Standard Operating Procedure: Collection Of Lake Water Quality Samples

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Approval Signatures

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LCI QAPP Update Log

Prepared/Revised By:	Date:	Revision No:	Summary of Changes:
Karen Woodfield Alene Onion	03/2019	1.0	Updates
Karen Woodfield Alene Onion Matt Kraft	04/2020	2	Updates: -
Karen Woodfield Alene Onion	04/2020	2.1	• Added revision log • Added COVID19 Modifications

Matt Kraft Karen Stainbrook			• Added Sediment Diatom Sampling

No substantive changes include updating references, correcting typographical errors, and clarifying certain language to make the document more useful and effective.

1 Scope and Applicability

COVID-19 modification: In order to minimize exposure and to protect staff while continuing to execute the core mission of Division of Water and its water quality monitoring programs, modifications were developed that incorporate social distancing recommendations driven by COVID-19. Modifications to protocols for collection of water quality data as part of DOW monitoring programs are contained within this SOP. All sampling scenarios described within recommend that samplers wear gloves throughout the day to minimize direct contact with equipment surfaces. This is in addition to described protocols requiring samplers to wear gloves to minimize sample contamination. Where applicable, all protocols described below should be conducted in accordance with guidance provided in SOP#603-20 Guidance for Field Work During COVID-19 Pandemic.

2 Summary of Method

3 Health and Safety Warnings

3.1 When working with potentially hazardous materials, follow EPA, OSHA, and NYS DEC DOW (2018) (https://nysemail.sharepoint.com/sites/DECInSite-DOW/DOW%20Documents/hasp2019.pdf?csf=1&e=idFtwD&cid=4b2bfa9d-ceff-4e16-aac8-bcb8b7bb588f) specific health and safety procedures and refer to Material and Safety Data Sheets of hazardous chemicals. Material safety data sheets (MSDS) are contained in a binder in the sample prep room and are on the S drive at S:BWAM:DOW H&S:RIBS

3.1.1 COVID19 Modification: follow additional health and safety procedures during COVID-19 detailed in “Guidance for Field Work During COVID-19 Pandemic” (SOP #603-20).

3.2 When conducting fieldwork, there should always be at least two people in a field team. A form of emergency communication (e.g., cell phone) and a first aid kit should be carried with the field team.

3.2.1 COVID19 Modification: If staff are using multiple watercrafts to perform field work, staff must be visible to each other when on the water.

3.3 When conducting sampling from a boat in an impoundment or flowing waters, follow appropriate boating safety procedures as described in the Division’s Boating Safety Program.

3.4 COVID19 Modification: As per SOP#603-20 COVID-19 Guidance, staff should practice social distancing by maintaining a distance of at least 6' from other people whether outdoors, in vehicles or indoors. The use of multiple vehicles is recommended when possible. When use of multiple vehicles is not practicable, each person must wear a mask or cloth face covering when more than one person is present in the vehicle. When sampling from an impoundment with suspected or known quantities of hazardous materials, employ health and safety procedures in a manner consistent with Division of Water's Health and Safety Program.

3.5 When collecting a grab sample from a suspected cyanobacteria harmful algal bloom (HAB) scum, use safety gloves to protect skin. If skin exposure occurs, rinse area with clean water.

4 Interferences

4.1 There are two primary interferences or potential problems with surface water sampling. These include cross-contamination of samples and improper sample collection. Cross-contamination problems can be eliminated or minimized using dedicated sampling equipment. If this is not possible or practical, then cleaning of sampling equipment is necessary.

4.1.2 Improper sample collection can involve using contaminated equipment, disturbance of the stream or impoundment substrate, and sampling in an obviously disturbed or non-representative area.

4.2 Following proper cleaning procedures, minimizing disturbance of the sample site, and choosing an appropriately representative sampling site will eliminate these problems.

5 Definitions

5.1 Deepwater samples: samples used to characterize uniformly cold waters and collected 1-2 m from the lake bottom (see section 11.5.2.2 for when and how to determine lake bottom sampling)

5.2 Water column: The vertical section of water between the waterbody surface and the stream bottom.

5.3 Epilimnion: upper layer of a lake that has a fairly uniform warm temperature

5.4 Hypolimnion: mass of uniformly cold, dense water at depth in a lake

5.5 Metalimnion: a region located below the epilimnion in which temperature decreases rapidly with depth; also referred to as the thermocline

5.6 Stratification: vertical profile of a ponded waterbody that is controlled largely by density differences, which are generally a consequence of temperature differences. For the purposes of this SOP, this term refers to thermal stratification, not chemical stratification or the development of other strata

5.7 Surface: for the purposes of this SOP, "surface" is defined as the first 2 meters of water depth or half the total depth in lakes less than 2 meters.

6 Personnel Qualifications

6.1 All field samplers are aquatic biologists, engineers, environmental program specialists, research scientists, or environmental technicians, or have been trained in appropriate field sampling techniques for qualified samplers (as cited above) prior to collecting samples submitted for analysis. All non-professional samplers, such as participants in state-sanctioned volunteer monitoring programs, have participated in a 4-hour training course conducted by qualified professional staff.

7 Equipment and Supplies

7.1 Equipment needed for collection of surface water samples includes all or an appropriate subset of the following:

7.1.1 Handheld depth finder

7.1.2 Kemmerer bottles[1]

7.1.3 Van Dorn bottles

7.1.4 Cables, lines, and messengers

7.1.5 Amber polyethylene sampling containers

7.1.6 Amber glass sampling containers for cyanotoxin analysis

7.1.7 Churn splitter or carboy

7.1.8 Secchi disk (limnological disk- 20cm alternating black and white quarters) with measured line

[1] The appropriate sampling device must be of proper composition. Samplers constructed of brass/stainless steel, PVC or PFTE (Teflon) should be used based upon the analyses to be performed. In general, PVC samplers are used for collecting samples to be analyzed for most inorganic constituents (except for chloride) and brass, Teflon or stainless-steel samples are used for organic constituents, such as pre-cursors to THM formation potential, for which PVC could contaminate the sample.

7.1.9 Single- or multi-probe electronic meter for temperatures, dissolved oxygen, pH, conductivity, OPR, depth and other parameters

7.1.10 Tethered rake with marked retrieval line

7.1.11 Field filtration apparatus (funnel, centerpiece, receiving flask, hand-operated or compressed air field vacuum pumps, graduated cylinders)

7.1.12 Wisconsin-type plankton net (30cm diameter, No. 10 or equivalent, mesh = 80 µ, 153µ or 243 µ), collection barrel, weighted collar, and measured line

7.1.13 Carbonated water

7.1.14 Formalin-rose Bengal solution

7.1.15 Distilled or deionized water

7.1.16 Forceps

7.1.17 Filters: glass fiber (0.45µm) for chlorophyll a and DOC, and phosphorus free membrane filters (0.45µm) for all other filtered samples

7.1.18 Field biological collection bottles, including kick sample jars, polyethylene (1 qt.)

7.1.19 Ethylene alcohol preservative

7.1.20 Sieve bucket with handle, U.S. std No 35, 500 µ mesh Sieve bucket with handle, U.S. std No 35, 500 µ mesh

7.1.21 Petite ponar with tethered marked line

7.1.22 Concrete block(s) with float labeled with DEC-DOW tag

7.1.23 Plastic tray for floating aquatic plants or holding material collected from petite ponar or artificial substrate

7.1.24 Pre-labeled analyte aliquot bottles

7.1.25 Re-sealable plastic zipper storage bags

7.1.26 Ice/ice packs

7.1.27 Cooler(s)

7.1.28 Chain of custody forms

7.1.29 Field data sheets

7.1.30 iPad and associated accessories

7.1.31 SPOT (Satellite Personal Tracking) System

7.1.32 Bathymetric maps or topographic maps with overlay sampling grids

7.1.33 Safety gloves, glasses, and shoes. COVID19 Modification: Additional personal protective gear should be worn during the COVID19 pandemic as per SOP 603-20 section 7.

