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**FLATHEAD LAKE
BIO STATION**
UNIVERSITY OF MONTANA

MONITORING MONTANA WATERS

Providing assistance with water monitoring efforts in Montana

STANDARD OPERATING PROCEDURES (SOP)

In order to collect scientifically credible data, standardized methods must be followed. Following the same methodology as water quality professionals in Montana ensures errors are not made and that data are accurate. Having completed standard operating procedures (SOPs) and sampling and analysis plans (SAPs) ensures that groups collect the types of samples needed and that the correct methods are used. The Department of Environmental Quality (DEQ) uses standard operating procedures listed in their [Water Resource Monitoring Methods Selection Guide](#) (MMSG). By following these approved DEQ methods, data collected by citizen-led sampling efforts can be stored and used by FLBS and DEQ in their respective databases, and ensures that data are of sufficient quality to be used for management efforts.

Most of the analytes that are included in monitoring plans are chemical constituents collected in water samples. Monitoring these analytes provides a general picture of watershed health and abnormal values can indicate a water quality problem. Though the sample collection procedures are the same for many analytes, the sample holding times and preservation methods can vary so it is important for watershed groups to know the details for each analyte of interest (see Table 1 in the Appendix). Other analytes, like chlorophyll-*a*, require unique equipment and have specific procedures to follow during sample collection and storage. The most important thing for sample collection is for groups to be familiar with all procedures and equipment before going into the field. All methods listed in this document come from the [MMSG](#), mostly in original form. Detailed methods for additional variables of interest (e.g. riparian vegetation surveys, substrate surveys) can be found in the [MMSG](#) document listed on the [MT DEQ Monitoring Water Quality](#) website under Volunteer Monitoring.



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CHEMICAL CONSTITUENTS

Unfiltered and Filtered Samples

Listed below are the general procedures for collecting water samples for the purpose of analyzing chemical constituents like nutrients and metals. Some of these analytes require specific equipment and collection and storage procedures.

The following methods are known as “grab procedures”. These procedures detail how to collect raw samples directly from a body of water. For small analytes, sampling will require filtration, and these procedures are listed at the end of this section. For both sampling methods, you must also perform the field blank methods with clean water. “Field blanks are used to check for contamination introduced by transporting equipment in the field, improper procedures used by field crews...” and other



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contamination scenarios ([MMSG](#)). You can find the procedures for using field blanks at the end of this section. The proper number of duplicate samples must also be collected per sampling event. The Appendix includes examples (Tables 1 and 2) illustrating the analyte, collection vessel, preservation method and holding time. Groups analyzing water chemistry samples at the FLBS Freshwater Research Lab will receive a table detailing analytes, collection vessels, and preservation method in each cooler that is shipped for each sampling event.

Considerations:

- Sample collection method selection depends on site conditions (e.g., wadable, watercraft needed, sampling surface only or multiple depths)
- Analytical requirements per analyte will determine whether samples must be filtered or not, acid preserved or not, on ice or frozen, etc.
- Samples will either be hand-delivered or shipped to the analytical laboratory; care must be taken to ensure required sample temperatures are maintained and that samples will arrive at the lab within required holding time limits.

Equipment and Software:

- Sample bottles (Provided by MMW)
- Permanent marker
- Clear tape to cover labels
- Preservative, if needed
- Cooler(s) for sample storage (Provided by MMW)
- Ice for sample preservation
- Filters and syringes (if using)
- Extension pole sampler with collection bottle (if using)
- Decontamination procedures and supplies, if using collection equipment at multiple sites

Unfiltered Samples: Grab Technique

1. Label the bottle and cover the label with clear tape to prevent water damage.
2. Carry the bottle to a suitable sampling location:
 - Sampler can safely wade and stand or access the water from a boat.
 - Water column is well-mixed and deep enough to allow sampler to avoid surface scum and bottom sediments.



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- Upstream or away from any disturbance to water column or bottom sediments.
3. Triple-rinse the bottle and lid: face upstream into the direction of the flow, collect a small volume of water in the bottle, replace the lid, and shake gently. Discard the rinse water downstream. Repeat this process three times to triple-rinse.

NOTE: Do not rinse bottles that have preservatives added to the bottle prior to sampling (e.g., *E. coli*, mercury, and dissolved organic carbon sample bottles are often pre-preserved).

