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Subject: EMDF Baseline Surface Water Monitoring by TDEC
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Attachments: [image001.png](#)
[73212 EMDF BaselineSW TDEC 02 07 2023.pdf](#)

Dennis,

Good afternoon. The attached plan was prepared to support TDEC's baseline surface water monitoring at/near EMDF.

TDEC plans to initiate this effort as soon as resource scheduling and access clearing allow. Weather and runoff conditions will also factor into the timing, as our objective is to monitor representative baseflow conditions as opposed to stormwater runoff. Don Gilmore is our point of contact for coordinating the field logistics.

Based on our discussion at the project team meeting last week, TDEC understands DOE does not plan to co-sample. Just let us know if that changes.

As always, let me know if you have any questions.

Thanks,



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Civil & Environmental Consultants, Inc. Scope of Services
325-255 01.1 (WO#071422): EMDF Baseline Surface Water Sampling

Sampling near the proposed Environmental Management Disposal Facility (EMDF) on the Department of Energy (DOE) Oak Ridge Reservation (Site) will be conducted in order to establish baseline surface water conditions prior to construction of the landfill. TDEC has requested Civil & Environmental Consultants, Inc. (CEC) to collect water samples from three flumes (SF-1, SF-3, and SF-6), a spring (Spring D10W), two Bear Creek locations (BCK 7.6 and BCK 8.63), and one upstream NT-10 location (NT10US) to support DOE’s surface water baseline assessment. The seven locations and coordinates, as provided by TDEC, are included in the table below.

Station	ID	Rationale	Longitude	Latitude
Flume 1	SF-1	North Tributary 11 (NT-11) downstream	-84.312827	35.952823
Flume 3	SF-3	North Tributary 11 (NT-11) upstream	-84.313879	35.955775
Flume 6	SF-6	North Tributary 10 (NT-10) downstream	-84.308372	35.955965
Spring D10W	Spring D10W	Upgradient groundwater discharge to surface water ¹	-84.311482	35.959546
Bear Creek Kilometer 7.6	BCK 7.6	Bear Creek downstream (downstream of NT-11)	-84.313963	35.951111
Bear Creek Kilometer 8.63	BCK 8.63	Bear Creek upstream (upstream of NT-10)	-84.306950	35.954
North Tributary 10	NT10US	North Tributary 10 (NT-10) upstream	-84.309	35.959

¹ Spring D10W discharges to stream D-10W, which DOE plans to divert into NT-10.

Task Planning

- CEC Knoxville Environmental Staff participating in the field effort will have the appropriate DOE training and associated “green” badges to support the field effort.
- CEC understands that TDEC will make the required notifications to DOE in order to access the sampling locations, including gaining access to DOE’s on-site Haul Road. Two CEC field personnel will participate in the required haul road training in order to drive a CEC vehicle on the Haul Road. It is anticipated the briefing can be completed in conjunction with the site reconnaissance.
- CEC understands that DOE plans to co-sample during this initial baseline event. As such, DOE may need additional time to coordinate with their selected analytical laboratory and to prepare the bottleware order for this sampling effort. CEC will provide flexibility such that the sampling event can be scheduled at a mutually agreeable date with the various parties.

- A task-specific Job Safety Plan (JSP) will be prepared to outline field efforts, associated risks, mitigation approaches and controls, as well as general emergency response procedures should one arise. The JSP can be provided to TDEC for review and comment upon request.
- An equipment list including personal protection equipment (PPE) and materials required to perform the scope of services will be prepared for the sampling effort. Consideration will be made when selecting equipment and materials to procure PFAS-free materials to lessen the potential for sample contamination from its use. The Michigan Department of Environmental Quality *PFAS Surface Water Sampling Guidance* document is provided as an attachment to this task order as a reference for equipment and materials that are acceptable for use. The list will be provided to TDEC for review and discussion purposes, if requested. Once finalized, CEC will obtain the equipment and materials for use in performing the sampling
- The analytical laboratory (laboratory), as selected in coordination with TDEC, will be engaged to discuss the analytical testing suite and anticipated reporting levels, as well as provide PFAS-free containers. The laboratory will also be engaged to supply PFAS-free water for use in collecting field quality control samples. PFAS-free sample containers and water will be delivered to the CEC Knoxville Office in support of subsequent sampling efforts.
- A site reconnaissance will be performed by CEC field personnel to familiarize itself with the sampling locations. It will also allow CEC to assess access restrictions, site conditions, and plan for the necessary equipment needed to collect samples. It is understood TDEC will be available to accompany CEC on the reconnaissance to share their experience with the sampling locations and to coordinate access.
- It is assumed all sampling will be performed in one day by two CEC Knoxville environmental personnel. Additional time is included in the budget to prepare for the sampling effort and to process and label the sample bottles and prepare the chain-of-custody. CEC will also coordinate pick-up by the lab courier.
- This initial sampling event is intended to be a baseflow sampling event as opposed to stormwater sampling. The budget provided below assumes one sampling event will be performed.

Field Procedures

- The sampling team (CEC, TDEC, and DOE) will meet at the EMWMF office prior to field activities for a safety briefing. CEC will host a safety tailgate talk to review the JSP and Site-specific hazards. The tailgate talk will be documented on a CEC field form and attendees will be asked to sign the form acknowledging the items that were discussed. The DOE contractor will also provide a safety briefing prior to mobilization to the first sampling location.

- Sampling will be broken into two separate “mobilizations” being performed during the same, 10-hour workday. The Site reconnaissance visit will be performed in advance of the sampling mobilizations. PFAS samples across all seven locations will be collected during the first mobilization. All the remaining analytes will be collected during the second mobilization. The separate mobilizations will reduce the potential for cross-contamination during the sample collection effort due to the sensitive nature of the PFAS sampling process.
- PFAS sampling procedures are listed in greater detail in a subsequent section of this scope.
- During the second mobilization, field water quality parameters will be measured at each location. Water quality parameter readings will not be collected during the PFAS sampling mobilization. Water quality parameters to be collected include conductance, pH, temperature, dissolved oxygen, oxidation-reduction potential, and turbidity. CEC anticipates utilizing a Horiba-U52, or similar, to collect the aforementioned water quality parameters. The Horiba U-52 will be calibrated by CEC, or the third-party rental company, prior to deployment in the field. Calibration information will be recorded on a designated CEC field form and provided to TDEC as part of the final deliverable.
- CEC anticipates sampling each location via container immersion, with the exception of the Spring D10W location which may require the use of a peristaltic pump due to its shallow nature. A dedicated transfer container (i.e., with no preservatives) will be utilized at each location to avoid the need to decontaminate sampling equipment. The transfer container will be filled by immersing it within the flowing surface water via a nitrile-gloved hand. Once the dedicated container is filled, CEC will fill the laboratory provided sampling containers in order from the most volatile to least volatile. Care will be taken to prevent the loss of any pre-preserved chemicals within the laboratory sample containers. At Spring D10W, dedicated, PFAS free, sample tubing will be deployed to evacuate spring water into the laboratory containers. If possible, CEC will fill the Spring D10W containers without the use of a transfer container. Should CEC make observations during the Site reconnaissance effort that require a change in sampling methodology, it will be discussed with TDEC. CEC will not change the sampling approach without approval from TDEC.
- In general, sampling will begin at the most downstream location and proceed upstream to reduce potential disturbance. As samples are collected, they will be immediately stored in an iced cooler ice and under proper chain of custody protocols. Sample containers will be appropriately labeled with the sample ID, sample time, date, and requested analytical analysis, at a minimum. PFAS sample containers will be managed and stored separately from the other containers. Samples will be relinquished to a laboratory provided courier for transport services.
- Field notes, including weather conditions, sample times, observation, etc., will be documented in a Rite in the Rain® notebook using a fine point sharpie (per Michigan, 2018 these materials have been determined to be PFAS-free).