7.1.34 Hand-held Global Positioning System (GPS)

7.1.35 Buoys and/or anchors

7.1.36 Logbook and waterproof pen

7.1.37 Sediment diatom sampling equipment including a gravity corer, sediment core extruding apparatus, metal spatula, measuring tape (in cm), sectioning stage, wash bottle, and rubber stopper and cap for sediment core tube

7.1.38 Sample bottle labels (for un-labeled sample aliquot bottles or biological sample bags)

7.1.39 Approved QA project plan (QAPP)

7.1.40 Approved field health and safety plan

7.1.41 Equipment checklist

8 Sample Collection- Preparation

COVID19 Modification: As per SOP#603-20 COVID-19 Guidance, staff should practice social distancing by maintaining a distance of at least 6’ from other people whether outdoors, in vehicles or indoors. To minimize exposure between field staff, equipment and tasks should be assigned to each individual before field work begins. Unless equipment is disinfected between uses, staff should maintain consistent use of that equipment and performance of specified tasks to prevent cross contamination between coworkers. The wearing of a mask or cloth face covering is required during field work and other times when staff must be in close proximity with each other and social distancing measures cannot be maintained.

8.1 Determine the extent of the sampling effort and the sampling methods to be employed.

8.2 Secure necessary sampling and monitoring equipment.

8.2.1 COVID19 Modification: Unless equipment is disinfected between uses, staff should maintain consistent use of that equipment and performance of specified tasks to prevent cross contamination between coworkers. To minimize exposure between field staff, equipment and tasks should be assigned to each individual before field work begins. To comply with SOP#603-20 Guidance, each team will consist of two staff. STAFF 1 will be responsible for the collection of field observations, secchi, the depth profile, macrophyte samples, sediment diatom sampling and/or D-net macroinvertebrate sampling while STAFF 2 will be responsible for water column sample collection, HABs samples, depth, bathymetry, and/or ponar macroinvertebrate sampling. Each staff will prepare the equipment for his/her assigned sampling methods.

8.3 Calibrate equipment that requires calibration.

8.3.1 COVID19 Modification: This task is completed by one staff to prevent COVID19 exposure. Multiple sondes may be calibrated at one time as long as proper disinfectant procedures (below) are followed afterwards.

8.4 Clean equipment and ensure that it is working properly. Ensure that sample collection devices do not have any significant scratches or any leaks (SOP#103-19 Equipment Cleaning).

8.5 Prepare scheduling and coordinate with sampling staff.

8.5.1 COVID19 Modification: As per SOP#603-20 COVID-19 Guidance, staff should practice social distancing by maintaining a distance of at least 6' from other people whether outdoors, in vehicles or indoors. To minimize exposure between field staff, equipment and tasks should be assigned to each individual before field work begins. Unless equipment is disinfected between uses, staff should maintain consistent use of that equipment and performance of specified tasks to prevent cross contamination between coworkers. The wearing of a mask or cloth face covering is required during field work and other times when staff must be in close proximity with each other and social distancing measures cannot be maintained. The use of multiple vehicles is recommended when possible. When use of multiple vehicles is not practicable, each staff member must wear a mask or cloth face covering when more than one person is present in the vehicle.

8.6 Prepare a float plan to identify times and location s where sampling staff are expected to complete on-water sampling procedures and depart from boats, and to identify instructions for float plan holders to follow in the event of overdue boaters (instructions are listed on the float plan itself). Identify a primary and a secondary float plan holder for each excursion.

8.7 Perform a general site survey prior to site entry in accordance with the site-specific health and safety plan and QA Project Plan.

8.8 Use buoys or GPS coordinates to identify and mark all sampling locations. GPS units are operated according to manufacturer's specifications. If required, the proposed locations may be adjusted based on site access, property boundaries, and surface obstructions.

9 Sample Collection - Cleaning and Procedures

9.1 Cleaning Procedures for Water Chemistry Sampling Equipment

Equipment should be cleaned prior to initial use to eliminate any contaminants introduced in the manufacturing or processing of the equipment or introduced during exposure to any contaminants while monitoring the water quality. When sampling in dilute systems, sampling equipment should be cleaned prior to use. Sampling equipment should also be cleaned when sampling from different layers (i.e., epilimnion and hypolimnion) within the stratified waterbody. Cleaning procedures can be minimized for equipment dedicated solely to sampling a specific waterbody.

9.1.1 General NYS DEC DOW equipment procedures should be followed (SOP#103-19 Equipment Cleaning).

9.1.2 The following procedures should also be used for cleaning lake water sample collection equipment:

9.1.2.1 Prior to initial use, wipe down the surfaces of any PVC-based equipment with hexane to remove any particulates introduced in the manufacturing process and rinse with copious amounts of distilled water to remove any dislodged debris.

9.1.2.2 Rinse all collection equipment with distilled water prior to each sampling run. Wipe and rinse (with distilled water) all surfaces to remove any detritus deposited during previous sampling runs or storage.

9.1.2.3 Collect a water sample from the stratum to be evaluated and discharge a small portion (at least 10-20ml) of the contents through the collection port of the sampling equipment to further rinse the entire collection device. This should be conducted as devices are transitioned from project waterbodies or from one stratum to another within the same waterbody

9.1.2.4 If the compositing container was appropriately rinsed at the previous sampling location, discharge a portion of the water sample from the stratum to be evaluated into the compositing container and rinse the composting container with it to acclimatize the compositing container for sample collection.

9.1.3 COVID19 Modification: Additional cleaning procedures during COVID19 Pandemic:

9.1.3.1 COVID19 Modification: As per SOP#603-20 COVID-19 Guidance, staff should practice social distancing by maintaining a distance of at least 6' from other people whether outdoors, in vehicles or indoors. Unless equipment is disinfected between uses, staff should maintain consistent use of that equipment and performance of specified tasks to prevent cross contamination between coworkers. The wearing of a mask or cloth face covering is required during field work and other times when staff must be in close proximity with each other and social distancing measures cannot be maintained.

9.1.3.2 COVID19 Modification: Clean ipads using Clorox disinfecting wipes as per SOP#603-20 and Apple recommendations:

- Using a 70 percent isopropyl alcohol wipe or Clorox Disinfecting Wipes, you may gently wipe the hard, nonporous surfaces of your Apple product, such as the display, keyboard, or other exterior surfaces. Don't use bleach.
- Avoid getting moisture in any opening, and don't submerge your Apple product in any cleaning agents.
- Unplug all external power sources, devices, and cables.
- Keep liquids away from the product, unless otherwise noted for specific products.
- Don't get moisture into any openings.
- Don't use aerosol sprays, bleaches, or abrasives.
- Don't spray cleaners directly onto the item.

9.1.3.3 COVID19 Modification: Clean YSI sondes with a bleach solution: 1/3c bleach / 1 gallon water as per SOP#603-20 and YSI recommendations.

9.1.3.4 COVID19 Modification: If not specified above, equipment should be disinfected according to SOP#603-20.

9.1.3.5 COVID19 Modification: All areas of field equipment which may have been touched needs to be disinfected according to SOP#603-20. This includes but is not limited to:

- churn handle, spigot and outside surface;
- Van Dorn rope, handle, spigot and outside surface;
- integrated tube outside surface;
- Macrophyte rake handle and rope;
- YSI handheld surface;
- YSI sonde cable, storage cup, and outside surface;

- YSI case handles, zipper, and outside surface; secchi disk surface and rope;
- Deep water bottle outside surface
- Anchor handle and rope
- Paddle handles and shafts
- PFDs including clasps and neckline
- Cooler handles, clasp and outside surface
- Scissors and packing tape dispensers

10 Sample Collection – Choice of Sampling Device

10.1 The deciding factors or questions to be considered during the selection of a sampling device for sampling surface waters in lakes, ponds, surface impoundments, and other ponded waters are as follows:

10.1.1 Will the sample be collected from the shore or from a boat?

10.1.2 What is the desired depth at which the sample is to be collected?

10.1.3 Are data required from individual strata or from composited strata?

10.1.4 Is the sample intended to replicate representative conditions at the time of collection, or some other measure (worst case conditions, unusual events, etc.)?

10.1.5 Will the desired dataset be compromised by subjecting the sample to atmospheric conditions or the time required to perform analyses on discrete samples?

10.1.6 Will the sample be contaminated by the collection device?

10.1.7 What will the sample be analyzed for (i.e., volatile organics)?

10.1.8 Does the strength and dexterity of the sampler (professional or volunteer) require the use of a sampling device that requires the ability to set a spring-loaded sampler or balance a long integrating tube?

11 Sample Collection – Method Options and Procedures

11.1 COVID19 Modification: Sampling activities will be divided between two staff to maintain social distancing. In general, STAFF 1 will be responsible for the collection of field observations, secchi, the depth profile, macrophyte samples, sediment diatom sampling and/or D-net macroinvertebrate sampling while STAFF 2 will be responsible for water column sample collection, HABs

samples, depth, bathymetry, and/or ponar macroinvertebrate sampling. Each staff will prepare the equipment for his/her assigned sampling methods.