4. Collect the sample:
 - For wadable locations, submerge the bottle so the mouth is below the water surface but above the bottom and allow the bottle to fill.
 - For lake surfaces (from a boat), submerge the bottle until the sampler's elbow is at the water surface and allow the bottle to fill.
5. Leave appropriate headspace:
 - For most samples, the bottle should be filled to the shoulder or line that denotes the target volume; this will leave a small amount of head space, especially necessary if preservative will be added to the sample.
 - If samples are to be frozen, leave sufficient head space to allow the sample to expand when it freezes without the bottle breaking.
 - If samples require zero headspace (e.g., volatile organic analysis (VOA) or ultra-low-level mercury (ULL-Hg)), submerge the container and lid, remove the lid, allow to fill completely, and secure the lid, all while submerged; verify there is no head space or air bubbles; if head space or air bubbles remain, use the lid to add a small amount of water until a convex meniscus (slight bulge of water surface) forms, then secure the lid.
6. If preservative is required to be added to the sample, put on gloves, carefully unscrew the lid, pour the entire contents of the preservative vial into the sample bottle, replace the lid, and gently invert the sample bottle three times to mix the preservative into the sample. Discard the empty preservative vial.
7. Store samples upright according to sample preservation and storage requirements (e.g., in a cooler on regular ice at $\leq 6^{\circ}\text{C}$, or frozen on dry ice).
8. Deliver samples to the analytical laboratory within required holding times.

Filtered Samples: Grab Technique

1. Label the bottle and cover the label with clear tape to prevent water damage.
2. Carry the bottle, filter and syringes to a suitable sampling location:
 - Sampler can safely wade and stand or access the water from a boat.



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- Water column is well-mixed and deep enough to allow sampler to avoid surface scum and bottom sediments.
 - Upstream or away from any disturbance to water column or bottom sediments.
3. Open a new 60 cc syringe package, remove the syringe, and discard the packaging. Triple-rinse the syringe by drawing ambient (stream or lake) water into the syringe, gently shaking, and compressing the syringe to force the water out; repeat this three times.
 4. Fill the syringe with ambient water (draw water into the syringe from below the water surface at wadable locations or from the Van Dorn or Kemmerer, if using).
 5. Open a new 0.45 μm filter package by gripping the blue ring and peeling the cover open. Screw the filter onto the syringe and discard the packaging. Pass a small amount of water through the filter to “prime” the filter.
 6. Triple-rinse the sample bottle with filtered water: plunge a small amount of water (approximately 10-20ml) from the syringe through the filter into the sample bottle. Replace the lid, shake gently, and then discard the rinse water downstream. Repeat this process three times to triple-rinse the bottle with filtered water. When finished rinsing, unscrew and discard the filter used for rinsing.

NOTE: Do not rinse bottles that have preservatives added to the bottle prior to sampling (e.g., *E. coli*, mercury, and dissolved organic carbon sample bottles are often pre-preserved).

7. Refill the syringe with ambient water, open and attach a new filter, and pass a small amount of water through the filter to “prime” it.
8. Collect the sample: fill the bottle with filtered water. Often, to fill the bottle will require multiple refills of the syringe; when the syringe is empty, grip the filter’s blue ring, unscrew the filter and refill the syringe, taking care not to contaminate the filter. If the filter is not clogged, screw the filter back onto the syringe and continue filtering until the bottle is sufficiently full. If the filter clogs mid-way throughout filtering, unscrew and discard the clogged filter, refill the syringe, screw on a new filter, pass a small amount of water through the new filter, and continue filtering. Repeat this process until the sample bottle is full.
9. Leave appropriate headspace:
 - For most samples, the bottle should be filled to the shoulder or line that denotes the target volume; this will leave a small amount of head space, especially necessary if preservative will be added to the sample.
 - If samples are to be frozen, leave sufficient head space to allow the sample to expand when it freezes without the bottle breaking.
 - If samples require zero headspace (e.g., volatile organic analysis (VOA) or ultra-low-level mercury (ULL-Hg)), submerge the container and lid, remove the lid, allow to fill completely, and



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secure the lid, all while submerged; verify there is no head space or air bubbles; if head space or air bubbles remain, use the lid to add a small amount of water until a convex meniscus forms, then secure the lid.

