New Baseline Volunteer Checklist

Complete these steps to become a stream monitoring volunteer!
Questions? Contact WAV staff at wav@extension.wisc.edu.

UW–MADISON EXTENSION

- [ ] Complete the **Online Introduction to WAV** course *prior to* attending a field stream monitoring training: www.wateractionvolunteers.org/get-involved/become-a-volunteer/

- [ ] **Attend a 3-4 hour field training** to learn the monitoring methods, meet your local WAV Coordinator, and pick up your equipment. Sign up for a training in spring at www.wateractionvolunteers.org/events/

- [ ] Work with your Coordinator and WAV staff to **choose a stream monitoring site to monitor.** Your site should be wadeable (can you safely walk across the stream?), accessible, and of interest to you! Your monitoring site will have a unique *Station ID* and *Station Name* in the Wisconsin DNR's SWIMS surface water database.

- [ ] **Identify your stream monitoring partner(s).** You can monitor with a friend, family member, or another volunteer. We strongly encourage monitoring with a teammate for ease and for safety.

- [ ] **Get set up to enter data** in the Wisconsin DNR's SWIMS database. Instructions are available at www.wateractionvolunteers.org/data/submit-data/

  1. Create a **MyWisconsin ID**.
  2. Email wav@extension.wisc.edu to ask WAV staff to set up your profile in SWIMS. Provide your full name, MyWisconsin ID email address, phone number, Station ID that you are monitoring, and the name of the WAV Coordinator or organization you are volunteering with (if applicable).
  3. Now you will be able to log in to SWIMS to enter data for your station.

- [ ] **Watch our quick 9-minute tutorial video when you're ready to submit your data**, to learn how to submit and edit data in SWIMS correctly: www.wateractionvolunteers.org/data/submit-data/

- [ ] **Sign up for the WAV monthly e-newsletter** to get program information, hear about new opportunities, and more. Go to www.wateractionvolunteers.org/ and click the button "Get WAV news".

Submitting your Stream Data to SWIMS

All WAV data is entered into the Wisconsin Department of Natural Resources' Surface Water Integrated Monitoring System (SWIMS) database. The SWIMS database is used by DNR staff and partners to enter and search for water quality data for assessing the health of our state's waters and to guide water protection and restoration decision-making.

Get access to SWIMS:

Step 1 - Create a MyWisconsin ID

You will use the State of Wisconsin's "MyWisconsin ID" system to log in to SWIMS each time.

- Go to apps.wisconsin.gov and click "Sign up" (small blue link below the login).
- Enter and verify your email address, create a password, and set up two-factor authentication (e.g., a text to your phone).

Step 2 - Email WAV Program Staff to create a SWIMS profile

Before you can log in to SWIMS, WAV staff need to create your profile in the database. Send an email to wav@extension.wisc.edu and provide:

- Your full name, the email address tied to your MyWisconsin ID, phone number, Station ID and Station Name, and the local Coordinator/organization you are monitoring with.

Learn how to submit data and avoid errors:

- Go to wateractionvolunteers.org/data/submit-data/ or scan the QR code to find resources to help you submit data to SWIMS.
- **Watch our 9-minute quick tutorial video** to learn how to correctly enter and edit data in SWIMS (available at the link above).
- Sign up for virtual training opportunities offered by WAV staff or your local WAV Coordinator.

Questions about entering data? Contact WAV staff at wav@extension.wisc.edu.

WAV Baseline Stream Monitoring Calendar

Every Month:

- Fill out the WAV datasheet for only the parameters you collect, in addition to your teammate's name, DNR station name and number, date and time, and your streamside observations.
- Enter your data into SWIMS when you get home: wateractionvolunteers.org/data/submit-data/
- Be on the lookout for aquatic invasive species. Collect a sample, take high quality photos, and then report any suspect AIS.

	Temperature	Transparency	Dissolved oxygen	Stream flow	Biotic index
May	☐	☐	☐	☐	☐
June	☐	☐	☐	☐	
July	☐	☐	☐	☐	
August	☐	☐	☐	☐	
September	☐	☐	☐	☐	☐ Complete fall sample in September or October
October	☐	☐	☐	☐	

Questions? Contact WAV staff at wav@extension.wisc.edu.

For more information, scan the QR code or go to wateractionvolunteers.org/baseline.

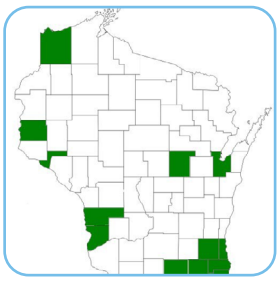
Aquatic Invasive Species

Extension
UNIVERSITY OF WISCONSIN–MADISON

WISCONSIN DEPT. OF NATURAL RESOURCES

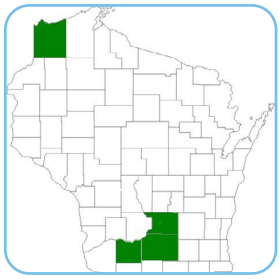
Freshwater Golden Clams

- While native fingernail clams are no larger than a dime (18mm), these invasive Asian clams can reach the size of a quarter (24mm).
- The concentric ridges of the clam's shell feel like a washboard, compared to the smooth shells of native fingernail clams. You can distinctly feel them with your finger.
- The clams have been found at densities of 20,000 clams per square meter. They can self-fertilize, so it only takes one to start a new population.
- The clams compete with native species for food and space, and can alter nutrient cycling within the stream.



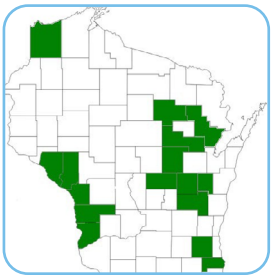
New Zealand Mudsnails

- New Zealand mudsnails are very small, no more than 4–6mm (1/4").
- Their shells, ranging in color from gray to light or dark brown, have 5–6 whorls and a right-side opening.
- New Zealand mudsnails can reproduce asexually by cloning and can become superabundant (500,000 snails per square meter) in productive streams.
- They compete with native stream species and alter food chains.
- Their small size allows them to be easily and unknowingly transported; their ability to seal their shells allows them to survive out of water for nearly a month.



Faucet Snails

- Faucet snails are small, generally growing to sizes up to 15mm (just over ½").
- Faucet snails range in color from pale brown to black, have a right-side opening shaped like a teardrop, and tend to have 5–6 whorls. Adults have concentric circles on the operculum (the structure that covers the shell opening).
- Faucet snails are filter feeders and have high growth rates, thus competing with native populations for habitat and food.
- Faucet snails act as hosts to a variety of parasites that pose a threat to native waterfowl that consume the snails.
- Faucet snails can live for up to a month in dry mud.