PFAS Sampling Method

- CEC personnel will prescribe to the following cautions:
 - Acceptable clothing for field personnel will follow the State of Michigan surface water PFAS sampling guidance.
 - It is understood from the State of Michigan guidance document that there has been no evidence of cross-contamination due to personal care products (PCPs). If any PCPs (including sunscreen or bug spray) are used by sampling personnel the day of sampling, they will be applied at the office, prior to mobilizing to the sampling location, and hands will be washed thoroughly after application. No PCPs will be applied in the field.
 - No food will be consumed during the period of the PFAS mobilization. Any pre-wrapped food will be consumed prior to leaving the office and hands will be washed thoroughly thereafter.
- Out of an abundance of caution, PFAS samples will be collected using the Modified Clean Technique (EPA Method 1669) or the “Clean hands/dirty hands” procedure. Powderless nitrile gloves will be worn and changed frequently throughout sampling (at a minimum, nitrile gloves will be changed at every sampling location prior to commencing).
- As discussed above, PFAS samples are anticipated to be collected via container immersion using a dedicated container at each location. The sample will be directly poured from the transfer container into the laboratory provided sample container. Both the transfer container and sample container will be laboratory certified PFAS-free. At the stream locations, care will be taken to not disturb settled sediment to avoid affecting the turbidity of the sample.
- The Spring D10W location may require the use of a peristaltic pump. The sample tubing will consist of HDPE/LDPE certified PFAS free material. The elastomeric tubing at the pump head will consist of silicone rubber. Alternative materials may be considered if they are certified PFAS free.
- After sampling, the sample bottles will be immediately double bagged in LDPE storage bags (Ziploc®) and placed into the cooler on ice. All PFAS samples will be stored in one cooler with no other sample analytes to minimize any potential for cross-contamination.
- It is assumed decontamination of equipment will not be required since only disposable equipment will be used.
- Fine or ultra-fine point sharpies will be used to label the sample bottles. Labelling will occur only when the lid is firmly secured on the sample bottle and if nitrile gloves are changed after use of the sharpie.

- The trip blank will be provided by the laboratory with certified PFAS free water. A field blank will be prepared at one location during the sampling event using certified PFAS free water. One equipment blank will be prepared using a dedicated transfer bottle to pour PFAS free water into the sample bottle. The trip blank, equipment blank and field blank will be submitted to the laboratory for analytical testing.

Analytical Testing

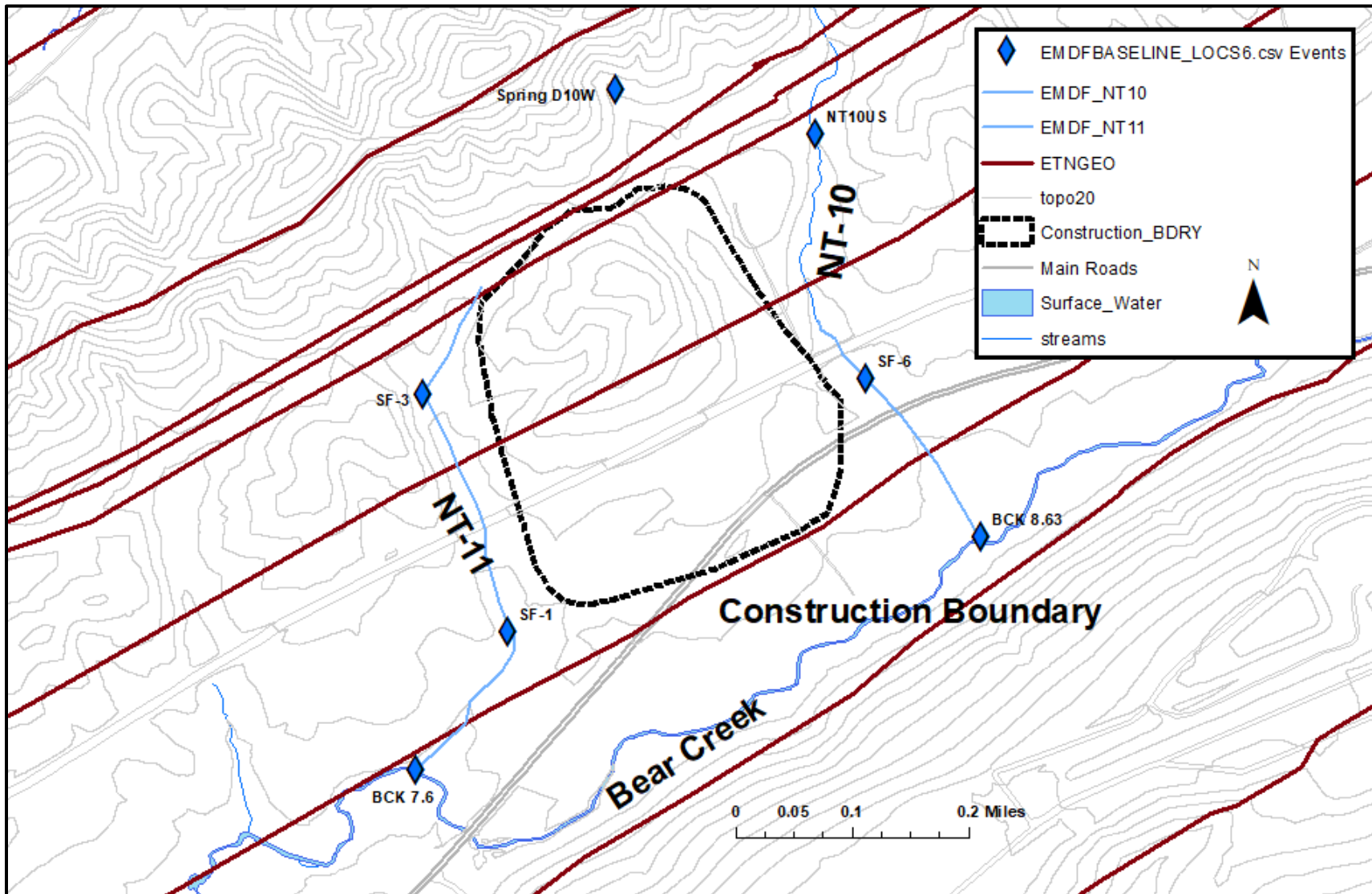
- The analyte list, methods of analysis, and minimum detectable concentrations (MDC) are attached to this scope of services in the form of table. MDCs are to be at or below the most stringent Tennessee Ambient Water Quality Criteria (AWQC), when available. If no AWQC is available for an analyte, the EPA maximum contaminant level (MCL) will be utilized, followed by the EPA secondary MCL, and finally, a lab recommendation based upon their reporting capability. Please note that these methods are subject to change upon request from TDEC. If the laboratory recommendation is used for a given analyte, and the analyte was quantified at that location, then a reinvestigation may be warranted to analyze that contaminant to a more stringent MDC as directed by TDEC.
- The samples will be delivered to the laboratory via courier.
- CEC has budgeted for the following analytical testing:
 - 6 surface water samples to be subjected to the full analytical suite (the feasibility of collecting Whole Effluent Toxicity (WET) testing samples at Flume 3 (upstream of NT-11) and North Tributary 10 (upstream location of NT-10) will be evaluated during the site reconnaissance and reported back to TDEC);
 - The Spring D10W location will be subjected to the full analytical suite with the exception of WET testing;
 - One field duplicate to be subjected to the full analytical suite (the location of the duplicate sample can be specified by TDEC). A WET testing duplicate will not be collected;
 - One trip blank to be analyzed for VOC and PFAS compounds;
 - One equipment blank to be analyzed for PFAS compounds; and,
 - One field blank to be analyzed for PFAS compounds.
- CEC will perform a cursory quality review of the analytical results prior to delivery to TDEC. The quality review will consist of reviewing the laboratory narrative, confirming all analytical results have been received, and reviewing any potential data qualifications. As directed by TDEC, CEC will pass the laboratory reports and EDDs along to TDEC without tabulating.