10. If preservative is required to be added to the sample, put on gloves, carefully unscrew the lid, pour the entire contents of the preservative vial into the sample bottle, replace the lid, and gently invert the sample bottle three times to mix the preservative into the sample. Discard the empty preservative vial.
11. Store samples upright according to sample preservation and storage requirements (e.g., in a cooler on regular ice at $\leq 6^{\circ}\text{C}$)
12. Deliver samples to the analytical laboratory within required holding times.

FIELD BLANKS

Field blanks are samples of clean (e.g., laboratory-grade deionized) water prepared in the field following the same rinse, collection, preservation and storage procedures used for routine samples. Field blanks are used to check for contamination introduced by transporting equipment in the field, improper procedures used by field crews, etc.

NOTE: Typically, one set of field blanks is prepared per sampling event for each analyte collected during the sampling event. For example, if a field crew collects samples at multiple sites over multiple days, they would prepare a set of field blanks (i.e., one blank per analyte collected during the sampling) at the end of the trip before they depart from the field at their last site.

1. Label the field blank sample bottles with the date and personnel information, and add the words 'field blank' to the label or add a code to the site name to differentiate blanks from duplicate and routine samples. Cover the labels with clear tape to prevent water damage.
2. In the field (e.g., at your vehicle near your last site), assemble the supplies needed to prepare field blanks (e.g., deionized water, filters and syringes if applicable, preservatives if applicable).
3. Rinse the field blank bottles following the same procedures used to rinse the bottles for routine samples (e.g., triple-rinse with unfiltered or filtered water depending on the analyte), using deionized water rather than ambient stream water.
4. Pour or filter, depending on the analyte, deionized water into the sample bottle.
5. If preservative is added to the routine samples for a parameter, add acid to the field blanks for that analyte as well.
6. Store field blanks following the same procedures used for routine and duplicate samples.



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7. Deliver field blanks to the analytical laboratory alongside the routine and duplicate samples collected during the sampling event.

DUPLICATE SAMPLES

Duplicate samples are two samples taken from and representative of the same population and carried through all steps of the sampling and analytical procedures in an identical manner. Duplicate samples are used to assess variance of the total method including sampling and analysis.

NOTE: Duplicate samples are typically collected at a rate of 10% of the total number of routine samples being collected for a project for each parameter. Collect duplicates in the same manner for a parameter and label using the same site, date, and personnel information, but include “duplicate” in the labeling.

BIOLOGICAL CONSTITUENTS

Chlorophyll-*a*

Chlorophyll-*a* is a useful analyte to give an idea on how much algae and similar organisms may be present in your water body. While chlorophyll-*a* levels fluctuate throughout the year, they are typically highest in mid to late summer. Sampling for chlorophyll-*a* requires special filtration equipment and procedures.

Like any field sample, you must also perform the field blank methods using clean water. “Field blanks are used to check for contamination introduced by transporting equipment in the field, improper procedures used by field crews...” and other contamination scenarios ([MMSG](#)). You can find the procedures for using field blanks in the previous section.

Considerations:

- Chlorophyll-*a* breaks down readily in sunlight; efforts should be taken to minimize exposure of the sample to sunlight, including performing filtering in a shaded location, using a dark bottle to collect the sample, and setting up the filter apparatus prior to sample collection to minimize time between sampling and filtration.
- The volume of water filtered must be recorded.

Equipment and Software:

- Field form
- 1L dark Nalgene bottle (Provided by MMW)
- 100-250ml graduated cylinder
- Petri dishes or centrifuge tubes



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- Squeeze bottle with tap water
- Vacuum hand pump vacuum with tubing
- Nalgene filtering unit
- Tweezers or forceps
- GF/F glass fiber filters (0.70 μm)
- Aluminum foil
- Cooler with ice (Provided by MMW)

Procedure

1. Identify an appropriate place to collect the sample:
 - Water can be safely accessed.
 - Water column is well-mixed.
 - Water is deep enough to submerge the collection unit (graduated cylinder or bottle).
 - A shaded location is available nearby in which to perform filtering.
2. Set up the filter apparatus:
 - Ensure the filter unit is clean and free of debris.
 - Use clean forceps to place a glass fiber filter (0.70 μm) on the filter holder. Use a small amount of tap water (not stream or lake water) from a squirt bottle to settle the filter.
 - Place the top of the filter flask on top of the filter, grasp the cuff and carefully screw it on tight without tearing the filter.
 - Attach the vacuum hand pump with tubing.
3. Triple-rinse the dark Nalgene bottle with ambient stream or lake water; avoid disturbing the water column in the area where the final sample will be collected.
4. Grab a water sample from an undisturbed location using 1L dark Nalgene bottle and transport the sample to a shaded location for filtering.