Rusty Crayfish

- Adults are up to 15cm (6") in length – larger than most native crayfish and with larger claws.
- They are easily identified by rust- colored spots on each side of the carapace (shield projecting backwards from its head).
- Females carry fertilized eggs under their tails for many months and can spread the population as they move or are transported.
- Rusty crayfish, with a higher metabolism than native crayfish, feed voraciously day and night, thus outcompeting the native crayfish that typically feed only at night. The aggressive rusty crayfish can eat vegetation, native crayfish, fish eggs, young fish, tadpoles, and macroinvertebrates, thus altering the stream ecosystem.

Photographing AIS Specimens

If target AIS are found, volunteers should always photograph the specimens and the monitoring location.

Materials:

- Camera or phone with camera

Additional photo guidance:

http://dnr.wi.gov/topic/Invasives/report.html

Process:

1. Photograph the landscape illustrating the location and extent of the occurrence so that the site is easy to recognize for follow-up visits. Include landmarks such as signs, beaches, public structures, memorable environmental features, or anything that is unique and would be easy to locate again. Take multiple landscape photos to ensure at least one is in focus.
2. If possible, take a GPS point. The GPS coordinates will help to relocate the population if needed.
3. Take several close-up photos of the specimen to show the various identifying characteristics. Include the data collection sheet in each picture to show the location (station name, number, date, etc.) Take multiple photos to ensure at least one is in focus.
4. Photographs should be submitted to local AIS contacts and emailed to: DNRInvasivePhotos@wisconsin.gov.

Collecting AIS Specimens

If target AIS are found, and volunteers are able, specimens should be collected and labeled.

Materials:

- Zip-top storage bags or sample vials/jars
- Labels for preserved specimens
- Pencil

Process:

1. Collect up to five specimens of the species, if possible.
2. Preserve specimens in bag or plastic/glass container with enough water to keep moist.
3. Label the specimens. Using a pencil, write the stream name, site description (e.g., Rocky Creek at CTH H), county, date, and your name on a slip of paper small enough to fit in the preservation vessel or to be applied to the outside of the specimen vessel.
4. Insert the paper into the container, or apply it to the outside. Close securely. Freeze specimens such as snails or crayfish as they will smell bad.
5. Specimens should be submitted to local AIS contacts.

Cleaning Your Gear

After monitoring each site, volunteers should clean, dry, and disinfect equipment and footwear.

Materials:

- Stiff bristle brush
- Spray bottle with tap water
- Rubber tote (18–50 gallons)
- Bleach solution (1 Tbsp bleach per 1 gallon water)
- Rubber gloves

Process:

1. Before leaving the stream:
   - **INSPECT** equipment.
   - **REMOVE** sediment, plants and animals by scrubbing equipment with a stiff brush.
   - **RINSE** equipment with tap water.
   - **DRAIN** all water from equipment.
2. Before entering another stream, it is recommended that volunteers switch to a completely new set of gear or take one of the following disinfection steps:
   - Far away from surface water, **SOAK** waders and other equipment in bleach solution for 20 minutes in rubber tote, then rinse. Wear gloves when handling bleach; or
   - **STEAM CLEAN** equipment; or
   - **SOAK** equipment in 140°F water for several minutes; or
   - **FREEZE** equipment for 8 hours.

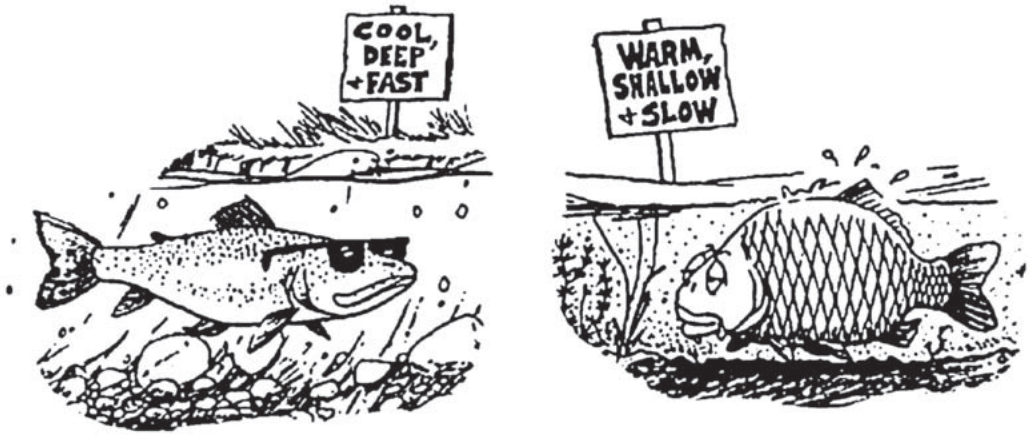
*New Zealand mudsnails do not react to bleach solution. Gear must be steamed, frozen, soaked, or treated with Virkon solution

Find your local AIS Contact at http://dnr.wi.gov/topic/Invasives/report.html

WAV WATER ACTION VOLUNTEERS

Temperature:
Its Role in Aquatic Habitats

Volunteer Monitoring Factsheet Series 2023



Why are we concerned?

- Temperature changes can affect all aquatic life. For example, warm water holds less dissolved oxygen than cold water and triggers higher plant growth and respiration rates. The lowered oxygen levels of warmer waters are further reduced when plants and animals die and decay.
- Although most aquatic life has adapted to survive within a range of water temperatures, some fish species, (trout, for example) require cooler waters. The metabolic rate of organisms, or the rate at which they convert food into energy, also increases with higher water temperatures, resulting in even greater demands on oxygen.
- Research also shows that extreme temperature fluctuations can make fish and insects more susceptible to disease, parasites and the harmful effects of toxic waste.

Time Needed:
10 minutes

Equipment Needed:
- Hipboots
- Thermometer
- Datasheet
- Pen/pencil

When to Measure:
Monthly from May to October

Temperature Conversion Chart

Fahrenheit	33	34	35	36	37	38	39	40	41	42	43	44	45	46	47	48	49	50	51
Celsius	.6	1.1	1.7	2.2	2.8	3.3	3.9	4.4	5	5.6	6.1	6.7	7.2	7.8	8.3	8.9	9.4	10	10.6

Fahrenheit	52	53	54	55	56	57	58	59	60	61	62	63	64	65	66	67	68	69	70
Celsius	11.1	11.7	12.2	12.8	13.3	13.9	14.4	15	15.6	16.1	16.7	17.2	17.8	18.3	18.9	19.4	20	20.6	21.1

Fahrenheit	71	72	73	74	75	76	77	78	79	80	81	82	83	84	85	86	87	88	89
Celsius	21.7	22.2	22.8	23.3	23.9	24.4	25	25.6	26.1	26.7	27.2	27.8	28.3	28.9	29.4	30	30.6	31.1	31.7

Background on Temperature

Stable water temperature is a critical factor in maintaining the health of a stream and its inhabitants. Temperatures over 78˚ F, (25.6˚ C) for example, are usually fatal to brook trout, which need waters in the range of 55˚- 65˚ F (12.8˚-18.3˚ C) in order to thrive. Other fish such as the smallmouth bass can survive an upper limit of 86˚ F (30˚ C) and carp can live in even warmer waters. So as temperature increases, cool water species will gradually be replaced by warm water ones.