Reporting

Deliverables for this sampling effort will include:

1. CEC equipment calibration form(s) and field notes
2. Field water quality parameter summary table
3. Laboratory Level IV analytical reports
4. Laboratory provided EDD

TDEC Provided Figure Showing Sampling Locations



Michigan Department of Environmental Quality *PFAS Surface Water Sampling Guidance*

SURFACE WATER PFAS SAMPLING

Guidance

Introduction

This sampling guidance contains the processes, decontamination procedures, and acceptable items and materials for sampling surface water for Per- and Polyfluoroalkyl Substances (PFAS). This guidance will be used to support the sampling objectives and procedures based on the Quality Assurance Project Plan (QAPP) developed prior to any field activities. This guidance assumes staff has basic familiarity with and/or understanding of basic surface water sampling procedures.

NOTE: Review the **General PFAS Sampling Guidance** prior to reviewing this guidance document.

The Michigan Department of Environment, Great Lakes, and Energy (EGLE) intends to update the information contained within this Surface Water PFAS Sampling Guidance document as new information becomes available. The user of this Surface Water PFAS Sampling Guidance is encouraged to visit the Michigan PFAS Action Response Team webpage (michigan.gov/PFASresponse) to access the current version of this document.

PFAS has been detected in surface water in Michigan at concentrations of over 19,000 parts per trillion (ppt). Because PFAS compounds can be analyzed at concentrations in the parts per trillion (ppt) range, precautions must be taken to prevent cross-contamination. Therefore, there is a high possibility of false positives if decontamination procedures are not followed diligently. This sampling guidance covers both the collection of samples from shallow and deep surface water bodies.

This Surface Water PFAS Sampling Guidance discusses the collection of surface water samples and methods to prevent cross-contamination that can occur from:

- Field clothing and personal protective equipment (PPE)
- Personal care products (PCPs)
- Food packaging
- Sampling equipment
- Equipment decontamination
- Filtering of surface water
- Sample collection and handling
- Sample shipment

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NOTE: Additional information about PFAS testing can be found on the Michigan PFAS Action Response Team (MPART) website:

michigan.gov/PFASresponse

1. Potential Sources for PFAS Cross-Contamination

Potential sources for PFAS cross-contamination include items and materials used within the sampling environment, such as sampling equipment, field clothing, personal protective equipment (PPE), sun and biological protection products, personal hygiene, personal care products (PCPs), and food packaging. A detailed discussion about potential sources for PFAS cross-contamination is included in the **General PFAS Sampling Guidance**, which should be reviewed before reading this document. However, a high-level summary is presented in this guidance.

All of the items and materials discussed in each of the EGLE’s PFAS Sampling Guidance Documents are divided into three major groups:

- Prohibited (●) identifies items and materials that should not be used when sampling. It is well documented that they contain PFAS or that PFAS are used in their manufacture.
- Allowable (■) identifies items and materials that have been proven not to be sources of PFAS cross contamination and are considered acceptable for sampling.
- Needs Screening (▲) identifies items and materials that have the potential for PFAS cross-contamination due to a lack of scientific data or statements from manufacturers to prove otherwise. These items and materials are further sub-divided into two categories:
 - o **Category 1:** Items and materials that will come in direct contact with the sample. These should not be used when sampling unless they are known to be PFAS-free, by collecting an equipment blank sample prior to use.
 - o **Category 2:** Items and materials that will not come in direct contact with the sample. These should be avoided, if possible, unless they are known to be PFAS-free by collecting an equipment blank sample prior to use.

● - Prohibited ■ – Allowable ▲ - Needs Screening

Please note that at this time no published research is available that documents the use of various materials and effect on sample results. Therefore, a conservative approach is recommended, and the guidance is based on the collection of multiple environmental samples at various PFAS Sites. Sampling staff should take practical and appropriate precautions to avoid items that are likely to contain PFAS at the sampling site as well as avoid specific items during the sampling event.

A general overview of PFAS contamination sources during sampling can be found in **Section 4.2** of the **General PFAS Sampling Guidance**. Any items or materials utilized that are not identified in this guidance or not discussed in **Section 4.2** should be evaluated as described in **Section 4.2.1**.

Sampling staff should take practical and appropriate precautions to avoid items that are likely to contain PFAS at the sampling site as well as avoid specific items during the sampling event (see below).

1.1 Field Clothing and Personal Protection Equipment (PPE)

A general overview of field clothing and PPE can be found in **Section 4** of the **General PFAS Sampling Guidance**.

As with any field mobilization, it is the responsibility of all personnel to be aware of the physical, chemical and biological hazards associated with a particular site. Personal safety is paramount. The safety of staff should not be compromised by fear of PFAS-containing materials without any scientific basis. Any deviation from this guidance, including those necessary to ensure the health and safety of sampling personnel, should be recorded in field notes and discussed in the final report.

Depending on the project objectives and sampling plan, the collection of surface water samples could be as simple as a grab sample or as complex as a sample collected using a Van Dorn® sampler from a boat. Generally, for surface water sampling, approved field clothing (discussed in **Section 4** of the **General PFAS Sampling Guidance**) is required. Life jackets made of PFAS-free materials should be used. The coatings used on waders are of particular concern during surface water sampling. Ensure the waders are made from PFAS-free materials before use.

- Do not use waders made of Gore-Tex or other known PFAS containing materials.
- Life jackets made of polyethylene foam and nylon shell fabric may be used.
- Waders made of Neoprene or other PFAS-free materials may be used.

NOTE: Special attention should be given to clothing that has been advertised as having waterproof, water-repellant, or dirt and/or stain characteristics. They are likely to have PFAS in their manufacturing.

NOTE: Life jackets may have protective coatings that contain PFAS.

Any field clothing and/or PPE items that might be required for surface water sampling and not discussed in this guidance should be evaluated as described in **Section 4.2.2** of the **General PFAS Sampling Guidance**.

NOTE: Both field clothing and PPE should be kept dust and fiber free. During the sample collection, extra care should be taken so that no dust or fibers can fall into the sample bottle.

Powderless nitrile gloves should frequently be changed any time there is an opportunity for cross-contamination of the sampling including, but not limited to, the following activities:

- Each time sampling equipment is handled.
- Prior to sample collection.

