a formal audit report.

An ADQ is a quantitative evaluation of the verification test data. The objective of the ADQ is to determine if the test data were collected according to the requirements of the Test Protocols and associated SOPs and whether the data were accumulated, transferred, reduced, calculated, summarized, and reported correctly. The ADQ assesses data accuracy, completeness, quality, and traceability. The ACT QA Manager conducts the ADQ after data have been 100% verified by the ACT Chief Scientist. The ADQ entails tracing data through their processing steps and duplicating intermediate calculations. A representative set of the data (10%) is traced in detail from raw data and instrument readouts through data transcription or transference through data manipulation through data reduction to summary data, data calculations, and final reported data. The focus is on identifying a clear, logical connection between the steps. Particular attention is paid to the use of QC data in evaluating and reporting the data set. Problems that could impact data quality are immediately communicated to the ACT Chief Scientist. The results of the ADQ are documented in a formal audit report with conclusions about the quality of the data from the verification and their fitness for their intended use.
Data Quality Assessment – ACT reviews technology testing data to ensure that only sound data that are of known and documented quality and meet ACT technology testing quality objectives are used in making decisions about technology performance. Data assessment is conducted in two phases. The first phase consists of reviewing and determining the validity of the analytical data – data verification and validation. The second phase consists of interpreting the data to determine its applicability for its intended use – usability assessment. Data verification is the process of evaluating the completeness, correctness, and consistency of the test data sets against the requirements specified in the Test Protocols. Data verification is conducted by the ACT QA Manager. The process includes verifying that:

- the raw data records are complete, understandable, well-labeled, and traceable;
  - a. all data identified in the Test Protocols has been collected;
  - b. instrument calibration and QC criteria were achieved;
  - c. data calculations are accurate.

Corrective action procedures are implemented if data verification identifies any non-compliance issues.

Data validation evaluates data quality in terms of accomplishment of measurement quality objectives, such as precision, bias, representativeness, completeness, comparability, and sensitivity. Data validation:

- a. establishes that required sampling methods were used and that any deviations were noted;
- b. ensures that the sampling procedures and field measurements met performance criteria and that any deviations were noted;
- c. establishes that required analytical methods were used and that any deviations were noted;
- d. verifies that QC measures were obtained and criteria were achieved; and that any deviations were noted.

Data validation is performed by the ACT QA Manager. Any limitations on the data and recommendations for limitations on data usability are documented. Data usability assessments determine the adequacy of the verified and validated data as related to the data quality objectives defined in the Test Protocols. All types of data and associated information (e.g., sampling design, sampling technique, analytical methodologies) are evaluated to determine if the data appear to be appropriate and sufficient to support decisions on technology performance. A data usability assessment has an analytical and a field component. An analytical data usability assessment is used to evaluate whether analytical data points are scientifically valid and of a sufficient level of precision, accuracy, and sensitivity. The field data usability assessment evaluates whether the sampling procedure (e.g., sampling method, sample preservation and hold times) ensures that the sample that is collected for analysis is representative.

Corrective Action - Corrective action is implemented in response to any situation that compromises the quality of testing or data generated by ACT. The need for corrective action can be identified by any ACT personnel and implemented with the prior approval of the ACT Chief Scientist, in consultation with the QA Manager. The Chief Scientist is responsible for determining appropriate corrective action to address an issue. Any findings that have a direct impact on the conduct of the verification test will be corrected immediately following notification of the finding. Implementation of corrective actions must be verified by the ACT QA Manager to ensure that corrective actions are adequate and have been completed. This will be done in real-time if corrective actions can be immediately performed. All corrective actions are
documented. Any impact that an adverse finding had on the quality of the test data is addressed in the test report.

*Audit Reporting* - The ACT QA Manager is responsible for all audit reports. These written reports:

a. identify and document problems that affect quality and the achievement of objectives required by the Test Protocols and any associated SOPs;
b. propose recommendations (if requested) for resolving problems that affect quality;
c. independently confirm implementation and effectiveness of solutions;
d. identify and cite noteworthy practices that may be shared with others to improve the quality of their operations and products;
e. provide documented assurance that when problems are identified, further work performed is monitored carefully until the problems are suitably resolved.