Think Like a Scientist!

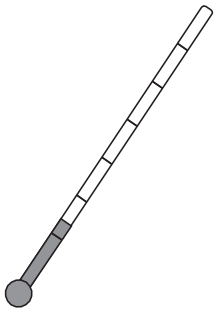
Follow the directions VERY CAREFULLY! Accuracy is a must for valid data

One of the most drastic ways that stream temperature is increased is by thermal pollution. Thermal pollution occurs when warm water is added to the stream. Industries such as power plants, paper mills and cheese factories may discharge heated water used in the manufacturing process into the streams. Runoff, in a more indirect way, can also add warm water to streams. Rainwater running off warmed surfaces, especially parking lots, roof tops and roads, increases stream temperatures.

Mill ponds and impoundments also increase water temperature because they contain a large surface area of slow-moving water which is warmed by the sun, affecting water temperature downstream.

Removing all overhanging trees that shade and cool the stream can also negatively impact stream temperatures. Another factor contributing to higher stream temperatures is eroding soil. Turbid water that results from eroded soil heats up quickly because the suspended sediments absorb the sun's radiant heat. Sediment also makes stream channels shallow. A shallow stream warms up faster than deep waters.

Collecting the Sample



1. To insure consistency in a long-term monitoring effort, the sampling location should be marked in some way. You can tie a piece of surveyor's tape to a tree or drive in a stake above the highest water line. Make sure you have any necessary permission before you mark a site. Record the air temperature before you take the stream temperature.
2. Test in the middle of the stream where the water is moving, not in pools or backwater areas.
3. You can use a standard alcohol thermometer for the measurement. Lower the thermometer about four inches below the surface, as close as possible to the middle of the stream.
4. Leave the thermometer immersed until the reading has stabilized. This usually takes about two minutes. Try to take the reading with the base of the thermometer still immersed. Record your measurement. The standard unit WAV uses to report water temperature is degrees Celsius (C).

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Water Action Volunteers is a cooperative program between the University of Wisconsin-Madison Division of Extension and the Wisconsin Department of Natural Resources. For more information, go to https://wateractionvolunteers.org/

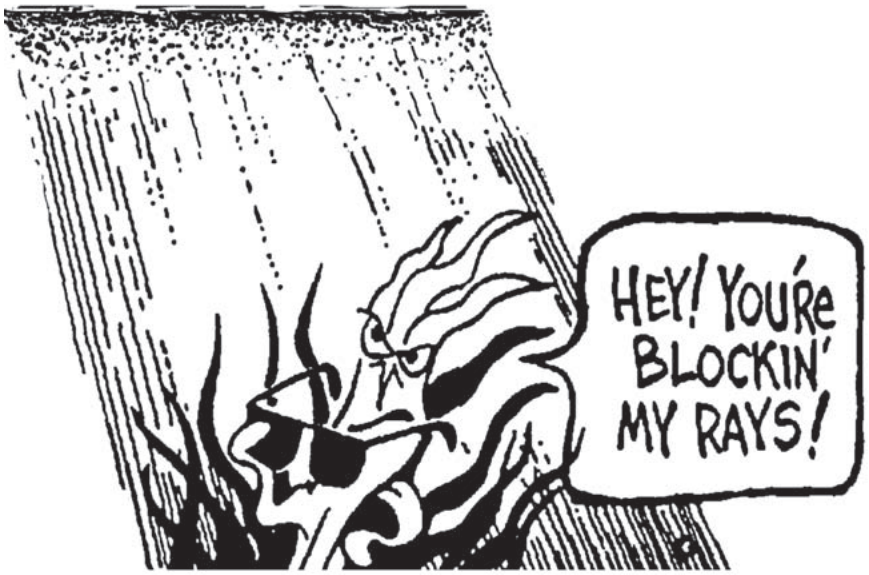
Transparency:
A Water Clarity Measure

WATER ACTION VOLUNTEERS

Volunteer Monitoring Factsheet Series 2023

Why are we concerned?

- Water clarity is one of the most obvious measures of water quality.
- Water clarity can be a useful indicator of runoff from construction sites fields, logging activity, industrial discharges and other sources.
- Monitoring transparency before, during and immediately after rain can provide a useful picture of potential runoff problems.



Time Needed:
10-20 minutes

When to Measure:
Monthly from May to October

Equipment Needed:
- Hipboots (if wading)
- 120cm transparency tube
- Datasheet
- Pen/pencil
- Bucket (optional)

Definition of Terms

Turbidity: The amount of suspended particles in the water.

Transparency: A measure of water clarity.

Transparency Tube: A tube with a black and white disc in the bottom, which is marked in centimeters or inches along its side. It is used for assessing the clarity of stream water

Suspended Material: Small particles floating in the water.

Sediment: Soil or other bits of eroded material that run off land and settle in still water.

NTU: Nephelometric Turbidity Units, which is a measure of the amount of light scattered by suspended material in the sample.

Background on Turbidity / Transparency

Murky water is easily seen as unhealthy. However, natural substances which are not harmful to the water can sometimes make water appear brown and murky. How do we know if the murky water is a cause for concern? Scientists have found a way to quantify the cloudiness of water by measuring its turbidity, which refers to the amount of suspended particles in the water. These small particles of soil, algae or other materials generally range in size from the microscopic level to about one millimeter, (about as thick as a pencil lead). More free-floating particles cause greater turbidity, resulting in less light penetration through the water. This hinders photosynthesis, necessary for healthy aquatic plant growth and production of dissolved oxygen. The water also becomes warmer because the suspended particles absorb heat, and warmer water holds less dissolved oxygen than cold water. The faster a stream flows, the more energy it has and the more sediment it can carry. Sources of turbidity include:

- erosion from fields, construction sites
- urban runoff from rainstorms and melting snow
- large number of bottom feeders (such as carp) which stir up bottom sediments
- excessive algal growth

Think Like a Scientist!

Follow the directions VERY CAREFULLY! Accuracy is a must for valid data comparisons.

Since we assess water clarity visually, we don't directly measure how many suspended particles are in the water. Instead we measure the transparency of the water, which takes into account both color and suspended particles. We do, however, have you use a conversion chart to estimate the turbidity measurement in nephelometric turbidity units (NTU) as well as recording in cm or inches.