● - Prohibited ■ – Allowable ▲ - Needs Screening

- After handling any sample, including QA/QC samples such as field reagent blanks or equipment rinsate blanks.
- After the handling of any non-dedicated sampling equipment, contact with non-decontaminated surfaces, or when judged necessary by field personnel.
- During and after decontamination of non-dedicated sampling equipment.

1.2 Personal Care Products (PCPs)

A number of sampling guidance documents recommend that personal hygiene and personal care products (PCPs) (e.g., cosmetics, shampoo, sunscreens, dental floss, etc.) not be used prior to and on the day(s) of sampling because the presence of PFAS in these products has been documented (OECD, 2002, Fujii, 2013, Borg and Ivarsson, 2017). However, if the EGLE's sampling SOPs are followed, these items should not come into contact with the sampling equipment or the sample being collected. As of the date of this sampling guidance, cross-contamination of samples due to the use of PCPs has not been documented during the collection of thousands of samples. However, field personnel should be aware of the potential of cross-contamination if the sampling equipment or actual samples would come into contact with these products. The following precautions should be taken when dealing with personal hygiene or PCPs before sampling:

- Do not handle or apply PCPs in the sampling area.
- Do not handle or apply PCPs while wearing PPE that will be present during sampling.
- Move to the staging area and remove PPE if applying personal care products becomes necessary.
- Wash hands thoroughly after the handling or application of PCPs and, when finished, put on a fresh pair of powderless nitrile gloves.

1.3 Food Packaging

PFAS has been used by the paper industry as a special protective coating against grease, oil, and water for paper and paperboards, including food packaging since the late 1950s (Trier et al., 2018). PFAS application for food packaging includes paper products that come into contact with food such as paper plates, food containers, bags, and wraps (OECD, 2002). Pre-wrapped food or snacks (such as candy bars, microwave popcorn, etc.) must not be in the sampling and staging areas during sampling due to PFAS contamination of the packaging. When staff require a break to eat or drink, they should remove their gloves and any other PPE, if worn, in the staging area and move to the designated area for food and beverage consumption. When finished, staff should wash their hands and put on a fresh pair of powderless nitrile gloves at the staging area, before returning to the sampling area.

- Do not handle, consume, or otherwise interact with pre-wrapped food or snacks, carry-out food, fast food, or other food items while on-site during sampling.
- Move to the staging area and remove PPE prior to leaving the sampling and staging areas if consuming food on site becomes necessary.

2. Surface Water Sampling Equipment

Surface water sampling equipment that is also used for non-PFAS sampling such as dippers, Kemmerer[®], or Van Dorn[®] samplers, should be decontaminated prior to collecting PFAS samples, by triple-rinsing with laboratory certified, PFAS-free deionized water before use and between sampling events, to avoid cross contamination. This non-dedicated equipment (equipment used for more than one water body or location) should be verified to be PFAS-free through the collection of an equipment blank prior to use. Surface water sampling equipment can fall into **Category 1** or **Category 2**:

● - Prohibited ■ – Allowable ▲ - Needs Screening

Category 1: Surface water sampling equipment that will come into contact with the surface water sample includes sample containers and various surface water samplers or tubing. Sample containers should be provided by the laboratory and known to be PFAS-free. Any surface water samplers, tubing, or materials that will come into contact with the surface water samples should be screened and known to be PFAS-free. The tubing should always be kept in the original cardboard or bag in which it was shipped. The tubing should always be stored in a clean location free of dust and fibers.

NOTE: As a precautionary action, an equipment rinsate blank should be collected even if the sampling materials are made of materials that are not expected to contain PFAS.

Category 2: Examples of field equipment that do **not** come into contact with the surface water samples include cell phones, GPS receivers, notebooks, clipboards, and multiparameter water quality sonde units. The surface of some of these pieces of field equipment, or the storage boxes in which they are kept, might contain PFAS.

Do not use any equipment that contains any known fluoropolymers including, but not limited to:

- Do not use polytetrafluoroethylene (PTFE), that includes the trademark Teflon® and Hostaflon®, which can be found in many items, including but not limited to the lining of some hoses and tubing, some wiring, certain kinds of gears, and some objects that require the sliding action of parts.
- Do not use Polyvinylidene fluoride (PVDF), that includes the trademark Kynar®, which can be found in many items, including but not limited to tubing, films/coatings on aluminum, galvanized or aluminized steel, wire insulators, and lithium-ion batteries.
- Do not use Polychlorotrifluoroethylene (PCTFE), that includes the trademark Neoflon®, which can be found in many items, including but not limited to valves, seals, gaskets, and food packaging.
- Do not use Ethylene-tetrafluoro-ethylene (ETFE), that includes the trademark Tefzel®, which can be found in many items, including but not limited to wire and cable insulation and covers, films for roofing and siding, liners in pipes, and some cable tie wraps.
- Do not use Fluorinated ethylene propylene (FEP), that includes the trademarks Teflon® FEP and Hostaflon® FEP, and may also include Neoflon®, which can be found in many items, including but not limited to wire and cable insulation and covers, pipe linings, and some labware.
- Do not use low density polyethylene (LDPE) for any items that will come into **direct contact** with the sample media. LDPE can be found in many items, including but not limited to containers and bottles, plastic bags, and tubing.
- ▲ **However**, LDPE may be used if an equipment blank has confirmed it to be PFAS-free. LDPE does not contain PFAS in the raw material but may contain PFAS cross-contamination from the manufacturing process.
- LDPE bags (e.g. Ziploc®) that **do not** come into direct contact with the sample media and do not introduce cross contamination with samples may be used.
- Use materials that are either made of high density polyethylene (HDPE), polypropylene, silicone, or acetate.
- Use only powderless nitrile gloves (which can be found at some hardware and major retail outlets).

Note: Manufacturers can change the chemical composition of any product. As a result, all materials that will come into direct contact with the sample media should be tested to confirm they are “PFAS-free”, i.e. will not contaminate samples at detectable levels. **There is no guarantee that materials in the ‘Allowable’ category will always be PFAS-free.**

- Transport loose sampling supplies such as tubing, pipette tips, transfer pipettes, or sampling containers in their original packaging or LDPE bags (e.g. Ziploc®).
- Store sampling supplies in a clean location free of dust and fibers.
- ▲ Latex gloves should be screened before use.
- ▲ Post-It® Notes should be screened before use.

NOTE: Depending on the project objectives, boats might be required to be used during surface water sampling. Boats might have various parts that may contain PFAS, including protective water repellent coatings. When boats are used on rivers, samples should always be collected on the upgradient side of the boat.

Surface water samples can be collected at wadeable and non-wadeable locations: At locations deeper than 1 foot in depth, the sub-surface grab sample should be collected approximately 6-inches below the water surface. At locations that are less than 1 foot in depth, the sub-surface grab sample should be collected at the mid-point of the water column and caution should be taken to avoid contaminating the sample with sediment and/or surface scum. If the depth is too shallow to properly submerge the sample collection apparatus, a surface sample should be collected with care not to contaminate the sample with sediment and/or surface foams. Any departure from the 6-inch [0.5 foot] depth sampling rule for PFAS should be noted in the field notes.