NOTE: Avoid exposing samples to direct sunlight during processing.

5. Measure 20 ml or more of sample water into the graduated cylinder, pour into the filter funnel, and draw the sample through the filter using the vacuum hand pump.

NOTE: To avoid rupture of fragile algal cells, do not exceed 9.0 inches Hg on the vacuum gauge.

6. Continue measuring additional sample water using the graduated cylinder, adding it to the filter flask and passing it through the filter until a sufficient sample has been filtered. The volume of water



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filtered may vary from 20ml to 1000ml or more. When filtration slows and the filter has developed a distinct green (or green-brown) color, sufficient sample has been filtered.

NOTE: Do not allow the filter to clog! If a filter completely clogs while water remains in the upper half of the apparatus, discard the filter and start again, using less water volume.

7. When finished filtering, unplug the hand pump, remove the top of the filter flask, and use forceps to fold the filter in half with the colored side folded in on itself, and place the filter in the petri dish or centrifuge tube. Label the petri dish with site ID, date, and the sampler's name, and cover the label with clear tape. Wrap the labeled petri dish in aluminum foil to eliminate light exposure and place the sample in a ziploc bag.
8. Immediately place the sample in a cooler with dry ice and store it frozen until it is received by the analytical laboratory. If dry ice is not used to freeze samples while in the field, samples must be stored in a cooler surrounded by regular ice at $\leq 6^{\circ}\text{C}$ while in the field, then frozen solid in a freezer prior to shipping.
9. Record the total volume of water filtered on the field form!

NOTE: It is very important to keep track of and record the total volume of water that you filtered to collect the sample.

MULTIMETER INSTRUMENTS IN THE FIELD

Multimeter instruments are useful tools in the field to acquire data for multiple parameters without having to use laboratory analysis. Dissolved oxygen, temperature, conductivity, and pH are typically measured in the field using multimeter instruments for data that best represents natural conditions in the water body. Due to short recommended holding times, samples for most of these parameters cannot be shipped to the lab for analysis. Before using multimeter instruments, review the manufacturer operations manuals for instruction on use and calibration. The methods for general use of multimeter instruments in this section come from DEQ's [*Water Quality Planning Bureau Field Procedures Manual for Water Quality Assessment Monitoring*](#). If you have questions about specific instruments or instrument calibrations reach out to MMW for support and assistance.

Dissolved Oxygen, Specific Conductivity, and Water Temperature

1. Immediately upon arrival at the site, turn the YSI 85 (or similar model) instrument on, open the case and allow it to remain undisturbed for ≥ 15 minutes in a shaded location.
2. Perform field calibration of dissolved oxygen, using the calibration values appendix in the operations manual to verify measurement accuracy.



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3. At the site, submerge the probe in the water, shake vigorously to remove any air bubbles trapped near the probe, and position it facing upstream into the flow. Ensure that there are no obstructions in front of the probe (i.e., rocks, macrophytes, debris). If the water is not flowing, gently move the probe from side to side to circulate the water around the probe.
4. Allow a few moments for measurements to stabilize and record dissolved oxygen (mg/L), specific conductivity (μS), and water temperature ($^{\circ}\text{C}$). Be sure “ready” option is showing prior to making the reading or that the measurement has stabilized and is not drifting up or down.

pH

1. At the site, submerge the probe in the water. Allow a few moments for instrument measurements to stabilize and record pH.
2. While in the field at the end of each day of sampling, perform a two-point calibration check to verify performance of the meter.

Turbidity

1. At the site, submerge the probe in the water and position it facing upstream into the flow. Ensure that there are no obstructions in front of the probe (i.e., rocks, macrophytes, debris), be cautious not to disturb the substrate around the probe, and allow it to stabilize for several moments. Record turbidity (NTU) measurement results.