Collecting the Sample

In general, collect the sample away from the river bank in the main flow area. Be careful not to collect water that has sediment from bottom disturbances (toss out the sample and try again if you get bottom sediment in your sample).

Wading Streams

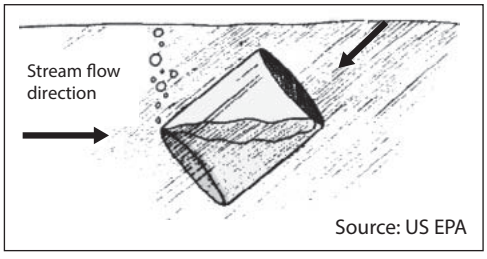
- Walk into the water downstream from the sampling location. Be careful not to stir up the bottom sediment upstream of your sampling location.
- Face upstream (into the current) in the middle of the stream.
- Collect your water sample by plunging your transparency tube or collection bucket 8-12 inches beneath the surface or halfway down from the surface. Scoop away from your body and into the current.
- Scoop water into the tube so it is filled to the top, or use a bucket to collect additional water from the stream at the site to fill the tube to the top.
- Return to shore with the sample.

From Shore

- To collect a sample from the shore, use a bucket or sample bottle attached to a pole. Scoop from below the surface in the upstream direction. Be careful not to stir up the sediment upstream of your sample.

From a Bridge

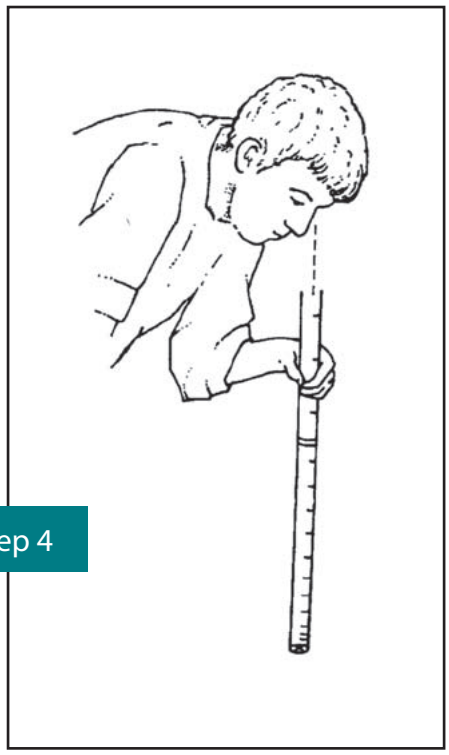
- If you are collecting a sample from a bridge, lower the bucket and get a sample from below the surface.



It's important to scoop your bucket down and into the current flow. Avoid sampling surface water.

Using the Transparency Tube

1. Walk upstream and lower the transparency tube into the water in the main part of the stream channel where water is flowing. Fill the tube, avoiding collecting only water from the surface. Tip the tube up slightly to release any air bubbles. Then, place your hand over the end of the tube and lift it out of the water. Carry the full tube to shore.
2. Stand out of direct sunlight. If you cannot get to a shady place, use your body to cast a shadow on the tube.
3. If you are wearing sunglasses, remove them. Then look for the black and white secchi disc on the bottom of tube. If the disc is immediately visible, record the max length of the tube (120 cm) on the datasheet.
4. If the secchi disc is not immediately visible, have your partner let water out a little at a time using the valve at the bottom of the tube until the disc is just visible. That is, have them stop letting water out immediately when you can just see the contrast between black and white on the disc.
5. Read the height of water using the measurements on the side of the tube.
6. Record the measurement on your datasheet.
7. Dump contents of the tube on the ground.
8. Collect a new sample then repeat steps 1 through 6.
9. Record the second measurement on your datasheet.
10. Add both readings, divide by 2, and record this average transparency on your datasheet.
11. (*Optional*) If you would like to compare your transparency value to a turbidity value, use the conversion chart on the next page.



US EPA

Try to stand out of sunlight when taking your measurements.

Transparency Conversion Chart

Centimeters	Inches	Turbidity Values*
<6.4	<2.5	<240
6.4 to 7.0	2.5 to 2.75	240
7.1 to 8.2	2.5 to 3.25	185
8.3 to 9.5	3.26 to 3.75	150
9.6 to 10.8	3.76 to 4.25	120
10.9 to 12.0	4.26 to 4.75	100
12.1 to 14.0	4.76 to 5.5	90
14.1 to 16.5	5.6 to 6.5	65
16.6 to 19.1	6.6 to 7.5	50
19.2 to 21.6	7.6 to 8.5	40
21.7 to 24.1	8.6 to 9.5	35
24.2 to 26.7	9.6 to 10.5	30
26.8 to 29.2	10.6 to 11.5	27
29.3 to 31.8	11.6 to 12.5	24
31.9 to 34.3	12.6 to 13.5	21
34.4 to 36.8	13.6 to 14.5	19
36.9 to 39.4	14.6 to 15.5	17
39.5 to 41.9	15.6 to 16.5	15
42.0 to 44.5	16.6 to 17.5	14
44.6 to 47.0	17.6 to 18.5	13
47.1 to 49.5	18.6 to 19.5	12
49.6 to 52.1	19.6 to 20.5	11
52.2 to 54.6	20.6 to 21.5	10
>54.7	>21.6	<10

*Roughly NTUs

Chart developed by Kevin Fermanich

What Do These Turbidity Values Mean?

All streams have background turbidity/transparency, or a baseline standard for a natural amount of turbidity/transparency. Fish and aquatic life that are native to streams have evolved over time to adapt to varying levels of background water clarity. For example, native fish and aquatic life in the Mississippi River are very happy with their murky environment. What causes problems in any stream or river are unusual concentrations of suspended particles and how long the water stays at a deviated level. When you collect transparency samples, it is important to note any fluctuations in values, which can help detect trends in water quality.

Time is probably the most influential factor in determining how turbidity affects the aquatic environment. The longer the water remains at unusually high values, the greater effect it has on fish and other aquatic life. Fish in particular become very stressed in waters that remain highly turbid for a long time. Signs of stress include increased respiration rate, reduced growth and feeding rates, delayed hatching and in severe cases, death. Fish eggs are ten times more sensitive to turbidity than adult fish. To further understand how time and turbidity impact fish, look at the graph that is included: "Relational Trends of Fresh Water Fish Activity to Turbidity Values and Time."

High turbidity levels affect humans, too. Acceptable turbidity levels for recreation is 5 NTU and acceptable levels for human consumption ranges from 1-5 NTU.