A depth-integrated sample can be collected using a simple weighted bottle constructed to allow gradual water inflow (e.g., chlorophyll sampler). Depth-specific samples can be collected using a Van Dorn® or Kemmerer® sampler. The decision to collect a depth-integrated sample, or depth-specific sample should be made by the project leader and based on the project's objectives.

Collecting samples at the surface of a water body or solely sampling the surface microlayer is not recommended if the data are to be compared to Michigan's Part 4, Rule 57 Water Quality Values (WQVs). These values include the use of Bioaccumulation Factors (BAFs). BAFs are used to determine the relationship between the concentration of the contaminant in fish fillets to the concentration of the contaminant in the water body. Since fish are not routinely exposed to the microlayer, comparing surface concentrations of PFAS to the WQVs is not scientifically defensible. At this time, EGLE is unaware of any study that has derived a fish tissue BAF based on surface and/or microlayer PFAS concentrations.

2.1 Container Immersion

Two types of immersion sampling equipment are available for surface water sampling: extension rods and submersible devices. Extension rods can be used to immerse the actual sample container, different types of beakers, or peristaltic pump tubing into the surface water. Submersible devices (i.e., Kemmerer Bottle, Van Dorn Sampler) are fully immersed into the surface water using a rope.



2.1.1 Extension Rods

The most common extension rods are telescoping or swing samplers. Both types of sampling equipment are very similar in design and concept, and both facilitate the immersion of either the sampling container or various beakers or scoops. Lists of various extension rod designs are provided below:

- Pendulum or angular beaker.
- Fixed scoop.
- Fix or rotatable head container holder.

A peristaltic pump can also be used with extension rods by attaching the tubing to the extension rods and immersing both the rods and the connected tubing to the desired depth in the surface water.

- Use only sample collection equipment, tubing, beakers, and/or scoop materials that are known to be PFAS-free such as stainless steel, HDPE, polyvinyl chloride (PVC), polypropylene, acetate, or silicone.
- Extension rods made of materials such as aluminum that has been identified as being PFAS-free can be used.

A specialized extension rod that features a telescoping design for the handle could also be used as a subsurface grab sampler. The sample is collected using a cable from the handle, which has a ring that can be opened for the sample collection after the desired depth has been reached.

2.1.2 Submersible Devices

The most common submersible devices being used are weighted-bottle samplers, Kemmerer Bottles, or Van Dorn Samplers. These devices are primarily used when the samples are collected at depths greater than 5 feet from a boat and/or structure such as a bridge or pier. All submersible devices are submerged in the surface water using a rope.

NOTE: Careful evaluation of all submersible samplers' parts should be done. Any parts that might contain PFAS should be replaced with PFAS-free materials. Equipment rinsate blank samples should be collected to make certain the sampler is PFAS-free.



The weighted bottle sampler is typically constructed of a 1-liter Nalgene Tritan® bottle fixed inside a metal cage fitted with a weight below the bottle. This sampler is suspended vertically, which is ideal for sample collection at road or bridge crossings. The bottle is typically lowered to approximately 6-inches below the water surface. The sampler can also be fitted with a depth-integrating cap to allow the bottle to slowly fill while lowering the apparatus to the bottom of the water body.

The Kemmerer Bottle sampler is typically constructed of a stainless-steel tube with polyurethane end seals that can collect a total sample volume of 1.2 liters. The Kemmerer Bottle is not ideal for the collection of samples close to the surface, as the tube is immersed vertically in the water.

The Van Dorn® bottle sampler is typically constructed of 1-liter transparent acrylic tube with two end stoppers. The sampler is suspended horizontally, which is ideal for the sample collection in shallow water bodies as well as sampling at depth.

When submersible samplers are used, the following recommendations should be followed:

- Do not use any sampling bottle with Teflon end seals.
- Use a Kemmerer® Bottle made of stainless steel with polyurethane end seals.
- Use a Van Dorn® bottle sampler that uses stoppers made of PFAS-free materials.
- Use nylon line, stainless steel cable, or line or wires made of PFAS-free materials for sample collection.
- Use tubing for the sampling ports made of HDPE, polypropylene, silicone, PVC, or other PFC-free materials.

2.2 Direct Sampling



For surface water samples collected near the shore, the direct method can be used to collect the water samples directly into the sample container.

- Do not sample without powderless nitrile gloves.
- Never place the sample cap directly on the ground or boat deck during sampling.

- Use powderless nitrile gloves
- Hands should be well washed
- Use HDPE sample containers with Teflon®-free caps, provided by the laboratory.
- If sample containers that are known to be PFAS-free are not available, the sample container and lid should be rinsed with water that is known to be PFAS-free at least 3 times prior to collecting the sample.
- If samples are collected while wading in the water body, the container should be immersed inverted and upstream of the collector.
- If samples are collected from a boat, the containers should be submerged upstream of the boat.

NOTE: Unless specifically required by the project objectives, surface water samples should **not** be taken at the top layer of the water body or of surface scums. PFAS are expected to accumulate at the surface water air interface or be present in the surface runoff, so samples taken at the surface are likely to result in high biased results that are not representative of the bulk surface water.

2.3 Pipette Sampling



For surface water samples to be analyzed for PFAS using EPA SW-826 method 3512, the pipette method can be used to collect the sample. A pipette capable of collecting 10 milliliters (mL) of sample (e.g., the Gilson Pipetman Classic P10mL) should be used. The pipette tip should be constructed of PFAS-free material (e.g., polypropylene) and should be changed between samples. 5mL of sample should be dispensed into two laboratory-provided PFAS-free sample containers.

- Use powderless nitrile gloves
- Hands should be well washed
- Use polypropylene (or another PFAS-free equivalent material) pipette tips.
- Dispense the sample into the laboratory provided sample container(s).
- If samples are collected while wading in the water body, the pipette tip should be immersed upstream of the collector.
- If samples are collected from a boat, the pipette tip should be immersed upstream of the boat.
- ▲ Other pipette materials may be used if an equipment blank has confirmed it to be PFAS-free

3. Equipment Decontamination

Field sampling equipment that is used at multiple sites or sampling locations (non-dedicated equipment) could become contaminated with PFAS.

The following should be considered when decontaminating any equipment that contacts the sampling media:

- Do not use Decon 90®.
- Laboratory supplied PFAS-free deionized water is preferred for decontamination.
- Alconox®, Liquinox®, and Citranox® can be used for equipment decontamination.
- Sampling equipment can be scrubbed using a polyethylene or Polyvinyl chloride (PVC) brush to remove particulates.

● - Prohibited ■ – Allowable ▲ - Needs Screening

- Decontamination procedures should include triple rinsing with PFAS-free water.
- Commercially available deionized water in an HDPE container may be used for decontamination if the water is verified to be PFAS-free.
- ▲ Municipal drinking water may be used for decontamination purposes if it is known to be PFAS-free.
- ▲ Laboratory deionized water may be used for decontamination purposes if it is known to be PFAS-free.

4. Sample Collection and Handling

A preferred sampling sequence should be established prior to any sampling event to reduce the risk of cross contamination. In general, the sampling sequence should begin in areas expected or known to be least contaminated, proceeding to anticipated areas or identified to be most contaminated. If analytical results from past sampling events are available, the sampling sequence can be readily determined.

However, for many PFAS investigation sites, no PFAS sampling has been conducted. In these cases, all site information on possible PFAS uses and potential PFAS migration patterns (e.g., upgradient, downgradient) from PFAS sources at the site should be reviewed prior to the sampling event to help establish the sampling sequence.