STREAM DISCHARGE

Stream discharge is the measure of how much water is flowing through a river over a given time period. To calculate discharge, the velocity of the water (flow) and area of the transect (width and average depth) must first be measured. Both the stream flow and stream transect methods included below are taken from DEQ’s [Water Quality Planning Bureau Field Procedures Manual for Water Quality Assessment Monitoring](#). While not perfectly accurate, these methods allow for a general estimate of stream discharge.

If a group is interested in the load of an analyte then discharge must be measured. Load is a measure of how much of an analyte is being discharged through a river over a period of time (e.g. 10 pounds of nitrogen per year), while concentration simply measures the mass of an analyte in a given volume of water at one moment (e.g. 10 milligrams of nitrogen per liter).

Equipment and Software:

- Stakes or flags for marking start/end points
- Measuring tape/range finder
- Field notebook



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- Flow meter (if available)
- Biodegradable stick (if using the floating stick method)
- Waterproof digital camera

FLOW/VELOCITY

Floating Stick/Ball Method

If a flow meter is not available then the floating stick/ball method can be used to make a general estimate of water velocity. It is a semi-quantitative method that tends to underestimate the flow due to slower velocity near the surface, but it is more accurate than a visual estimate.

1. Find a stream reach that is straight and uniform in width and depth; a glide is preferred. This will assure that laminar flow is achieved to the greatest extent possible. Measure a length at least twice the mean wetted width (≥ 50 ft is preferable) and mark each end by hanging flagging or driving a stake or rebar into the ground at the high water line.
2. Determine the mean width (from the water's edge) by measuring at least three cross-sections (if wadable), using a rangefinder, or by making a visual estimate.
3. Determine the mean depth by measuring depth at multiple points throughout the reach (if wadable) or by making a visual estimate.
4. Record the measured distance and a description of each stake's location in the field notes for high flow. Note landmarks and make a sketch if necessary to help identify stake locations in the event that they are no longer in place during subsequent flow measurements. Photograph both stakes to record their location along the streambank and the water level.
5. Toss a small stick or other biodegradable floating object heavy enough to stay in and move consistently with the main current into the middle of the stream above the upstream marker of the measured reach. Begin timing when the object passes the upstream marker. Count (with a watch or stopwatch) the seconds it takes the object to reach the downstream marker. The object must stay in the main current. If it does not, repeat the measurement. Complete three measurable floats. Remove the stakes upon completion unless subsequent site visits requiring flow measurement are anticipated.
6. Record the following information in your field notes for high flow:
 - Reach length (ft or m)
 - Mean depth (ft or m)
 - Mean width (ft or m)



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- Float times (sec)
7. Complete the following calculations in your field notes for high flow:
- **Cross-sectional area** (m^2 or ft^2) = Mean width **x** Mean depth
 - **Average float time** (sec) = (Float time 1 + Float time 2 + Float time 3) / 3
 - **Float velocity** (ft/sec or m/s) = Reach Length / Average float time
 - **Discharge** (ft^3/sec or m^3/sec) = Cross-sectional area **x** Float velocity

Flow Meter

1. Assemble the meter and adjust the settings
 - Refer to the instrument operations manual for further details on use, calibration and maintenance.
 - Set the flow meter to the “fixed point average” (FPA) setting, which provides an average of velocities over a fixed period of time, and specify a FPA interval of 10 seconds. Set the units to either ft/s or m/s.
2. The flow meter method is appropriate for narrow streams where 10-15 points along a cross-section can be measured, or wide streams where 20-30 points along a cross-section can be measured. A flow meter with a wading rod can only be used in streams that have sufficient water depth to reliably use the instrument (≥ 0.2 in).
3. Choose a location for the cross-section in a straight reach with laminar flow; a glide is preferred. Consider the following guidance:
 - Location should be free of disturbances (i.e., boulders, aquatic growth, pipe joints, inflowing or out flowing side channels or tributaries, other obstructions)
 - Flow should be free of swirls, eddies, vortices, backward flow, and dead zones
 - Avoid areas downstream of sharp bends, upstream or downstream of vertical drops or where stream empties into a stationary body of water
 - Use best judgment in choosing the best site when all of the above criteria do not exist.
4. Stretch a tape (ft or m) between end-points of your cross-section, ensuring that it is oriented perpendicular to flow. The tape should be stretched at a minimum from water’s edge to water’s edge; however, it is acceptable to extend the tape beyond water’s edge on either bank to allow for ease of securing the tape. Use bank pins or stakes to secure the ends of the tape in place.