11.2 Location Finder (STAFF 2)

11.2.1 When possible, sampling locations are pre-determined with the use of GPS coordinates, shoreline triangulation, or other means identified during desktop evaluations, field reconnaissance, or previous sampling sessions.

11.3 Depth Finder (STAFF 2)

11.3.1 A hand-held depth finder is used to determine and verify sampling locations (based on known bathymetry) and provide incidental depth information for aquatic vegetation and benthic samples.

11.3.2 Depth measurements are collected as follows:

11.3.2.1 Loop depth finder around wrist and hold sensor below surface, pointing toward the lake bottom and perpendicular to the lake surface

11.3.2.2 Depress and release button

11.3.2.3 Record depth to nearest 0.1 meters (or feet)

11.4 Discrete Sampling by Direct Measurement (STAFF 1)

11.4.1 Direct measurement of water quality indicators is conducted with the use of electronic meters connected to field probes with cables. Analyses are completed through the use of multi-probe units, dual probe units (such as temperature/ oxygen meters) or single probe field meters.

11.4.2 The general instructions for conducting depth profiles are as follows:

11.4.2.1 Make sure that the equipment has been appropriately calibrated. Follow the manufacturer's directions when calibrating and using the meter. Replace membranes in accordance with the schedule and methodology provided in the manufacturer’s instructions. Record all calibration measurements in a logbook and download field calibration log files weekly.

11.4.2.2 When taking pH measurements, make sure that the equipment has been appropriately calibrated using standard buffers that bound the expected pH of the lake.

11.4.2.3 If data logging is planned, use the instruments instructions to create a data log file for the waterbody, noting the waterbody

name and date. Location within the waterbody should also be noted if multiple sites are scheduled for conducting profiles with data logging.

11.4.2.4 Lower the probe into the water until the aperture of the probe is completely submersed.

11.4.2.5 Verify readings have stabilized, as indicated by the multi-probe displays. Record all pertinent analytes either manually or by logging onto the display. For displays that exhibit the results for only a single analyte at a time, toggle between settings for the pertinent analytes.

11.4.2.6 Lower the probe until either the next pertinent interval is displayed (on units that record water depth), or to the next mark on the calibrated cable line. Repeat step 11.4.2.5.

11.4.2.7 Repeat steps 11.4.2.5 and 11.4.2.6 until the probe is within 1 meter of the bottom of the sampled waterbody.

11.4.2.8 Repeat steps 11.4.2.3 through 11.4.2.7 for any additional sites on the lake

11.4.2.9 Water temperature can be alternatively determined by inserting a thermometer to the immersion line in a bucket (or CSLAP collapsible container) of sample water that has been placed in the shade after collection. Permit the mercury column to stabilize (approx. 2 minutes) and read and record the temperature while the thermometer is still immersed.

11.5 Discrete Sampling with Kemmerer Bottles (STAFF 1)

11.5.1 A Kemmerer bottle may be used in most situations where site access is from a boat or structure (e.g., bridge or pier) and where samples at depth are required. Kemmerer bottles can be used in monitoring programs (waterbody-specific sampling) in which all sampled waterbodies exceed 3 meters in depth. For programs where depth ranges are not known in advance, Van Dorn bottles should be used.

11.5.2 General sampling procedures for the use of Kemmerer bottles are as follows:

11.5.2.1 Using a Kemmerer bottle that has been appropriately cleaned, set the pre-calibrated sampling device so that the sampling end stoppers are positioned away from the sampling tube, allowing the sampled substance to easily pass through the tube.

11.5.2.2 Lower the sampling device to the pre-determined depth. Surface samples are collected at a depth of 1.5 meters or ½ the depth of

the sampled waterbody, whichever is shallower. Deepwater samples (i.e., within the hypolimnion) are collected at a depth of 1-2 meters (usually 1.5 meters) above the bottom or at the maximum deployable depth, whichever is shallower. Use thermal profile to determine if lake is thermally stratified, defined as >5°C difference from surface to bottom. If screening data from previous year indicated thermal stratification, then assume deep samples should be collected in all intensive (next year) visits. If lake was thermally stratified (previous year or present sampling session), collect deep sample from 1.5 meters from lake bottom. If deep sample has any evidence of sediments or non-algal turbidity, dispose sample and collect again. If second sample also indicates sediment, retain second sample and note sediment in sample on field sheet. Avoid bottom disturbance to prevent sediment introduction into the sample. Any samples with visible suspended sediment must be discarded and the sample must be re-collected, unless visual observations of the sampling environment indicate high ambient turbidity.

11.5.2.3 When the Kemmerer bottle is at the required depth, send down the messenger to close the sampling device.

11.5.2.4 Retrieve the sampler and discharge the first 10 to 20 mL of sample to clear any potential contamination on the valve and, if not already rinsed, the compositing container. If suspended sediment is visible in the sample and not in the ambient environment prior to collecting the same, the sample will be discarded and re-collected.

11.5.2.5 Transfer the remaining sample to the appropriate compositing container.

11.5.2.6 Record the sample information in the field notebook.

11.6 Discrete Sampling with Van Dorn Bottles. (STAFF 2)

11.6.1 A Van Dorn bottle may be used in the same situations as the Kemmerer bottle, but Van Dorn's are preferred when any sampled waterbodies do not exceed 3 meters in depth or waterbody depth is not known in advance (during program planning stages). To maintain consistency in comparing results within each monitoring program, the same (choice of) collection device should be used in all sampled waterbodies within each monitoring program:

11.6.2 Sampling procedures for the use of the Van Dorn bottle are as follows

11.6.2.1 Ensure that the Van Dorn bottle has been properly cleaned, set the sampling device so that the tether cord attached to the sampling end stoppers are looped around the trigger posts and

away from the sampling tube to permit the water sample to easily pass through the tube.

11.6.2.2 Lower the calibrated sampling device to the pre-determined depth. Surface samples are collected at a depth of 1.5 meters or ½ the depth of the sampled waterbody, whichever is shallower. Deepwater samples are collected at a depth of 1.5 meters above the bottom or at the maximum deployable depth, whichever is shallower. Samples will be collected to avoid bottom disturbance.

11.6.2.3 When the Van Dorn bottle is at the required depth, send down the messenger to close the sampling device.

11.6.2.4 Retrieve the sampler and discharge a sufficient volume (at least the first 10 to 20 mL) to clear any potential contamination on the valve and, if not already fully rinsed - the compositing container. If suspended sediments are visible in the sample and not in the ambient environment prior to collecting the sample, the sample will be discarded and re-collected

11.6.2.5 Transfer the sample to the appropriate compositing container or directly into labeled sample containers.

11.6.2.6 Record the sample information in the field notebook.

11.7 Discrete Sampling by Dipping with Sterilized Bottles (STAFF 2)

11.7.1 While properly cleaned Kemmerer and Van Dorn bottles may be appropriate for collecting samples that must be transferred to unsterilized bottles, samples requiring transport and storage in sterile conditions should be collected directly in sterilized bottles. This includes samples collected for bacteriological analyses but applies only to those samples in which the appropriate sample depth (representative of surface conditions) is less than 1 meter.

11.7.2 Sampling procedures are as follows:

11.7.2.1 Label the sterilized bottles provided by the laboratory with the sample time, location and site names, and field identification number.

11.7.2.2 Immerse the inverted sterilized bottle. If there is any discernible flow, point the bottle in the direction of the flow, and always away from the sampler

11.7.2.3 Lower the sampling bottle to the appropriate sampling depth (6-10 inches below the surface for bacteriological sampling).

11.7.2.4 Uncap the bottle underwater to avoid introducing surface scum into the sample.

11.7.2.5 Tilt the container at a 45° angle and hold the container steady.

11.7.2.6 Allow the bottle to fill with water.

11.7.2.7 Bring the bottle out of the water and, if completely filled, pour out enough sample to leave sufficient headspace for sample expansion during freezing.

11.7.2.8 Cap the bottle, and place it in a cooler with ice

11.7.2.9 Record the sample information in the field notebook.

11.8 Discrete Lake Outlet Sampling by Dipping Directly with Compositing Bottles (STAFF 2)

11.8.1 While properly cleaned Kemmerer and Van Dorn bottles may be appropriate for collecting subsurface samples that must be transferred to compositing bottles, near surface samples at shallow outlets of lakes should be collected directly in unsterilized compositing bottles.