When collecting and handling surface water samples:

- Do not insert any materials inside the sample container.
- Dust and fibers must be kept out of sample containers.
- The sample cap should never be placed directly on the ground during sampling. If sampling staff must set the sample container cap down during sample collection and a second member of the sampling crew (wearing a fresh pair of powderless nitrile gloves) is not available, set the cap on a clean surface (cotton sheeting, HDPE sheeting, triple rinsed cooler lid, etc.).
- Regular/thick size markers (Sharpie® or otherwise) are to be avoided; as they may contain PFAS.
- Fine or Ultra-Fine point Sharpies® may be used to label the empty sample container while in the staging area provided the lid is on the sample container and powderless nitrile gloves are changed following sample container labeling.
- Ballpoint pens may be used when labeling sample containers. If ballpoint pens do not write on the sample container labels, preprinted labels from the laboratory may be used.
- Hands should be well washed and gloved.
- Use HDPE, or polypropylene sample containers with Teflon®-free caps, provided by the laboratory.
- Containers should only be opened immediately prior to sampling.
- Containers should be capped immediately after collecting the sample.
- Samples should be double bagged using resealable low density polyethylene (LDPE) bags (e.g., Ziploc®).
- Adequate blanks should be collected to ensure no contamination in the field setting, laboratory setting, or sampling equipment has occurred. A trip blank should be prepared by the laboratory and should remain with the samples until analysis. A field blank should be prepared at one chosen field site by the sampling crew for each project and remain with the samples until analysis. Equipment blanks should be prepared for each sampling apparatus that is used to collect surface water samples unless a direct collection method was used.
- Follow any guidance or requirements in the PFAS analytical reference method that will be used for testing samples, for sample collection, storage, preservation, and holding times.

● - Prohibited ■ – Allowable ▲ - Needs Screening

If a published testing method is not used, and in the absence of formal United States Environmental Protection Agency guidance for PFAS sample storage, the documentation in USEPA Method 537 Rev. 1.1 should be used as a guide for thermal preservation (holding temperature), and holding times for surface water or other samples. Samples must be chilled during storage and shipment, and must not exceed 50°F (10° C) during the first 48 hours after collection.

NOTE: USEPA Method 537 Rev. 1.1 was developed for the analysis of finished drinking water samples only. It was not designed for testing surface water or other matrices that could cause significant interferences to the method.

Surface water samples should be extracted as soon as possible but must be extracted within 14 days (EPA Method 537 Rev. 1.1, EPA Method 8327 Rev. 0.). Extracts must be stored at room temperature and analyzed within 28 days after extraction if analyzed by EPA Method 537 Rev. 1.1. Extracts must be stored at room temperature and analyzed within 28 or 30 days after extraction if analyzed by EPA Method 537.1 or EPA Method 8327, respectively.

5. Filtering of Surface Water

Since PFAS can sorb to particulate matter, unfiltered samples may result in high-biased results. PFAS are known to adsorb to various filters. As a result, filtering of surface water samples prior to delivery to the lab should be avoided unless called for in the project data quality objectives. To reduce the need for filtering, samples should be collected with as minimal disturbance to sediments as possible.

If it is known beforehand that samples will need to be filtered the procedure should be discussed with the laboratory and sample handling methods and responsibilities should be described in the sampling workplan and QAPP.

NOTE: It is recommended that filtering of the samples should **only be performed in the laboratory** in order to reduce the possibility of cross contamination.

If the surface water is highly turbid at the time of collection, it is recommended to allow the sample to settle until all visible suspended material has settled on the bottom of the container. The sample should then be decanted into a secondary sample container prior to shipment to the laboratory. This turbidity should be noted in the field notes.

The following recommendations should be used when considering filtering of the samples:

- **Field filtration of the sample is generally not advised.**
- ▲ If filtering is absolutely necessary, or if specifically requested by staff, the lab, or for other reasons:
 - Do not use any filters that contain any PFAS, such as PTFE filters.
 - Do not use nylon filters.
 - Consider use of a centrifuge in the laboratory to reduce the need for sample filtering.

6. Sample Shipment

When prepping samples for shipping:

- Check the cooler periodically to ensure samples are well iced and at the proper temperature.
- Refresh with wet ice, if needed, double bagged in LDPE resealable storage bags if needed.
- Wet ice should be used to cool and maintain the sample at or below the proper temperature.
- ▲ Chemical or blue ice may be used if it is known to be PFAS-free and it is absolutely certain that the sample is cooled and maintained at or below the proper temperature during collection and through transit to the laboratory.

● - Prohibited ■ – Allowable ▲ - Needs Screening

- Chain of Custody and other forms should be double bagged in LDPE (Ziploc®) storage bags and taped to the inside of the cooler lid.
- The cooler should be taped closed with a custody seal and, if shipping, shipped by overnight courier.
- The laboratory must be contacted prior to shipment to ensure they have staff available to accept the shipment, particularly important for weekend deliveries.
- Samples should be shipped as soon as possible (e.g. overnight) to ensure the samples arrive within the analytical holding time specified by the lab.

Analytical Testing Suite and Level of Quantification

Table 1				
Analyte	Method	Laboratory RL	DQO	Unit
Metals				
Arsenic	6020B	1	10	µg/L
Barium	6020B	1	2000	µg/L
Cadmium	6020B	1	1.8	µg/L
Chromium	6020B	1	100	µg/L
Chromium (VI)	SM-3500CrB-2011	10.0	16	µg/L
Copper	6020B	1	13	µg/L
Lead	6020B	1	5	µg/L
Mercury	EPA-1631E	0.0005	0.05	µg/L
Nickel	6020B	1	100	µg/L
Selenium	6020B	1	20	µg/L
Silver	6020B	1	3.2	µg/L
Thallium	6020B	1*	0.24	µg/L
Uranium (total)	ICPMS (200.8/6020)	0.067	30 ^A	µg/L
Radionuclides^B				
Americium-241	EiChroM AM-01	< 1.0	1.88	pCi/L
Carbon-14	Beta LSC	< 10	0.0753	pCi/L
Cesium-137	EPA-901.1	< 10	0.645	pCi/L
Chlorine-36	Beta GPC	< 50	289	pCi/L
Cobalt-60	Beta GPC	< 10	35.5	pCi/L
Europium-154	Beta GPC	< 10	32.7	pCi/L
Iodine-129	Rad-I-129 by Leps	< 5.0	10.2	pCi/L
Neptunium-237	Alpha Spec	< 1.0	23.4	pCi/L
Plutonium-238	EML Pu-01 Mod	< 1.0	0.0169	pCi/L
Plutonium-239/240	EML Pu-01 Mod	< 1.0	0.0165	pCi/L
Radium-226	EPA 903.0 Mod	< 1.0	0.534	pCi/L
Radium-228	EPA 905.0	< 2.0	10.5	pCi/L
Strontium-90	EiChroM Sr-01	< 2.0	47.9	pCi/L
Technetium-99	EiChroM Tc-01	< 5.0	1000	pCi/L
Thorium-228	EML Th-01 Modified	< 1.0	21.9	pCi/L
Thorium-230	EML Th-01 Modified	< 1.0	84.2	pCi/L
Thorium-232	EML Th-01 Modified	< 1.0	75.3	pCi/L
Tritium	EPA 906.0	< 500	465,000	pCi/L
Uranium-233/234	EPA 905.0 Mod	< 1.0	317	pCi/L
Uranium-235/236	EPA 905.0 Mod	< 1.0	455	pCi/L