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5. For narrow streams, divide the distance from left water's edge to right water's edge by 10-15; for wide streams, divide the distance from 20-30 to determine the number of equidistant points along the cross-section at which flow measurements will be collected. It is acceptable to round to nearest 0.25 ft or 0.1 m for ease of determining the distance between points of measurement.
6. Flow is measured at multiple equidistant points from left water's edge to right water's edge to account for complexity and variability in channel shape and flow patterns. Start at left water's edge and call out the location on the tape to the person recording the data. At left water's edge and right water's edge (the initial and final points of measurement, respectively) the depth and velocity will each be recorded as "0".
7. From left water's edge, move across the channel cross-section toward right water's edge, locate the next point of measurement on the tape (calculated previously) and, holding the wading rod vertical and steady with the base on top of the substrate, position the probe directly into (parallel to) the flow. Stand downstream from the tape and meter and at least 18" off to the side of the wading rod to avoid disrupting the flow measurement.
8. Read the depth on the graduated hex main rod at this point to the person recording the data. Each mark on the rod is 0.1 ft. Double marks are at 0.5 ft and triple marks are at 1 ft.

IMPORTANT: Be careful not to push the base of the wading rod down into the substrate when measuring flow in streams with soft substrates.

9. Position the probe to 0.6 depth by adjusting the round setting rod to the depth of the water (from the previous step). Slide the round sliding rod to line up the foot (or meter) scale on the sliding rod with the tenth scale on top of the main hex rod. For example, if the water depth is 2.7 ft, line up the 2 on the round sliding rod with the 7 on the tenth scale on the top of the main hex rod.
10. Once positioned to begin recording measurements, wait for a new averaging interval to begin or hit the reset button (ON/C). Allow the flow meter to cycle through three 10-second fixed point average intervals, then call out the average of these three values to the person recording the data.
11. Fill in information in the field notebook:
 - Waterbody name
 - Activity ID
 - Transect letter nearest to flow cross-section
12. Fill in the following information in the field notebook with proper units
 - Distance on tape (ft or m)
 - Depth (ft or m)
 - Velocity at point (ft/s or m/s)

- Comments (i.e., “left water’s edge”, “right water’s edge”, channel irregularities, unavoidable obstructions like channel islands, etc.)