11.8.2 Sampling procedures are as follows:

11.8.2.1 Rinse the compositing bottle (using procedure 9.1.2) and use a temporary label with location or (if needed) site name and depth to distinguish this compositing bottle from others collected during the sampling run

11.8.2.2 Point the bottle in the direction of the flow and immerse the bottle.

11.8.2.3 Lower the sampling bottle to the appropriate sampling depth (the shallower of elbow depth or ½ the depth of the outlet site).

11.8.2.4 Uncap the bottle underwater to avoid introducing surface scum into the sample.

11.8.2.5 Tilt the container at a 45° angle and hold the container steady.

11.8.2.6 Allow the bottle to fill with water.

11.8.2.7 Bring the bottle out of the water and, if completely filled, pour out enough of the sample to leave sufficient headspace for sample expansion during freezing.

11.8.2.8 Cap the bottle, and place it in a cooler with ice

11.8.2.9 Record the sample information in the field notebook.

11.9 Shore bloom Harmful Algal Bloom Sampling (STAFF 2)

11.9.1 Procedures are established in SOP# 212-19 Collection of Harmful Algal Bloom Samples (HAB sample type SB) to collect a shore bloom surface skim sample when a HAB is visually apparent.

11.9.2 When HABs are visually apparent, SB samples are collected at the location along the shoreline with the perceived densest concentration of HABs, often at the windward shoreline or along the shore in protected coves, where material can accumulate and tends not to mix. SB samples are direct grab samples that that are collected by skimming the water surface to collect HABs material. Samples are to be collected directly in sample bottles.

11.10 Integrated Sampling with Integration Tubes (STAFF 2)

11.10.1 An integrated sampler is useful for non-homogeneous waterbodies, greater than 2.5 meters in depth, that require composited or integrated samples within a vertical plane.

11.10.2 This method may be for water samples or harmful algal bloom (HAB) samples. Procedures are established in SOP# 212-19 Collection of Harmful Algal Bloom Samples for the use of integrated tubes to sample HABs (HAB sample type TU).

11.10.3 Although discretely collected samples can be composited within a churn splitter or collection carboy, integrated hose (tube) samplers are preferred for discrete samples.

11.10.4 Sampling procedures are as follows for the use of integrated tubes for water samples:

11.10.4.1 If necessary, screw together the two sections of the sampling tubes, making sure the threads line up and connections do not leak.

11.10.4.2 If a tethered stopper is provided, remove the tethered stopper, make sure the valve is open, and submerge the connected sections into the water 2-3x to remove any debris and to acclimate the insides of the integrating tubes to the environmental sample.

11.10.4.3 For waterbodies more than 3 meters deep, submerge the tube until the surface of the water is just below the open end of the tube; for waterbodies less than 3 meters deep, submerge the tube to ½ the depth of the water.

11.10.4.4 Place the tethered stopper into the open end of the tube and retrieve the tube until the valve is just below the surface of the water. Close the valve.

11.10.4.5 Remove the tube from the water and place over the properly rinsed collecting churn splitter or, carboy. Open the valve and slowly discharge the sample. If any of the sample water escapes from the churn splitter or carboy, during discharge from the integrated sampling tube, discard the sample and return to step 12.8.2.3. Do not discharge partial integrated samples into the churn splitter or carboy, only entirely integrated samples.

11.11 Integrated Sampling with Plankton Nets

11.11.1 Plankton nets are used to integrate plankton trapped in a Wisconsin-type netting at discrete depths throughout either the entire water column or the photic zone.

11.11.2 Sampling procedures are as follows:

11.11.2.1 Using a clean plankton net and barrel, lower the plankton net and barrel from the lake surface to just above the lake bottom.

11.11.2.2 Raise the net from just above the bottom to the lake surface at a steady, consistent rate.

11.11.2.3 Raise the barrel above the lake surface and splash water through the outside of the meshing to free any trapped plankton and draw them into the barrel.

11.11.2.4 Agitate the barrel until most of the volume of water has dripped out.

11.11.2.5 Pour sample from the barrel into a 50 mL polypropylene vial, using a distilled water wash bottle to remove any remaining plankton.

11.11.2.6 Add carbonated water to within an inch of the cap to narcotize the zooplankton.

11.11.2.7 After 5 minutes, top off with formalin-Rose Bengal solution.

11.11.2.8 For thermally stratified lakes, repeat steps 11.11.2.1 through 11.11.2.7 on a second tow limited to the epilimnion, as determined by a vertical temperature profile.

11.12 Water Transparency Measurements with a Secchi Disk (STAFF 1)

11.12.1 A Secchi disk is used to measure water transparency as a surrogate for turbidity in ponded waters. Water clarity can be determined if measured transparency exceeds the water depth at the sampling site and if there is enough sunlight to illuminate the water column above the lowered disk.

11.12.2 Sampling procedures are as follows:

11.12.2.1 Lower the disk over the shady side of the boat until the disk just disappears from site. Record this depth to the nearest 0.1 meter.

11.12.2.2 Lower the disk one meter below the depth recorded in step 11.12.2.1. Raise the disk until the disk reappears in sight, and record to the nearest 0.1 meter. If this measurement varies from the first measurement by more than 0.5 meters or 10% of the depth in step 11.12.2.1, whichever is greater, repeat step 11.12.2.1 and this step.

11.12.2.3 Determine the reported Secchi disk transparency by computing the average of steps 11.12.2.1 and 11.12.2.2.

11.13 Use Impairment Assessment (STAFF 1)

11.13.1 Use impairment assessments are collected primarily in volunteer monitoring programs to evaluate recreational use conditions and public perception of water quality, and to link these assessments to conjointly collected water quality data. Assessments must be completed prior to collecting any other field measurements to avoid biasing impairment evaluations with direct water quality measurements. Shoreline assessments must be completed prior to open water assessments. All open water assessments are conducted at the corresponding sampling site.

11.14 Aquatic Plant Sampling with Rake Toss Sampling (STAFF 1)

11.14.1 Tethered rakes are used to collect and assess rooted aquatic plants, including submergent plants like Eurasian watermilfoil and floating plants like lily pads in a standardized format, using the sampling protocol established by the U.S. Army Corps of Engineers and Cornell University. This methodology is not appropriate for emergent plants, such as cattails; these will continue to be evaluated visually as presence/absence. Separate methods will be used at screening and intensive sites.

11.14.2 Sampling procedures for screening sites are as follows

11.14.2.1 While at the boat launch, identify GPS coordinates of the reference water quality sampling site. If the location of the reference site is not readily apparent, use landscape features to best determine where the deepest point of the lake is located.

11.14.2.2 Toss rake the length of the tethered line.

11.14.2.3 Retrieve rake slowly and bring rake and attached plants into boat.

11.14.2.4 Estimate overall plant abundance using USACE/Cornell abundance scale: “Zero” = no plants on rake; “Trace” = fingerful of plants on rake; “Sparse” = handful of plants on rake; “Medium” = most to all tines on rake covered with plants; “Dense” = difficult to bring into boat.

11.14.2.5 Remove plants from rake tines and separate into individual piles for each plant type (species), based on unique physical attributes.

11.14.2.6 If an invasive species is recovered, estimate its abundance using USACE/Cornell abundance scale cited in step 11.14.2.4, and identify each plant type/species on form by plant species name or assigned name (“Unknown #1”, “Unknown #2”, etc.)

11.14.2.7 Collect voucher specimen for any unknown, suspected exotic plant, or suspected protected plant species, and place in a labeled zipper storage bag. Each plant should be placed in a separate bag, labeled with the name of the lake, date, and species name/number.

11.14.2.8 Record GPS coordinates on Aquatic Plant Sampling Form.

11.14.2.9 Identify the next sampling point from the GPS by moving 100 m from the present site towards the reference water quality sampling site. Repeat steps 11.14.2.1 through 11.14.2.8

11.14.2.10 Complete step 11.14.2.9 again, moving another 100 m towards the reference site. At least three sites should be sampled with the rake toss between the boat launch and the reference site. If the water becomes too deep to sample three sites spaced 100 m apart, adjust sample spacing along the transect to accommodate three equally spaced samples within the littoral zone.