Uranium-238	EPA 905.0 Mod	< 1.0	210	pCi/L
VOCs				
TCL VOCs	SW846-8260B	See Table 2	See Table 2	--
Pesticides/PCBs				
4,4'-DDD	8081B	0.04*	0.0031	µg/L
4,4'-DDE	8081B	0.04*	0.0022	µg/L
4,4'-DDT	8081B	0.04	0.0022	µg/L
Aldrin	8081B	0.04	0.00049	µg/L
alpha-BHC	8081B	0.04*	0.026	µg/L
beta-BHC	8081B	0.04	0.091	µg/L
Chlordane	8081B	0.2	0.008	µg/L
Dieldrin	8081B	0.04	0.00052	µg/L
Endosulfan II (b-endosulfan)	8081B	0.04	0.22	µg/L
Endrin aldehyde	8081B	0.04	0.29	µg/L
Heptachlor epoxide	8081B	0.04	0.00039	µg/L
Lindane (gamma-BHC)	8082A	0.04	0.95	µg/L
PCB-1016	8082A	0.2	0.5	µg/L
PCB-1221	8082A	0.2	0.5	µg/L
PCB-1232	8082A	0.2	0.5	µg/L
PCB-1242	8082A	0.2	0.5	µg/L
PCB-1248	8082A	0.2	0.5	µg/L
PCB-1254	8082A	0.2	0.5	µg/L
PCB-1260	8082A	0.2	0.5	µg/L
PCB-1262	8082A	0.2	0.5	µg/L
PCB-1268	8082A	0.2	0.5	µg/L
SVOCs				
TCL SVOCs	SW846-8270D	See Table 3	See Table 3	--
SVOCs (PAHs)	8270SIM	See Table 4	See Table 4	--
Other				
Ammonia Nitrogen, total as N	SM-4500-NH3D-2011	0.100	NE	mg/L
Calcium	6010D	0.5	NE	mg/L
Carbonate	SM 2320B-2011	2	NE	mg/L
Bicarbonate	SM 2320B-2011	1	NE	mg/L
Chloride	EPA-300.0	0.4	250 ^A	mg/L
Cyanide	4500CNE-2016	5	22	µg/L
Hardness as CaCO ₃ (mg/L)	200.7	NE	NE	--
Magnesium	6010D	0.1	NE	mg/L
Nitrate (as nitrogen)	EPA-300.0	100	10,000	µg/L

Nitrate + Nitrite	EPA-300.0	NE	NE	--
Nitrogen, total as N	Calculated	NE	NE	mg/L
Phosphorus, total as P	365.4	0.5	NE	mg/L
Potassium	6020B	100	NE	µg/L
Sodium	6020B	100	NE	µg/L
Sulfate	EPA-300.0	1	250 ^A	mg/L
Total dissolved solids	2540C-2015	2.5	500	mg/L
Total Organic Carbon (TOC)	5310C-2014	1	NE	mg/L
Total suspended solids	2540D-2015	2	NE	mg/L
Whole effluent toxicity - chronic/acute	1000.1 and 1000.2	--	NE	--
Dioxins/Furans				
25 compounds	Method 1613B	See Table 5	1 ^C	pg/L
PFAS				
18 compounds	Method 537.1 Rev2	See Table 6	NE	--
11 compounds	Method 533	See Table 6	NE	--

RL: Reporting Limit

DQO: Data Quality Objective

When not listed, the DQO is determined by the most stringent State of Tennessee Ambient Water Quality Criteria (AWQC)

^A:EPA's Maximum Contaminant Level (MCL) is listed due to no AWQC established for that analyte

^B:For radiochemical data, the DQO was determined by the UCOR document "Development of Fish Tissue and Surface Water Preliminary Remediation Goals for Radionuclides of Interest for the Proposed Environmental Management Disposal Facility, Oak Ridge, Tennessee", Table 1: Surface water PRGs

^C:The AWQC listed is for Total Dioxin, which is calculated by multiplying each dioxin and furan isomer by a toxic equivalent factor, and then summing the concentrations

NE: Not established; no AWQC, MCL, or secondary MCL is established for that analyte

A **highlighted** cell indicates the laboratory MDL does not meet the DQO

*: The laboratory MDL is below the DQO

Table 2			
Analyte	Laboratory RL	DQO	Unit
TCL VOCs via SW846-8260B			
1,1,1-Trichloroethane	1	200	µg/L
1,1,2,2-Tetrachloroethane	1	1.7	µg/L
1,1,2-Trichloroethane	1	5	µg/L
1,1-Dichloroethane	1	NE	µg/L
1,1-Dichloroethene	1	NE	µg/L
1,2-Dichloroethane	1	3.8	µg/L
1,2-Dichloropropane	1	5	µg/L
2-Butanone	20	NE	µg/L
2-Hexanone	5	NE	µg/L
4-Methyl-2-pentanone	5	NE	µg/L
Acetone	20	NE	µg/L
Benzene	1	5	µg/L
Bromodichloromethane	1	0 ^A	µg/L
Bromoform	1	43	µg/L
Bromomethane	2	NE	µg/L
Carbon disulfide	1	NE	µg/L
Carbon tetrachloride	1	2.3	µg/L
Chlorobenzene	1	100	µg/L
Chloroethane	1	NE	µg/L
Chloroform	1	57	µg/L
Chloromethane	1	NE	µg/L
cis-1,2-Dichloroethene	1	NE	µg/L
cis-1,3-Dichloropropene	1	NE	µg/L
Dibromochloromethane	1	60 ^A	µg/L
Ethylbenzene	1	530	µg/L
Methylene chloride	5	46	µg/L
Styrene	1	100	µg/L
Tetrachloroethene	1	NE	µg/L
Toluene	2	1,000	µg/L
trans-1,2-Dichloroethene	1	NE	µg/L
trans-1,3-Dichloropropene	1	NE	µg/L
Trichloroethene	1	NE	µg/L
Vinyl chloride	1	0.25	µg/L
Xylenes (total)	NE	10,000	µg/L

Table 3			
Analyte	Laboratory RL	DQO	Unit
TCL SVOCs via SW846-8270D			
1,2,4-Trichlorobenzene	5	70	µg/L
1,2-Dichlorobenzene	5	420	µg/L
1,3-Dichlorobenzene	5	320	µg/L
1,4-Dichlorobenzene	5	63	µg/L
2,4,5-Trichlorophenol	5	1,800	µg/L
2,4,6-Trichlorophenol	5	14	µg/L
2,4-Dichlorophenol	5	77	µg/L
2,4-Dimethylphenol	5	380	µg/L
2,4-Dinitrophenol	10	NE	µg/L
2,4-Dinitrotoluene	5*	1.1	µg/L
2,6-Dinitrotoluene	5	NE	µg/L
2-Chloronaphthalene	5	1,000	µg/L
2-Chlorophenol	5	81	µg/L
2-Methylnaphthalene	2	NE	µg/L
2-Methylphenol	5	NE	µg/L
2-Nitroaniline	5	NE	µg/L
2-Nitrophenol	5	NE	µg/L
3-Methylphenol	5	NE	µg/L
4-Methylphenol	5	NE	µg/L
3,3'-Dichlorobenzidine	10	0.21	µg/L
3-Nitroaniline	10	NE	µg/L
2-Methyl-4,6-dinitrophenol	10	13	µg/L
4-Bromophenyl phenyl ether	5	NE	µg/L
4-Chloro-3-methylphenol	5	NE	µg/L
4-Chloroaniline	5	NE	µg/L
4-Chlorophenyl phenyl ether	5	NE	µg/L
4-Nitroaniline	5	NE	µg/L
4-Nitrophenol	10	NE	µg/L
Acenaphthene	2	670	µg/L
Acenaphthylene	2	NE	µg/L
Anthracene	2	8,300	µg/L
Benzo(a)anthracene	2	0.038	µg/L
Benzo(a)pyrene	2	0.038	µg/L
Benzo(b)fluoranthene	2	0.038	µg/L