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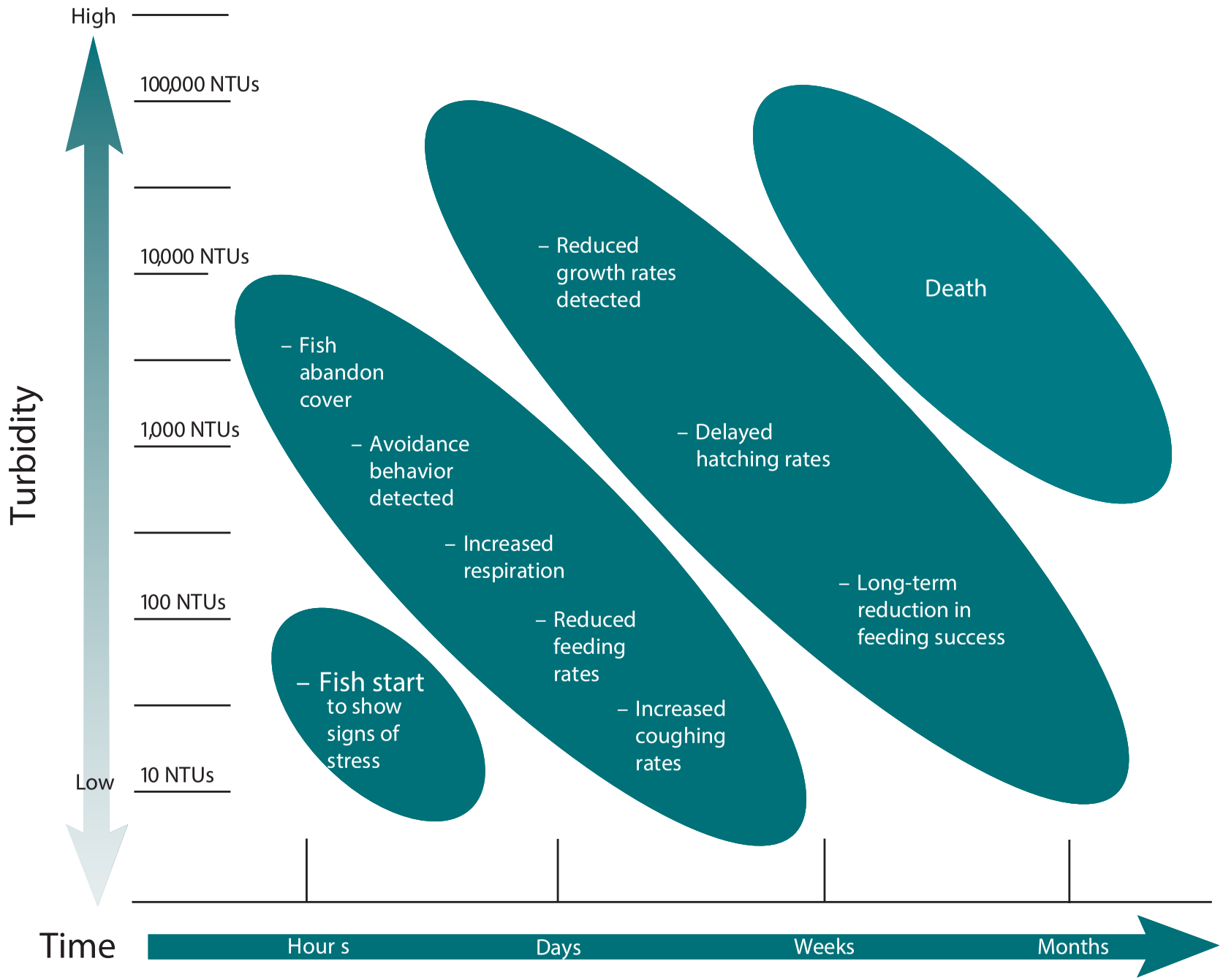


Dissolved Oxygen:
Aquatic Life Depends on It

Volunteer Monitoring Factsheet Series 2023

Why are we concerned?

- Both aquatic plants and animals depend on dissolved oxygen (D.O.) for survival.
- D.O. concentrations are influenced by many factors including water temperature, the rate of photosynthesis, the degree of light penetration (turbidity and water depth), the degree of water turbulence or wave action, and the amount of oxygen used by respiration and decay of organic matter.

Time Needed:
40 minutes

Equipment Needed:
- Hip boots
- Hach dissolved oxygen water test kit
- Safety glasses
- Disposable plastic/latex gloves
- Datasheet
- Pen/pencil

When to Measure:
Monthly from May to October

Definition of Terms

Cold-blooded: Animals whose body temperatures match that of their surroundings. Fish, invertebrates, snakes, frogs and toads are cold-blooded.

Diel: Involving a 24-hour time period.

Diffusion: The movement of molecules, for example oxygen molecules, from an area of higher concentration (e.g. the air) to an area of lower concentration (e.g. the water).

Endpoint: The completion of a chemical reaction. It is often determined by the change in color of an indicator solution.

Floc: Short for flocculent precipitate. These fine, suspended particles look like heavy snow.

Photosynthesis: The process in which green plants convert carbon dioxide and water, using the sun's energy, into simple sugars and oxygen.

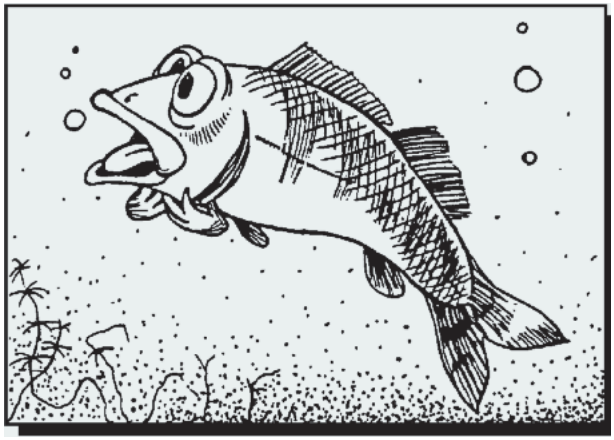
Respiration: The cellular process in which plants and animals use oxygen and release carbon dioxide. Basically, it is the reverse of photosynthesis because carbon dioxide, water and energy are released in the process.

Supersaturation: An indication that more oxygen is dissolved in water than would be in a state of equilibrium. Supersaturation could indicate that some processes are affecting the water's natural balance found in the state of equilibrium.

Titrant: The solution of known strength used for measuring the extent of a chemical reaction, in this case it is sodium thiosulfate.

Background on Dissolved Oxygen

Oxygen is a clear, colorless, odorless, and tasteless gas that dissolves in water. Small but important amounts of it are dissolved in water. It is supplied by diffusion of atmospheric (air) oxygen into the water and by production of oxygen from photosynthesis by aquatic plants. Wind, waves, and tumbling water in fast-moving streams increase the rate of diffusion.