11.14.3 Sampling procedures for intensive sites are as follows

11.14.3.1 Three sites will be sampled for aquatic plants during each of the last three intensive site visits in the following order - boat launch (July), major inlet (August), and outlet (September)

11.14.3.2 Identify the GPS coordinates of the reference water quality sampling location.

11.14.3.3 Follow the monthly sampling schedule outlined in step 11.14.3.1 for and repeat steps 11.14.2.2 through 11.14.2.10

11.15 Lake Macrobenthos Sampling using D Nets (STAFF 1)

11.15.1 Lake macrobenthos are sampled using the protocols established for stream macroinvertebrate sampling by the Stream Biomonitoring unit. These procedures are established in SOP# 208-19 Stream Biomonitoring.

11.15.2 Sampling procedures are as follows:

11.15.2.1 Identify sampling point from GPS coordinates and pre-determined sampling grid, at a distance of 10 meters from the shoreline. Verify depth of water is less than 1-meter-deep from visual observation or hand-held depth finder; if depth is greater than 1 meter, move toward the shoreline until the depth drops to 1 meter. Record GPS coordinates on Lake Biomonitoring Sampling Form. Enter water from boat or shoreline.

11.15.2.2 Stand parallel to shoreline. Start timer. Use wader boots to agitate lake bottom, moving a D-framed dip net in a figure 8 pattern perpendicular to the boots just above the lake bottom.

11.15.2.3 Move in a direction for one-meter parallel to the shoreline and continue agitating the lake bottom, sweeping net in figure 8 pattern.

11.15.2.4 After 30 seconds and 1 meter, turn 180 degrees and return to original site within 30 seconds, continuing to agitate the lake bottom and sweep the net in a figure 8 pattern.

11.15.2.5 Splash water on the outside of the net to dislodge any material on the inside of the net.

11.15.2.6 Place the mouth of the D net over a sieve bucket and removal all debris, splashing additional water on the outside of the net to dislodge any additional material. Sieve excess water through the bucket until material is mostly dewatered.

11.15.2.7 If habitat type (sandy, cobble, organic, macrophytes) has not previously been sampled at the lake, remove material from bucket and place into labeled 1-qt. polyethylene field collection bottles (labeled with lake name, site number, substrate type). Repeat steps 11.15.2.2 through 11.15.2.6.

11.15.2.8 If habitat type has previously been sampled, identify aquatic plants at site, keep sample in bucket and go to next sampling site.

11.15.2.9 Upon return to shore, add a small amount of lake water to the bucket to re-suspend material, and sieve excess water to fully mix the sample. Remove approximately ½ to ¾ quart of material from the bucket and place in labeled field collection bottle (labeled with lake name and defined as “composite” sample). Discard remaining

sample from bucket into lake and clean bucket and net as per the procedures outlined in (SOP#103-19 Equipment /Cleaning).

11.15.2.10 Place tape on top of bottle labels.

11.15.2.11 Fill collection bottles to shoulder with ethyl alcohol. Invert bottles to mix sample, decant alcohol from sample through bucket sieve to assure no loss of organisms, and refill bottle to shoulder.

11.16 Lake Macrobenthos Sampling using Petite Ponars (STAFF 2)

11.16.1 Lake macrobenthos are sampled using the protocols established for sediment grab sampling by the DEC contaminated sediment unit. These procedures are established in SOP#207-19 Sediment Sampling.

11.16.2 Sampling procedures are as follows:

11.16.2.1 Identify sampling point from GPS coordinates and pre-determined sampling grid, at a distance of 20 meters from the shoreline. Record depth and GPS coordinates on Lake Biomonitoring Sampling Form.

11.16.2.2 Trip open the petite ponar, and lower to within approximately one meter from the lake bottom.

11.16.2.3 Let petite ponar free fall to bottom and give a tug on the line to assure that tripping mechanism is engaged.

11.16.2.4 Bring petit ponar to the surface and, after verifying that the sample was collected, place on top of the open end of a sieve bucket.

11.16.2.5 Open gates and release sample into sieve bucket.

11.16.2.6 Sieve excess water to dewater sample.

11.16.2.7 Record sample depth and note any extraneous material (woody debris, aquatic vegetation, mussels, etc) found in sample

11.16.2.8 Move to next sampling site and repeat steps 11.16.2.1 to 11.16.2.7.

11.16.2.9 Upon return to shore, add a small amount of lake water to the bucket to resuspend material, and sieve excess water to fully mix the sample. Remove approximately ½ to ¾ quart of material from the bucket and place in labeled field collection bottle (labeled with lake name and defined as “composite” sample). Discard remaining sample from bucket into lake and clean bucket and ponar as per the procedure in SOP#103-19 Equipment /Cleaning)..

11.16.2.10 Place tape on top of bottle labels.

11.16.2.11 Fill collection bottle to shoulder with ethyl alcohol. Invert bottles to mix sample, decant alcohol from sample through bucket sieve to assure no loss of organisms, and refill bottle to shoulder.

11.17 Exotic Mussel Substrate Sampling

11.17.1 Exotic mussels are sampled using a modification of the protocols established for multiplate sampling established by the DEC stream macroinvertebrate monitoring unit in SOP# 208-19 Stream Biomonitoring. This monitoring may be conducted on lakes susceptible to zebra mussel infestation, based on water chemistry data and nearby infestations, and on lakes to be sampled at least monthly through a DEC monitoring program (CSLAP or LCI).

11.17.2 Sampling procedures are as follows:

11.17.2.1 Attach a plastic cable with a DEC-DOW identification tag to a 3 hole, 4”x8”x16” concrete block.

11.17.2.2 During the first sampling run at a lake, locate a secluded shallow area of the lake close to the boat launch site

11.17.2.3 Drop the concrete block to the bottom of the lake on a hard substrate (cobble or sand), making sure that the float is closely tethered and is on or just below the lake surface. If multiple blocks are available, sink second block in nearby secluded location.

11.17.2.4 Record GPS coordinates and narrative description of sampling site(s).

11.17.2.5 Upon next trip to the lake, return to sampling site(s), slowly retrieve concrete block and place in tray at bottom of the boat.

11.17.2.6 Visually observe all surfaces of the block and remove any mussels or related organisms. Use a hand lens to closely observe all surfaces for juvenile or adult mussels.

11.17.2.7 Record description of any removed material on field forms and place mussels or other biomaterials in labeled borosilicate vials (label includes name of lake, date, and location)

11.17.2.8 Re-deploy block(s), making sure float is closely tethered and is on or just below the surface.

11.17.2.9 Upon return to shore, fill vial(s) with ethyl alcohol

11.17.2.10 During last sampling session, remove floats, cables and blocks, and clean as per the procedures in (SOP# 103-19 Equipment Cleaning).

11.18 Sediment Diatom Sampling (STAFF 1)

11.18.1 The lake sediment diatom sampling method is outlined in the 2012 National Lakes Assessment Field Operations Manual (EPA, 2012). An abbreviated description of the method is described below.

11.18.1.1 Collect at least a 45 cm long sediment core from undisturbed sediments at or near the water quality sampling site and section off 2 cm of sediment from the top (at all lakes) and bottom (at natural lakes only) of the core sample for analysis. Sediment from the top will be used for the diatom analysis. Samples from the bottom of the core (natural lakes only) will be used for diatom analysis and radiometric dating analysis. In natural lakes, the composition and texture of the bottom will vary from lake to lake and, in some lakes, it will be impossible to get a 45 cm core because the bottom is too rocky, the sediments are too dense, or, if it is a shallow lake, there are macrophytes covering the bottom. It is essential that the GPS coordinates be recorded. If you collect a core less than 45 cm long on your first try, move to another location near the index site to find an area with a softer bottom. In addition, you can experiment with getting improved penetration by adding additional weight (if available) to the corer, and/ or by releasing the corer further above the sediments. If a core of at least 45 cm cannot be collected from natural lakes after attempting at least three locations, process the last core that you obtained. The procedures for collecting and processing sediment cores are presented below. If you collect a core longer than 45 cm long and there is water on top of the sediment core, this will be acceptable for use (see Figure 1).

11.18.1.2 Wear gloves at all times during sample collection to protect yourself from any potential contaminants in the sediments, and to prevent contamination of the sample. If the bottom has been disturbed during the initial depth determination or for any other reason, move at least 5 m to take the core. It is critical that the corer strikes undisturbed surface sediments.

11.18.1.3 Insert the core tube into the sampling housing apparatus and tighten the hose clamp screws to secure the tube. Ensure the messenger is attached to the sampler line. Set the release mechanism.