Benzo(g,h,i)perylene	2	NE	µg/L
Benzo(k)fluoranthene	2	0.038	µg/L
Benzoic acid	10	NE	µg/L
Benzyl alcohol	10	NE	µg/L
Bis(2-chloroisopropyl)ether	5	1,400	µg/L
Bis(2-chloroethoxy)methane	5	NE	µg/L
Bis(2-chloroethyl)ether	5	0.30	µg/L
Bis(2-ethylhexyl)phthalate	5	12	µg/L
Butyl benzyl phthalate	5	1,500	µg/L
Carbazole	5	NE	µg/L
Chrysene	2	0.038	µg/L
Di-n-butyl phthalate	5	2,000	µg/L
Di-n-octyl phthalate	5	NE	µg/L
Dibenzo(a,h)anthracene	2	0.038	µg/L
Dibenzofuran	5	NE	µg/L
Diethyl phthalate	5	17,000	µg/L
Dimethyl phthalate	5	270,000	µg/L
Fluoranthene	2	130	µg/L
Fluorene	2	1,100	µg/L
Hexachlorobenzene	5	0.0028	µg/L
Hexachlorobutadiene	5*	4.4	µg/L
Hexachlorocyclopentadiene	5	40	µg/L
Hexachloroethane	5	14	µg/L
Indeno(1,2,3-cd)pyrene	2	0.038	µg/L
Isophorone	5	350	µg/L
n-Nitrosodipropylamine	5	0.05	µg/L
n-Nitrosodiphenylamine	10	33	µg/L
Napthalene	2	NE	µg/L
Nitrobenzene	5	17	µg/L
Pentachlorophenol	10	1	µg/L
Phenanthrene	2	NE	µg/L
Phenol	5	10,000	µg/L
Pyrene	2	830	µg/L

Table 4			
Analyte	Laboratory RL	DQO	Unit
SVOCs (PAHs) via 8270SIM			
Acenaphthene	0.02	670	µg/L
Acenaphthylene	0.02	NE	µg/L
Anthracene	0.02	8300	µg/L
Benzo(a)anthracene	0.02	0.038	µg/L
Benzo(a)pyrene	0.02	0.038	µg/L
Benzo(b)fluoranthene	0.02	0.038	µg/L
Benzo(ghi)perylene	0.02	NE	µg/L
Benzo(k)fluoranthene	0.02	0.038	µg/L
Chrysene	0.02	0.038	µg/L
Dibenzo(a,h)anthracene	0.02	0.038	µg/L
Fluoranthene	0.02	130	µg/L
Fluorene	0.02	1100	µg/L
Indeno(1,2,3-cd)pyrene	0.02	0.038	µg/L
Naphthalene	0.2	NE	µg/L
Phenanthrene	0.02	NE	µg/L
Pyrene	0.02	830	µg/L

Table 5		
Analyte	Laboratory RL	Unit
Dioxins and Furans via Method 1613B		
2,3,7,8-TCDD	10.0	pg/L
Total TCDD	calculated	--
2,3,7,8-TCDF	10.0	pg/L
Total-TCDF	calculated	--
1,2,3,7,8-PeCDD	50.0	pg/L
Total-PeCDD	calculated	--
1,2,3,7,8-PeCDF	50.0	pg/L
2,3,4,7,8-PeCDF	50.0	pg/L
Total-PeCDF	calculated	--
1,2,3,4,7,8-HxCDD	50.0	pg/L
1,2,3,6,7,8-HxCDD	50.0	pg/L
1,2,3,7,8,9-HxCDD	50.0	pg/L
Total-HxCDD	calculated	--
1,2,3,4,7,8-HxCDF	50.0	pg/L
1,2,3,6,7,8-HxCDF	50.0	pg/L
1,2,3,7,8,9-HxCDF	50.0	pg/L
2,3,4,6,7,8-HxCDF	50.0	pg/L
Total-HxCDF	calculated	--
1,2,3,4,6,7,8-HpCDD	50.0	pg/L
Total-HpCDD	calculated	--
1,2,3,4,6,7,8-HpCDF	50.0	pg/L
1,2,3,4,7,8,9-HpCDF	50.0	pg/L
Total-HpCDF	calculated	--
OCDD	100.0	pg/L
OCDF	100.0	pg/L

Table 6		
Analyte	Laboratory RL	Unit
PFAS via Method 537.1 Rev2		
11-Chloroeicosafluoro-3-oxaundecane-1-sulfonic acid	4.0	ng/L
9-Chlorohexadecafluoro-3-oxanonane-1-sulfonic acid	4.0	ng/L
4,8-Dioxa-3H-perfluorononanoic acid	4.0	ng/L
Hexafluoropropylene oxide dimer acid	6.0	ng/L
Perfluorobutanesulfonic acid	2.0	ng/L
Perfluorodecanoic acid	2.0	ng/L
Perfluorododecanoic acid	2.0	ng/L
Perfluoroheptanoic acid	2.0	ng/L
Perfluorohexanoic acid	2.0	ng/L
Perfluorohexanesulfonic acid	2.0	ng/L
Perfluorononanoic acid	2.0	ng/L
Perfluorooctanoic acid	2.0	ng/L
Perfluorooctanesulfonic acid	2.0	ng/L
Perfluoroundecanoic acid	2.0	ng/L
N-ethyl perfluorooctanesulfonamidoacetic acid	2.0	ng/L
N-methyl perfluorooctanesulfonamidoacetic acid	2.0	ng/L
Perfluorotetradecanoic acid	2.0	ng/L
Perfluorotridecanoic acid	2.0	ng/L
PFAS via Method 533		
1H,1H, 2H, 2H-Perfluorohexane sulfonic acid	2.0	ng/L
1H,1H,2H,2H-Perfluorooctane sulfonic acid	2.0	ng/L
1H,1H,2H,2H-Perfluorodecane sulfonic acid	2.0	ng/L
Nonafluoro-3,6-dioxaheptanoic acid	2.0	ng/L
Perfluorobutanoic acid	2.0	ng/L
Perfluoro(2-ethoxyethane)sulfonic acid	2.0	ng/L
Perfluoroheptanesulfonic acid	2.0	ng/L
Perfluoro-4-methoxybutanoic acid	2.0	ng/L
Perfluoro-3-methoxypropanoic acid	2.0	ng/L
Perfluoropentanoic acid	2.0	ng/L
Perfluoropentanesulfonic acid	2.0	ng/L