**D.6 Quality Control Methods** (Johengen, 2017)

*Field Trip Blank* – Sample containers filled with reagent water (Type 1 reagent grade deionized water) are taken to the field and processed identically to field reference samples to evaluate contamination introduced during sampling, storage and transport. Four field trip blanks will be collected for each field test spaced evenly throughout the deployment period.

*Field Duplicates* – we will collect two reference sample water bottles simultaneously at approximately 10% of the sampling points to examine fine scale spatial heterogeneity within the mooring arrangement.
### Appendix E: Laboratory Test Concentrations and Volumes

<table>
<thead>
<tr>
<th>Component</th>
<th>Species Name</th>
<th>Stock Concentration</th>
</tr>
</thead>
<tbody>
<tr>
<td>Species A</td>
<td><em>Clorella</em> sp.</td>
<td>485</td>
</tr>
<tr>
<td>Species B</td>
<td><em>Cryptomonas</em> sp.</td>
<td>520</td>
</tr>
<tr>
<td>Species C</td>
<td><em>Aphanizomenon</em> sp.</td>
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</tr>
<tr>
<td>Species D</td>
<td><em>Peridinium inconspicuum</em></td>
<td>90</td>
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<tr>
<td>Turbidity</td>
<td>N/A</td>
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</tr>
<tr>
<td>CDOM</td>
<td>N/A</td>
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</table>

<table>
<thead>
<tr>
<th>Trial</th>
<th>Species</th>
<th>Target Concentration</th>
<th>Volume Needed</th>
<th>Total from Stocks</th>
<th>Media Base</th>
</tr>
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<td>10</td>
<td>41</td>
<td>41</td>
<td>1959</td>
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<td>38</td>
<td>38</td>
<td>1962</td>
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<tr>
<td>3</td>
<td>C only</td>
<td>10</td>
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<td>200</td>
<td>1800</td>
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<tr>
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<td>222</td>
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<td>A</td>
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<td>plus Turbidity</td>
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<td>Sample Date</td>
<td>Sample Time</td>
<td>Isolation Time</td>
<td>River Discharge (ft³/s)</td>
<td>Reference Sample</td>
<td>YSI EXO2 Multiparameter Sonde</td>
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<td>12.55</td>
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## Appendix G: Surface Mapping Deployment Data

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<th>pH</th>
<th>Turb (NTU)</th>
<th>Exo2</th>
<th>ODO (mg/L)</th>
<th>Chl-a (µg/l)</th>
<th>Chl-a (RFU)</th>
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Appendix H: IES Reflection

My time spent working at CIGLR was a wonderful learning experience. This summer provided me with opportunities to develop and fine tune my skills in field work, laboratory analysis, and data analysis. I worked on the in situ fluorometer evaluation from start to finish, not only collecting the samples, but also processing, analyzing, and evaluating them. I gained exposure to advancing fluorometry technologies, how to operate them, and their accuracies. I very quickly gained independence performing intricate laboratory analyses of chlorophyll-a and phycocyanin.

Working toward a degree at the Institute for the Environment and Sustainability provided me a well-rounded skill set that is very practical in the working world. In courses such as Environmental Protocols and Hydrogeography, I gained exposure to different types of surface water field work. Having those experiences opened doors to employment opportunities, such as this one, that were partially based on similar field work. Environmental Protocols also gave me a crash-course in Excel and the functions most useful when working with large datasets. I used those skills and developed them further during this fellowship. Additionally, the Professional Service Project I chose allowed me to more thoroughly investigate watershed characteristics on a comprehensive scale. Having that detailed foundation of knowledge and experience let me easily grasp concepts related to water quality during my time at CIGLR.

Having worked at the Ohio EPA and CIGLR, which is tied to the National Oceanic and Atmospheric Administration, I know first-hand how rewarding it is to research and monitor water quality problems that negatively impact environmental and human health. Since I began studying at Miami University, it has been a goal of mine to work directly with the HABs of Lake Erie. I used every possible academic opportunity to research and investigate water quality regarding the Great Lakes, and I achieved that goal this summer by working at CIGLR. I believe my background in government work and the comprehensive education I received from the Institute for the Environment and Sustainability will set me up to continue in this rewarding career path with the ultimate goal of improving water quality for all organisms that depend on it.