11.18.1.4 Slowly lower the corer through the water column until the bottom of the core tube is just touching the sediment surface. Raise the corer

1 m and while maintaining a slight tension on the line, lower the corer allowing it to settle into the bottom substrate. Immediately after the corer drops into the sediments, maintain line tension to prevent the corer from tilting and disturbing the core sample. [Keep in mind that the goal is to obtain a core 45 cm in length.

11.18.1.5 If this core length is not obtained the first time, the operation might need to be repeated at a new location using additional weights on the corer (if available) and/or a greater release height in order to improve penetration and obtain a longer core. If the core length exceeds the length of the core tube, as evidenced by the lack of a discernable sediment water interface (Figure 1), the operation will need to be repeated at a new location using less weight on the corer and/or a shorter release height.

11.18.1.6 Trip the corer by releasing the messenger weight so that it slides down the line. Keeping the line vertical and keeping tension on the line will help ensure that the messenger reaches the sampler and trips the mechanism.

11.18.1.7 Slowly raise the corer back to the surface, keeping the bottom of the core tube under the water.

11.18.1.8 While keeping the bottom of the core tube under water, reach under the surface and plug the bottom of the corer with a corer tube plug. To do this without disturbing the water-sediment interface, you cannot tilt the corer more than 45 degrees. (Note: core tube plugs are easily lost. Be sure to have spares available at all times).

11.18.1.9 Keeping your hand under the corer tube plug, raise the corer into the boat in a vertical position.

11.18.1.10 Measure the length of the core to the nearest 0.1 cm and record the interval on the Sample Collection form and on the two sample labels.

11.18.1.11 Determine the intervals by: a. Top Slice Interval: 0 to 2 cm b. Bottom Slice Interval: Calculate using the formula where L equals total length: (L-2) to (L-4) cm 1.

11.18.1.12 Put gloves on. Prepare containers and attach the labels to two plastic containers (for diatoms). Record the Site ID, date, and collection intervals on sediment core sample labels.

11.18.1.13 Detach the core tube from the corer. One crew member should hold the sampler in a vertical position while the second person dismantles the unit.

11.18.1.14 Position the extruder under the corer tube plug at the base of the coring tube. Supporting both the core tube and the extruder in a vertical position, slowly lower the coring tube onto the extruder until the sediment is approximately 1 cm below the top of the tube.

11.18.1.15 Remove the water above the sediment core by using a syringe with tube so that the surface sediments are not disturbed.

11.18.1.16 Secure the sectioning stage onto the top of the coring tube. Place the Plexiglas sectioning tube (marked with a line 2 cm from the bottom) on the stage directly over the coring tube.

11.18.1.17 Slowly extrude the sediment core into the sectioning tube until the top of the sediment reaches the 2 cm line on the sectioning tube (Figure 1). Slide the sectioning tube onto the flat part of the stage and scrape the top 2 cm section of sediment into a labeled clean plastic screw top container.

11.18.1.18 Before collecting the bottom section, remove the stage and sectioning tube and rinse in lake water. Also rinse the spatula, gloved hands and any other implements that have come in contact with the sediment. This procedure prevents contamination of the bottom sediment layer with diatoms from the top portion of the core. This step is critical because a small amount of sediment contains millions of diatoms that would contaminate the population structure needed to compare environmental conditions depicted by top and bottom core samples.

11.18.1.19 Continue extruding the sample, discarding the central portion of the sediment in the tube, until the top of the corer tube plug is approximately 5 cm from the top of the coring tube (Figure 1).

11.18.1.20 Rinse any sediment from your gloved hands. Re-affix the sectioning stage and sectioning tube to the top of the coring tube.

11.18.1.21 Extrude the sample into the sectioning tube until the top of the stopper reaches a point 4 cm from the top of the tube. Section the extruded sediment (approximately 1 cm) and discard. This operation results in exactly 4 cm of sediment remaining in the core tube.

11.18.1.22 Rinse the sectioning tube with lake water. Without removing the sectioning stage from the coring tube, slightly tilt the tube and wash the stage with a small amount of water from a squirt bottle. Make sure the rinse water runs off the stage and not into the coring tube with sediment.

11.18.1.23 Extrude the sample until the top of the sediment is at the 2 cm mark on the sectioning tube. Slide the sectioning tube onto the flat part of the stage.

11.18.1.24 Transfer 1/3 of the top 2cm sediment slice into a labeled plastic screw top container for diatom analysis and transfer the other 2/3 of the slice into a separate labeled plastic screw top container for radiometric dating analysis. Discard the unused 2 cm remaining in the sectioning tube.

11.18.1.25 Samples should be kept on ice and placed in a freezer in room 614 when returning to the DEC lab. Samples can be kept frozen indefinitely until analyzed.

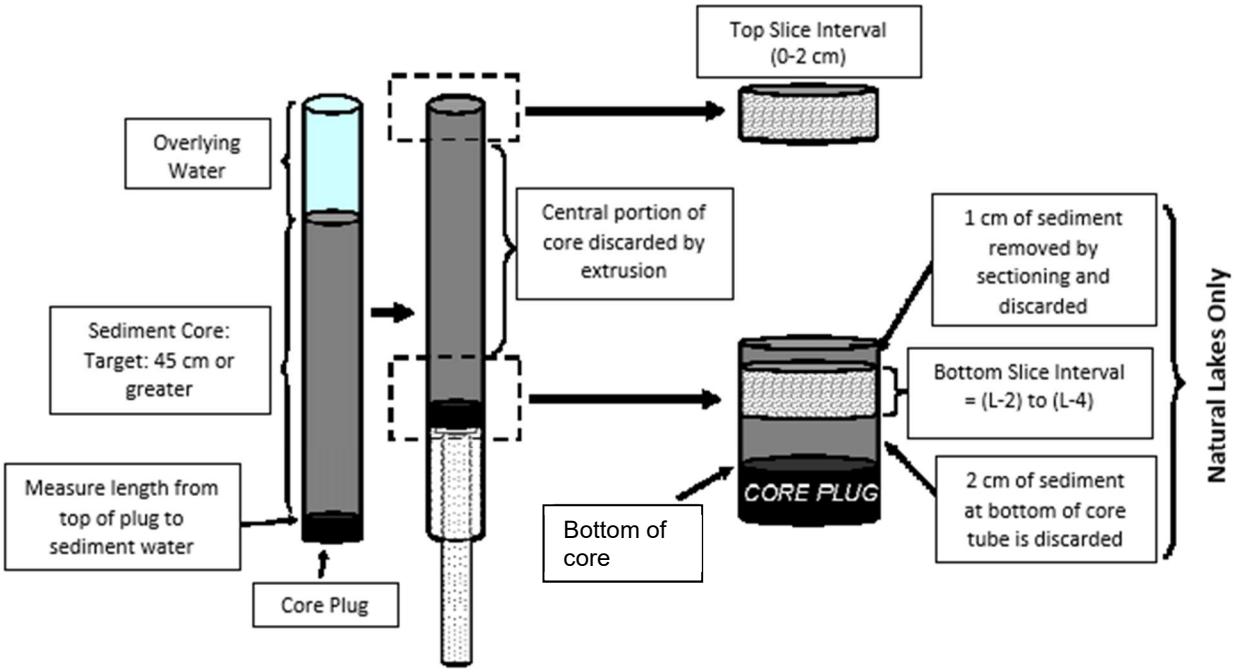

Figure 1: Gravity core and sediment sample schematic.

12 Sample Handling and Preservation

12.1 COVID19 Modification: all sample processing will be completed by STAFF 1.

12.2 General procedures for handling and preserving samples are provided in SOP# 101-20 Sample Handling, Transport, and Chain of Custody. Specific sample handling and preservation procedures for lake samples are discussed below.