Oxygen: Aquatic Life Depends on it

Both plants and animals depend on dissolved oxygen for survivial. Lack of dissolved oxygen can cause aquatic animals (e.g. fish, macroinvertebrates) to quickly leave the area or face death. Under extreme conditions, lack of oxygen can kill aquatic plants and animals. Measuring dissolved oxygen is probably the most significant water quality test to determine suitability of a stream for fish and many other aquatic organisms. However, these measures only provide a snapshot of the oxygen levels at that particular time. Levels can fluctuate widely throughout the day and year. Fish and other organisms have to live and breathe in that water all year long, A short time without oxygen can be fatal.

Dissolved oxygen (D.O.) is reported as milligrams of oxygen per liter of water (mg/L) which can be called parts per million by weight (ppm). Different aquatic organisms have different oxygen needs. Trout and stoneflies, for example, require high dissolved oxygen levels. Trout need water with *at least* 6 mg/L. Warm water fish like bass and bluegills survive nicely at 5 mg/L and some organisms like carp and bloodworms can survive on less than 1 mg/L. Based on this, there are stream classifications in Wisconsin that define the minimum amount of oxygen allowed at a site (see Table 1).

The oxygen demand of aquatic plants and cold-blooded animals also varies with water temperature. A trout uses five times more oxygen while resting at 80° F (26.7° C) than at 40° F (4.4° C).

TABLE 1: *Minimum dissolved oxygen levels allowed for waters with varied classification in Wisconsin.*

Stream Classification	Minimum Dissolved Oxygen Allowed
Trout Waters	6 mg/L (out of spawning season) and 7mg/L (during spring/fall spawning season)
Fish or aquatic life-designated waters	5 mg/L
Limited forage fish waters	3 ml/L
Limited aquatic life waters	1 mg/L

Factors Affecting Oxygen Levels

There are many factors that affect the amount of dissolved oxygen in the water (see inset boxes). A major one is photosynthesis. Aquatic plants produce oxygen by photosynthesis during daylight hours but they also use oxygen for respiration. High day-time levels of D.O. are often countered with low night-time levels (see a sample diel cycle for dissolved oxygen in Figure 1). This is due to respiration of living organ-isms, including fish, bacteria, fungi and protozoans, as well as the cessation of photosynthesis. Wide daily fluctuations of D.O. stress fish and other aquatic animals. Oxygen depletion can occur because of heavy plant growth. Complete depletion of D.O. can some-times be detected with your nose. Anaerobic decay results in a rotten egg smell (hydrogen sulfide gas). However, good management practices such as planting or maintaining vegetation that filters rainwater runoff and shades the water, cooler water temperatures and protecting the stream channel in other ways to maintain or increase turbulence all promote good dissolved oxygen levels.

Factors that could INCREASE the amount of dissolved oxygen in water	Factors that could DECREASE the amount of dissolved oxygen in water
• High atmospheric pressure • Clear water • Photosynthesis • A lot of turbulence/wave action • Cold water • Presence of excessive amounts of plants (during daytime)	• Respiration of animals and plants living in the water • Chemical reactions of the decaying process • Low atmosphere pressure • High levels of turbidity (such as from erosion) • Warm water • Very colored water • Presence of excessive amounts of plants (at nighttime) • Excessive organic materials (such as sewage, manure, or fertilizers)

Percent Saturation

Recording dissolved oxygen differs from other tests in that it requires two distinct calculations. We are interested in both the absolute amount of D.O. (mg/L or ppm) and how close the value is to the equilibrium value for that temperature and air pressure, which is the percentage of saturation. Values between 90% and 110% of saturation are excellent (see Figure at right). Supersaturated (over 100%) values may sound good but they can also indicate problems, such as excessive plant growth. You can assess the range of dissolved oxygen levels that aquatic plants and animals at your stream site must withstand by monitoring twice in one day – early in the morning, just before sunrise, and later in the afternoon when plants have been exposed to the most direct sunlight for an extended period.

Dissolved Oxygen Levels (% saturation)
Excellent: 91 - 110
Good: 71 - 90
Fair: 51 - 70

SOURCE: Field Manual for Water Quality Monitoring (13th Edition)