STREAM TRANSECTS

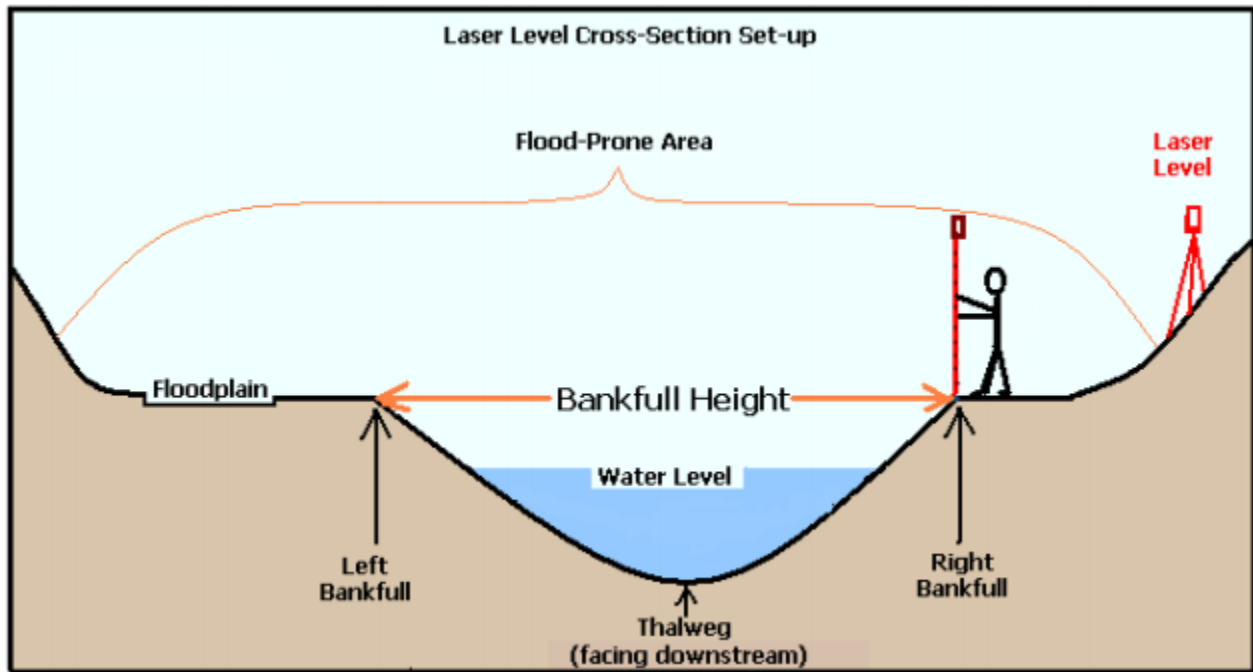


Figure 1. This diagram shows natural markers used in stream transects. Bankfull height is also known as the high water mark. This area will be where vegetation becomes more prevalent above the exposed rock or sediment in the stream bed. The thalweg is the lowest point in the streambed, regardless of the water level.

1. Identify a representative riffle cross-section in the stream reach which typifies the form of the stream. Pick a fairly straight section where the channel is somewhat constricted; avoid large boulders and large woody debris which alter the form and lateral extent of the channel.
2. Install stakes at both left bankfull and right bankfull elevation. Attach the tape first to the left bank stake, facing downstream. Pull the tape tight and attach it to the right bank stake. If the tape is so long that it tends to sag, install a nylon cord tightly at the same points on the stakes so you may have a more uniform horizontal plane to measure to.

IMPORTANT: To achieve a near-level bankfull elevation, measure the bankfull-to-water surface distance at the side with the most reliable bankfull indicator, and then adjust level of the tape at the other side of the channel to correspond with that measurement.



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- Determine the distance on the tape between left bankfull and right bankfull. Divide this distance by 15-30 to obtain 15-30 equidistant points along the cross-section within bankfull (use 30 points for larger streams).
- Measure the cross-section from left bankfull to right bankfull.

IMPORTANT: Measurements must be taken at left bankfull (LBF), Thalweg (THL), and right bankfull (RBF); if the 15-30 calculated points along the cross-section do not fall exactly on these points, add these three additional locations (distances on the tape) to the field notes.

Record the station (distance on the tape, ft) and the corresponding depth (measured from the channel bottom to the tape or nylon cord, ft) at each location in the field notes. Include notation indicating right bankfull (RBF), right water's edge (RWE), Thalweg (THL), left water's edge (LWE), and left bankfull (LBF).

APPENDIX

Table 1. FLBS Freshwater Research Lab's Pricing and Holding Time Example

Parameter	Units	Method Reference	Reporting Limit	Target Holding Time (days)	Filtration	Preservation	Price (\$)	Combo Price (\$)
NUTRIENTS								
Total Phosphorus as P	µg/L	USGS 03-4174, EPA 365.1	1.5	28	n.a.	Freeze or 0.2% H ₂ SO ₄ (v/v)	20	31
Total Nitrogen as N	µg/L	USGS 03-4174, EPA 353.2	25.0				21	
Soluble Reactive Phosphorus	µg/L	EPA 365.1	0.8		0.45µm		14	23
Nitrate-Nitrite as N	µg/L	EPA 353.2	1.5				15	
Ammonia as N	µg/L	DPA 350.1	1.5			14		
Reactive Silica as SiO ₂	mg/L	EPA 370.1	0.10			14		
Total Organic Carbon	mg/L	EPA 415.1	0.10		n.a.	0.2% H ₃ PO ₄ (v/v)	13	
Dissolved Organic Carbon	mg/L	EPA 415.1	0.10		0.45µm		13	



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Table 2. Shipping and Holding Methods Example

Analyte	Collection Vessel	Preservation
Total Nitrogen	60mL HDPE bottle (BLUE TAPE)	Frozen @ -20°C until delivery to lab
Total Phosphorus		
Nitrate + Nitrite	60mL HDPE bottle (RED TAPE)	Field Filtered to 0.45um; Frozen @ -20°C until delivery to lab
Soluble Reactive Phosphorus		
Total Suspended Solids	1000mL HDPE bottle	Refrigerated @ 4°C until delivery to lab
Total Dissolved Solids		
Chlorophyll	GF/F filter	Field Filtered onto GF/F; Frozen @ -20°C in foil until delivery to lab