12.3 Sample Handling Equipment and Materials

Equipment needed for handling and processing of surface water samples includes all or an appropriate subset of the following:

12.3.1 Compositing container - split churner, integrating carboy, or (other) integrating compositing container

12.3.2 Wear gloves when handling containers fixed with preservative

12.3.3 Filtration apparatus (funnel, centerpiece, receiving flask, hand-operated or compressed air field vacuum pumps, and graduated cylinders)

12.3.3.1 Filters - 0.45µ membrane for inorganics fractionation and chlorophyll *a* (chloroform-methanol method).

12.3.3.2 Filters - 0.45µ glass fiber for organics and chlorophyll *a* (acetone method)

12.3.4 Containers

12.3.4.1 Polysulfonate bottles (organic and inorganic samples)

12.3.4.2 Amber glass bottles (organic samples, HABs samples)

12.3.4.3 Clear plastic bottles (chlorophyll *a* filters)

12.3.4.4 50 mL polypropylene centrifuge vials (zooplankton, chlorophyll *a* filter, and raw water samples for un-extracted chlorophyll *a* analysis)

12.3.4.5 10 mL polypropylene centrifuge vials (cyanotoxins)

12.3.4.6 "Flip-top" in-line filtration apparatus (soluble reactive phosphorus)

12.4 Reagents - except where noted, all aliquots prepared and provided by the laboratory as per the most updated NYSDEC Prescribed Analytical Protocols (PAP):

12.4.1 H_2SO_4 for preserving nutrient samples- preserve to pH <2

12.4.2 HNO_3 for preserving metals samples- preserve to pH <2

12.4.3 Acid Lugols solution for preserving phytoplankton tows- add sufficient solution to stain sample

12.4.4 Carbonated Water (standard stock) and Formalin-Rose Bengal solution for preserving zooplankton tows- fill zooplankton sample vial to within

one inch of lid with carbonated water, top off with formalin-rose Bengal solution

12.4.5 5 mg/l $MgCO_3$ solution for preserving chlorophyll *a* samples- 0.5ml per 20 sq.cm of filter area- preservative applied directly to the filter prior to filtration

12.5 Harmful algal bloom samples

12.5.1 Prior to shipping, the cooler is carefully inspected. Broken and/or leaking coolers or outer cardboard boxes are replaced.

12.5.2 Place HAB samples directly into cooler ensuring that the bottles are caps are tight to prevent leaks.

12.5.3 Reusable ice packs are used to keep the sample chilled during shipping, and coolers are packed with non-paper packing materials to minimize sample bottle(s) shifting during shipment.

12.5.4 The laboratory chain of custody sheets and other forms are placed on top of cooler lid, along with notes to the laboratory reporting any equipment or supply needs.

12.5.5 The cooler is sealed with packing tape, and affixed with a pre-paid, pre-addressed UPS label, and the cooler is delivered to the nearest UPS vendor

12.5.6 Upon receipt at the laboratory, HAB sample bottles are removed, checked against the Chain of Custody form, logged, and kept preserved until analysis.

12.6 Preparation of Compositing Containers (Split Churner or Integrating Carboy)

12.6.1 Compositing containers are used to composite discrete samples, samples collected through integrated tubes, or to composite discrete samples prior to transfer to sample containers. Samples collected from Kemmerer or Van Dorn bottles, or from integrating tubes, should be transferred into compositing containers prior to distributing into sample aliquots to minimize differences between sub-strata within the bottles.

12.6.2 Sample transfer occurs as follows:

12.6.2.1 Regardless of the sample collection method used, collect enough water to sufficiently fill all sample containers plus an additional 1-2 liters to allow for proper mixing in churn or mixing carboy. If using mixing churn, position mixing disk properly in churn and place lid on churn.

12.6.2.2 When pouring samples, establish a uniform churning rate of about 9 inches per second for 10-15 seconds. Churning disk should touch the bottom of the tank, but not break the surface of the water. If using a mixing carboy or compositing bottle, make sure valves or lids are closed and invert carboy or container five times.

12.6.2.3 Open spout and flush with sample. Place the subsample containers under the spout or open mouth of the compositing bottle. Rinse untreated sample bottles with ambient water before filling. If using a mixing churn, churn at a uniform rate for several strokes, then open the valve while continuing to churn and fill each container. If using a mixing carboy or compositing container, repeat this step after each subsample container is filled.

12.6.2.4 Discard any remaining sample water in the churn splitter or carboy. After the last sample at each sampling location, and at the end of each day, rinse the churn/carboy/compositing container thoroughly with distilled/deionized water and discard rinse water. If using a mixing churn, add a liter or so of distilled/deionized water to keep the churn from drying out.

12.7 Sample Preparation and Preservation (Preserved/Unprocessed Samples)

12.7.1 Follow the steps below once the sample has been collected into the compositing churn.

12.7.2 If sample bottles are not acid-preserved (as provided by the laboratory) or pre-preserved, transfer a small amount of water from the compositing container or collection device into the sample bottle and cap, and invert to completely line the inner walls of the bottle. Remove water after acclimating the sample bottles. Repeat two times.

12.7.3 Transfer the sample(s) from the compositing container into suitable labeled sample containers.

12.7.4 Preserve the sample or use pre-preserved sample bottles, when appropriate.

12.7.5 Cap container and then place into a zipper storage bag. If the latter is unavailable, use plastic bags and secure closure with tape.

12.7.6 Load all sample containers into pre-chilled cooler(s) ensuring that bottles are not totally immersed in ice.

12.7.7 Record all pertinent data in the site logbook and on a field data sheet.

12.7.8 Complete the chain-of-custody form, place it into a seal zipper storage bag, and attach it to the top of the cooler with tape.

12.7.9 Rinse all sampling equipment with distilled or deionized water prior to the collection of additional samples.

12.8 Sample Preparation and Preservation (Preserved/Filtered Samples)

12.8.1 Filtration may be required to remove interferences in the analytical procedures required for the desired analytes or to determine soluble or suspected fractions of whole analytes. For most of these analyte fractions, a sufficient volume must be collected to adequately fill a sampling bottle. However, for some analytes, such as chlorophyll *a*, for which the retained portion on a filter is analyzed, a known and specified volume must be measured prior to filtration. The required volume is a function of the biological (algal) productivity of the ecological system being monitored, the sensitivity of the analytical instrumentation, and the precision by which sample volume can be measured. Sample volume required for chlorophyll *a* analyses is specified by the laboratory and identified in the QAPP for each program. Filtration apparatus used for lake sample processing include funnel/centerpiece/flasks equipped with vacuum pumps which are either hand or motor operated.

12.8.2 The funnel filtration is performed as follows:

12.8.2.1 For reusable polypropylene filtration (hand operated vacuum pump) apparatus, rinse the funnel and receiving flask with enough distilled water to remove any potential contaminants. If a motor operated vacuum pump is used, place a pre-labeled aliquot bottle (corresponding to the required filtration analysis) into the receiving flask

12.8.2.2 Place the centerpiece on the rinsed receiving flask of the filtration unit.

12.8.2.3 Using clean forceps, remove a single filter from the sterile filter storage container and place on the centerpiece.

12.8.2.4 If a reusable polypropylene filtration apparatus (with hand operated vacuum pump) is used, thread the cleaned funnel on top of the receiving flask, making sure the funnel is properly threaded to maintain a vacuum.

12.8.2.5 Transfer the sample(s) from the compositing container into a rinsed graduate cylinder or directly into the funnel. Measure sample volume, as determined by the laboratory, from the bottom of the meniscus.

12.8.2.6 Slowly transfer the sample from the graduated cylinder into the funnel of the filtration unit, making sure not to spill or splash any of the sample.

12.8.2.7 Draw a vacuum into the receiving flask by squeezing the hand-operated vacuum pump five times, making sure the pressure on the pump gage does not exceed 10 inches of mercury. For the power pumps this equates to 33.86kpa or 254mmhg.

12.8.2.8 Maintain sufficient vacuum to draw entire sample through the filter

12.8.2.9 Once filtration is completed, release pressure and transfer the sample from a port on the receiver flask into suitable, labeled sample container.

12.8.2.10 Preserve the sample or use pre-preserved sample bottles, when appropriate.

12.8.2.11 Cap container and then place container into zipper storage bag. If the latter is unavailable, use plastic bags and secure closure with tape.

12.8.2.12 Load all sample containers into pre-chilled cooler(s) ensuring that bottles are not totally immersed in ice.

12.8.2.13 Record all pertinent data in the site logbook and on a field data sheet.

12.8.2.14 Complete the chain-of-custody form and laboratory submission form.

12.8.2.15 Rinse all sampling equipment with deionized or distilled prior to the collection of additional samples.

12.9 Sample Storage and Transit

12.9.1 Samples should be shipped immediately or stored at 4°C (except as noted below) until transit can be initiated. Sample holding times must be within the analyte-specific times provided in 40 CFR Part 136.

12.9.2 Field sample preservation- temperatures and preservatives- are established by the sampling laboratories and should be completed prior to sample transit.