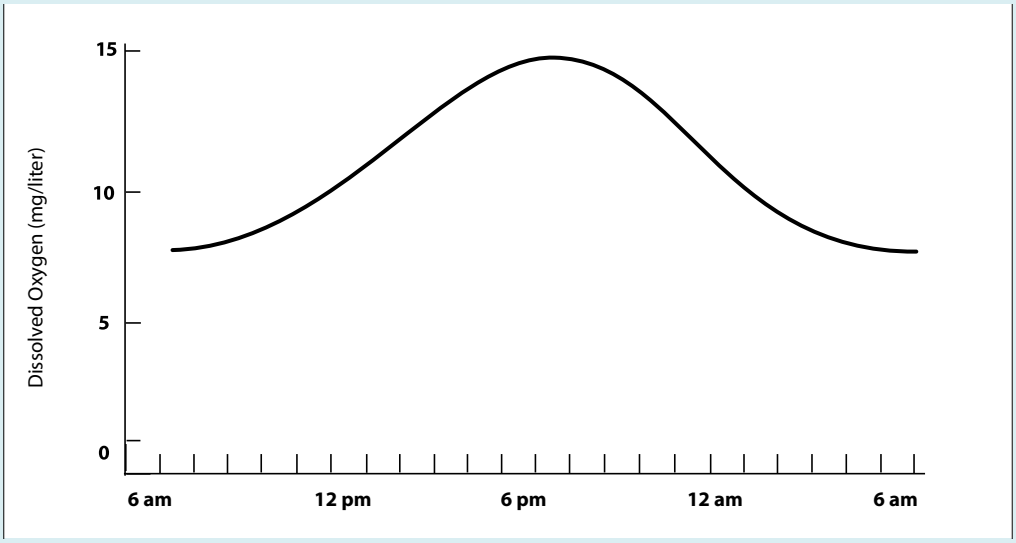


FIGURE 1: Diel Fluctuation in dissolved oxygen

Temperature Conversion Chart

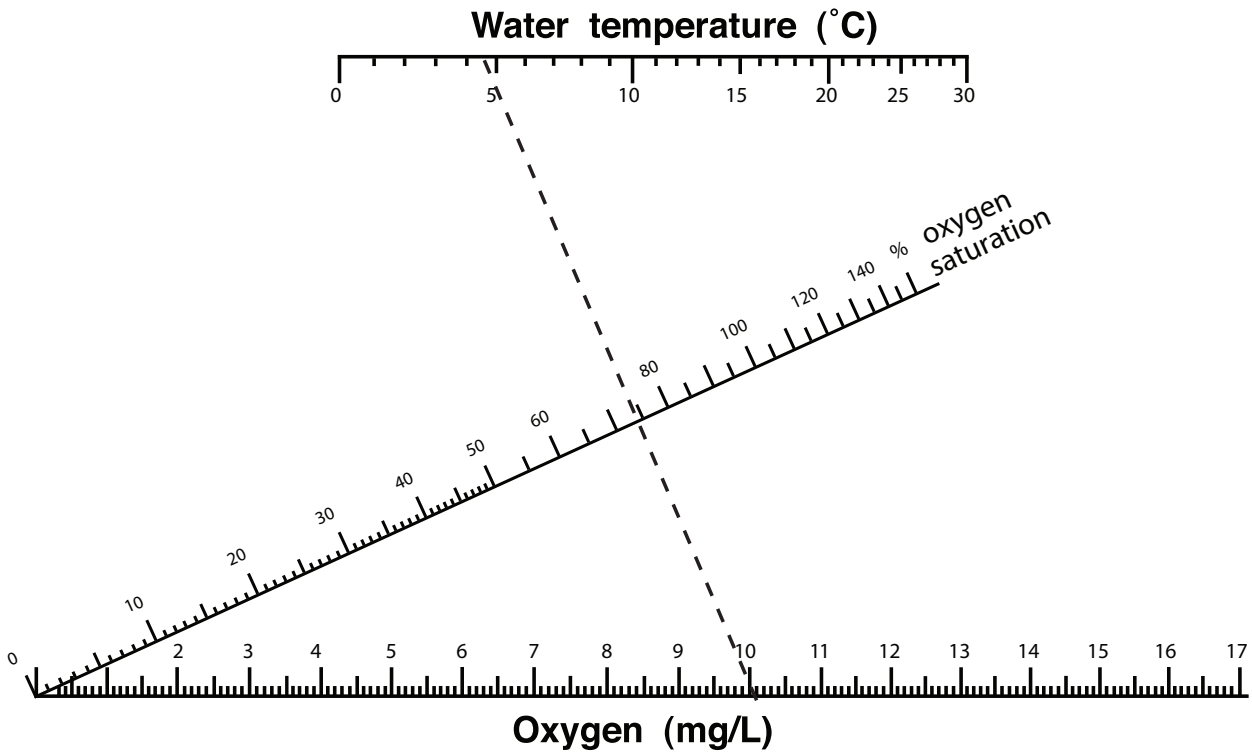
Fahrenheit	33	34	35	36	37	38	39	40	41	42	43	44	45	46	47	48	49	50	51
Celsius	.6	1.1	1.7	2.2	2.8	3.3	3.9	4.4	5	5.6	6.1	6.7	7.2	7.8	8.3	8.9	9.4	10	10.6

Fahrenheit	52	53	54	55	56	57	58	59	60	61	62	63	64	65	66	67	68	69	70
Celsius	11.1	11.7	12.2	12.8	13.3	13.9	14.4	15	15.6	16.1	16.7	17.2	17.8	18.3	18.9	19.4	20	20.6	21.1

Fahrenheit	71	72	73	74	75	76	77	78	79	80	81	82	83	84	85	86	87	88	89
Celsius	21.7	22.2	22.8	23.3	23.9	24.4	25	25.6	26.1	26.7	27.2	27.8	28.3	28.9	29.4	30	30.6	31.1	31.7

How to Find Percent Saturation:

Using a straight edge, find your water temperature (convert from Fahrenheit if necessary using above chart). Align with the oxygen (mg/L) scale. The measured percent saturation is on the point where the line connecting those two points crosses the oxygen saturation line. For example, 5˚C with 10 mg/L of oxygen aligns with 75% saturation, which is your answer.



Collecting the Sample

Remember that photosynthesis and respiration will continue after a sample is collected, so water can gain or lose oxygen while sitting in the sample bottle. Therefore, you should begin D.O. testing immediately upon reaching the shore after you have collected the sample.

You should measure water temperature at the same time and location that you collect the D.O. sample.

Think Like a Scientist!

Follow the directions VERY CAREFULLY! Accuracy is a must for valid data comparisons.

1. Use bottle with the stopper included in the Hach or LaMotte kit.
2. Collect your sample in roughly one-foot deep, normally moving water.
3. Facing upstream, slowly lower the bottle so opening of the bottle faces away from you and water current is entering the bottle.
4. Allow the bottle to fill with water gradually turning it to allow air bubbles to float out.
5. Cap bottle while still submerged and leave extra water in the neck of the bottle.
6. When lifting the bottle out of the water, look for bubbles. If you see any, take another sample using the same procedure.

Testing for Dissolved Oxygen

Using the Hach D.O. Test Kit (Model OX-2P)

Note: If you see any air bubbles trapped in the sample bottle during steps 2-4 below, discard the sample and start over.

1. Put on protective gloves and safety glasses. If your skin comes in contact with any powder or titrant, rinse the area liberally with water.
2. Remove the stopper and add the contents of D.O. powder pillow #1 (manganous sulfate powder) and D.O. powder pillow #2 (alkaline iodide azide powder) to the sample.
3. Insert the stopper, being careful not to trap an air bubble and shake vigorously, holding on to the top. If oxygen is present, a brownish-orange floc will form.
4. Allow the sample to stand until the floc settles halfway. Shake the bottle a second time and allow the floc to settle halfway again.
5. Remove the stopper and slowly add the contents of D.O. powder pillow #3 (sulfamic acid).
6. Stopper and shake vigorously until the acid is dissolved. The yellow color is from iodine. This is called the prepared sample. Prepared samples can be stored in the dark for a short time if it is more convenient or comfortable to return to your home/school to complete the analysis.
7. Transfer two plastic measuring tubes full of prepared sample to the square glass mixing bottle (your Hach kit instructions probably say one measuring tube full). Using two measuring tubes allows you to determine D.O. to the nearest 0.5 mg/L instead of 1 mg/L .
8. ***a)*** Holding the dropper vertically, add one drop at a time of **sodium thiosulfate** standard solution titrant to the square mixing bottle, and count each drop.
   b.) Swirl the solution after each drop.
   c.) Continue adding sodium thiosulfate drops until the sample is a very light yellow.
   d.) Pause and add 3 drops of **starch solution**. Do not include these drops in your count. The prepared sample will turn blue from the added starch solution. If you do not have starch solution, proceed with the next step but be aware that your sample will turn from yellow to colorless instead of blue to colorless.
   e.) Continue adding **sodium thiosulfate** drops, mixing and counting until the prepared sample turns from blue (or yellow) to colorless (the end point). Often this is just one or two more drops, so be careful.
9. The dissolved oxygen content of the water in mg/L is the total number of drops of titrant used to get to the endpoint divided by two if two measuring tubes of prepared sample were used. If only one measuring tube of prepared sample was used, the dissolved oxygen content is equal to the number of drops of titrant. Example: If you used two tubes of sample, you need to divide by two (13 drops divided by two tubes = 6.5 mg/L). If you only used one tube of sample, it's the actual number of drops of titrant used (6 drops with one tube = 6 mg/L)
10. Report dissolved oxygen (mg/L) on the datasheet.
11. Use instructions and chart on Page 4 to convert D.O. to % saturation. Report % saturation on the datasheet.