12.9.3 Once preserved, plankton samples can be stored at room temperature until analysis.

12.9.4 Sample transit should be avoided at times for which sample transit is suspended (Sundays and holidays using US Postal Service or USPS), unless otherwise addressed via an arrangement with another shipping vendor or analytical laboratory courier service.

13 Trophic State

13.1 The trophic state of a ponded water is calculated using the defined thresholds that follow.

13.2 The trophic state of lakes can be defined functionally or operationally. The functional definition is logistically difficult and costly because it requires measuring the actual biological production (biomass) in the system. The operational definition is preferable because it can be accomplished by measuring a few key indicators related to lake biomass: Chlorphyll a, phosophorus, and secchi. Chlorophyll a is a simple measure of algae biomass; phosphorus is the primary nutrient driving algae growth; and secchi records the changes in water transparency affected by high algae production. Each of these water quality indicators—total phosphorus, chlorophyll a, and Secchi disk transparency—are measured through CSLAP and LCI in each water sampling session and can be used to quantitatively define the trophic state of the lake.

13.3 Table 1 shows the trophic state ranges adopted in New York state and commonly used in other states (Carlson—see below). The small difference between these stems from the desire in New York state to use simple intervals, the recognition that trophic categories represent a continuum rather than clear delineations, and the fact that the New York state boundary between mesotrophic and eutrophic lakes are closely matched.

Table 1: Trophic Ranges for Water Quality Indicators

	Oligotrophic		**Mesotrophic**		**Eutrophic**	
	NYS	Carlson	NYS	Carlson	NYS	Carlson
Phosphorus	<10 ug/L	<12 ug/L	10-20 ug/L	12-24 ug/L	>20 ug/L	>24 ug/L
Secchi Disk Transparency	>5 m	>4 m	2-5 m	2-4 m	<2 m	<2 m
Chlorophyll *a*	<2 ug/L	<2.6 ug/L	2-8 ug/L	2.6-7.3 ug/L	>8 ug/L	>7.3 ug/L

14 Data and Records Management

14.1 All data, times, and field conditions must be documented on field data sheets or within site logbooks with permanent ink. Data sheets are archived as computer files (Microsoft Excel, PDF, or FileMakerPro) within one month of the sampling session and logbooks are transferred to electronic format. Electronic archiving and transmittal of data for federal purposes are outlined in SOP# 102-11 Data Handling and Archival.

15 Quality Control (QC) and Quality Assurance (QA)

15.1 Representative samples are required. In order to collect a representative sample, the hydrology and morphometrics (e.g., measurements of volume, depth, and etc.) of the sampled waterbody should be determined prior to sampling. This will aid in determining appropriate sample locations and depths.

15.2 All field QC samples required in the QAPP must be followed. Field blanks, field spikes, and collection of replicate and split samples are just some of the QC samples that may be requested.

15.3 QC samples

15.3.1 Equipment blanks are obtained by running distilled water through sample collection equipment after cleaning and sample collection. At least one equipment blank is to be collected during each sampling cycle.

15.3.2 Matrix spikes are collected as a normal sample and spiked in the lab by adding a known reference solution to a known volume of an unknown matrix (water) sample. Matrix spike samples are analyzed according to ELAP required frequency at the laboratory. Since most lake monitoring is performed on a large number of lakes, we can rely on the laboratoy frequency to provided matrix spike analyses. If a study contains fewer than ten lakes, a distinct sample must be collected for matrix spike analysis.

15.3.3 Duplicate samples are collected independently of unknown matrix samples, and split samples are obtained from a single sample collection. These are collected during each batch of samples (the more frequent of each weekly submission set or once every twenty samples).

15.4 All instrumentation must be calibrated and operated in accordance with operating instructions as supplied by the manufacturer, unless otherwise specified in the QAPP. Equipment checkout and calibration must occur prior to sampling/operation. Calibration information is recorded on the field sheet or logbook accompanying the field instruments.

15.5 Equipment is checked prior to being taken into the field to assure satisfactory operation. Backup equipment will be available as necessary and practical.

16 References

16.1 NYS DEC DOW. 2019. SOP#101-20 Data Handling, Transport, and Chain of Custody.

16.2 NYS DEC DOW. 2019 SOP#103-20 Equipment Cleaning

16.3 NYS DEC DOW. 2011. SOP#102-20 Data Handling and Archival.

16.4 NYS DEC DOW. 2019. SOP#201-20 Ambient Water Sampling.

16.5 NYS DEC DOW. 2019. SOP#207-19 Sediment Sampling.

16.6 NYS DEC DOW. 2019. SOP#208-19 Stream Biomonitoring.

16.7 NYS DEC DOW. 2019. SOP#212-19 Collection of Harmful Algal Bloom Samples.

16.8 NYS DEC DOW. December 2017. Health and Safety Program and Boating Safety Program.

16.9 NYS DEC DOW.2020. SOP#603-20 Guidance for Field Work During COVID-19 Pandemic.

Appendix 1 (Table 1)

Table 1: Sample Handling Specifications – Water Column

Parameter	Collection Method	Sample Processing	Sample Container	Filling
Alkalinity	Depth Integrated or grab	Composite	Plastic Glass	DO NOT AERATE
Ammonia	Depth Integrated or grab	Composite	Plastic Glass	
Anions	Depth integrated or grab	Composite	Plastic Glass	
Carbon, Total Organic	Depth integrated or grab	Composite	Amber glass	
Carbon, Dissolved Organic	Depth integrated or grab	Composite	Amber glass	Field filtered
Chlorophyll *a*, extracted	Depth integrated or grab	Composite	Clear glass or plastic	Field filtered
Chlorophyll *a*, Un-extracted	Depth integrated or grab	Composite	Plastic, glass	
Coliform-Total & Fecal (including E.coli)	Grab - direct into Sterile container	none	sterile	
Color, true	Depth integrated or grab	Composite	Plastic glass	Field filtered
Specific Conductance	Direct Field Measurement			
Harmful Algal Blooms	Depth integrated or grab	None	Glass	

Parameter	Collection Method	Sample Processing	Sample Container	Filling
Dissolved Oxygen	Direct Field Measurement			
Hardness	Depth Integrated or grab	Composite	Plastic Glass	
Kjeldahl Nitrogen	Depth Integrated or grab	Composite	Plastic Glass	
Metals, Total Recoverable	Depth Integrated or grab	Composite	Plastic Glass	HNO_3 field preserved
Metals, Dissolved	Depth Integrated or grab	Composite	Plastic Glass	HNO_3 field preserved
Mercury, Total	Depth Integrated	Composite	Plastic Glass	
Nitrogen, total	Depth Integrated or grab	Composite	Plastic Glass	
Nitrate-NO_3	Depth Integrated or grab	Composite	Plastic Glass	
Nitrite-NO_2	Depth Integrated	Composite	Plastic Glass	
Oil and Grease	Grab	Do Not Composite	Glass only	DO NOT AERATE
pH	Direct Field Measurement			
Phenolic Compounds	Grab - Steel Bucket	Do Not Composite	Glass only	
Phosphorous, Total	Depth Integrated or Grab	Composite	Plastic Glass	H2SO4 field preserved
Phosphorus, Soluble	Depth Integrated or Grab	Composite	Plastic Glass	H_2SO_4 field preserved
Phosphorus, Soluble Reactive	Depth Integrated or Grab	Composite	Plastic Glass	H_2SO_4 field preserved; field filtered
Solids: Total	Depth Integrated or grab	Composite	Plastic Glass	
Solids: Total Dissolved	Depth Integrated or grab	Composite	Plastic Glass	
Solids Total Suspended	Depth Integrated or grab	Composite	Plastic Glass	
Solids Total Volatile	Depth Integrated or grab	Composite	Plastic Glass	
Sulfate	Depth Integrated or grab	Composite	Plastic Glass	
Toxicity Testing Sample	Depth Integrated or grab	Composite	2 L plastic	

Parameter	Collection Method	Sample Processing	Sample Container	Filling
Turbidity	Depth Integrated or grab	Composite	Plastic Glass	
UV254	Depth integrated or grab	Composite	Plastic Glass	
Volatile Halogenated Organics	Direct Grab or D.O. Sample Bucket	Do Not Composite	Glass, Teflon lined septa	DO NOT AERATE
Sediment Diatom and Dating Samples	Gravity core, discrete	Do not composite	Plastic	