Using the LaMotte Test Kit Model 7414 or 5860

A) Fix your sample

1. Put on protective gloves and safety goggles. If your skin comes in contact with any powder or titrant rinse the area liberally with water.
2. Holding the reagent bottle completely upside down, add 8 drops of Manganous Sulfate solution (labeled "1" on bottle).
3. Holding the reagent bottle completely upside down, add 8 drops of Alkaline Potassium Iodide Azide (labeled "2" on bottle).
4. Cap and shake the bottle for 30 seconds. A white to brownish orange floc will cloud the sample bottle. Let the floc settle until the top half of the bottle is clear.
5. Shake the bottle again. Allow the floc to settle again.
6. Add 8 drops of Sulfuric Acid 1:1 (red cap on bottle) and shake for 30 seconds. The solution will turn from cloudy to clear in color (If you still see some dark "pepper specks" in the solution add 1 more drop). Your sample is now "fixed".
7. Pour your fixed sample into the graduated cylinder to the 20 ml mark and then pour it into the titration vial (glass vial labeled code 0299).

B) Prepare to titrate

1. Pick up the plastic titrator syringe (labeled code 1649) and push in the plunger to expel air.
2. Put the tip of the titrator syringe into the hole in the top of the titrating solution (bottle labeled Sodium Thiosulfate 0.025N). Fill the syringe by turning the bottle upside down and slowly pull back on the syringe plunger until the tip on the bottom of the plunger is well past the zero mark on the scale on the titrator. You may have to push the plunger in and out a few times to get rid of any air bubbles in the syringe.
3. Turn everything right side up.
4. Slowly push the plunger until the large ring on the plunger of the plastic titrator syringe is right at the zero mark on the titrator.
5. Remove the titrator from the sodium thiosulfate bottle.

C) Titrate the sample

1. Put the tip of the titrator into the opening on the plastic cap of the titration vial (code 0299) that contains your fixed sample.
2. Add the titrating solution one drop at a time by gently pushing the plunger. Swirl the solution between drops until the sample has turned pale yellow. If your solution is already pale yellow skip this step. If your solution is colorless you have zero mg/l dissolved oxygen (if this is the case you can proceed to step 24 for confirmation, if you like).
3. Pop off the plastic cap from the titration vial with the titrator still in the hole without moving the plunger in the syringe.
4. Add 8 drops of starch indicator solution to the pale yellow sample in the titration vial. The sample should now turn deep blue or black.
5. Put the cap back on the titration vial.
6. Swirl to mix the contents.
7. Continue to add sodium thiosulfate one drop at a time, swirling the solution between each drop Observe the color change from dark blue to light blue.
8. Stop when the solution turns from pale blue to colorless. (If no color change occurs by the time the plunger tip reaches the bottom of the scale on the titrator, refill the titrator by filling with titrant to the zero mark and continue the titration. Include both titration amounts in the final test results.)
9. Read the test result directly from where the scale intersects the ring of the plunger for plastic titrator. The titrator is marked at 0.2 ppm increments. So if the titrator ring is touching the third line below the line marked "7" the result would be 7.6 mg/l dissolved oxygen. (If the titrator has been refilled once before, the result would be 17.6 mg/l dissolved oxygen.)
10. Report dissolved oxygen (mg/L) and temperature on the recording form.
11. Use instructions and chart below to convert D.O. to % saturation. Report % saturation on the recording form.

-Modified from URI Watershed Watch

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Water Action Volunteers is a cooperative program between the University of Wisconsin-Madison Division of Extension and the Wisconsin Department of Natural Resources. For more information, go to https://wateractionvolunteers.org/

Streamflow:
Flow Speaks Volumes

Volunteer Monitoring Factsheet Series 2023

Why are we concerned?

Streamflow, or discharge, is the volume of water moving past a cross-section of a stream over a set period of time. It is usually measured in cubic feet per second (cfs). Streamflow is affected by the amount of water within a watershed, increasing with rainstorms or snowmelt, and decreasing during dry periods. Flow is also important because it defines the shape, size and course of the stream. It is integral not only to water quality, but also to habitat. Food sources, spawning areas and migration paths of fish and other wildlife are all affected and defined by streamflow and velocity. Velocity and flow together determine the kinds of organisms that can live in the stream (some need fast-flowing areas; others need quiet, low-velocity pools). Different kinds of vegetation require different flows and velocities, too.

Time Needed:
30 minutes

When to Measure:
Monthly from May to October

Equipment Needed:
- Tape measure
- Yardstick or marked D-frame net pole
- Surveying flags/ flagging
- Float (please use a tennis ball with a small amount of water in it)
- Net (can use D-frame net to catch the ball)
- Stopwatch
- Calculator
- Datasheet
- Pencil
- Hip boots or waders
- Stakes (optional)

Streamflow is affected by both forces of nature and by humans. In undeveloped watersheds, soil type, vegetation, and slope all play a role in how fast and how much water reaches a stream. In watersheds with high human impacts, water flow might be depleted by withdrawals for irrigation, domestic or industrial purposes. Dams used for electric power generation may affect flow, particularly during periods of peak need when streamflow is held back and later re-leased in a surge. Drastically altering landscapes in a watershed, such as with development, can also change flow regimes, causing faster runoff with storm events and higher peak flows due to increased areas of impervious surface. These altered flows can negatively affect an entire ecosystem by upsetting habitats and organisms dependent on natural flow rates. Tracking streamflow measurements over a period of time can give us baseline information about the stream's natural flow rate.

Definition of Terms

Discharge: Another term for streamflow, or the volume of water moving past a designated point over a set period of time

Flow Regime: The pattern of streamflow over time, including increases with stormwater runoff inputs and decreases to a base-flow level during dry periods.

Impervious Surface: A surface that does not allow water (e.g., rain) to pass through (infiltrate).

Rating Curve: A graphical representation of the relationship between the stage height and the discharge (flow).

Run: An area of a stream that has swift water flow and is slightly deeper than a riffle (a run will be about knee/thigh deep).

Stage Height: Height of the water in a stream above a baseline.

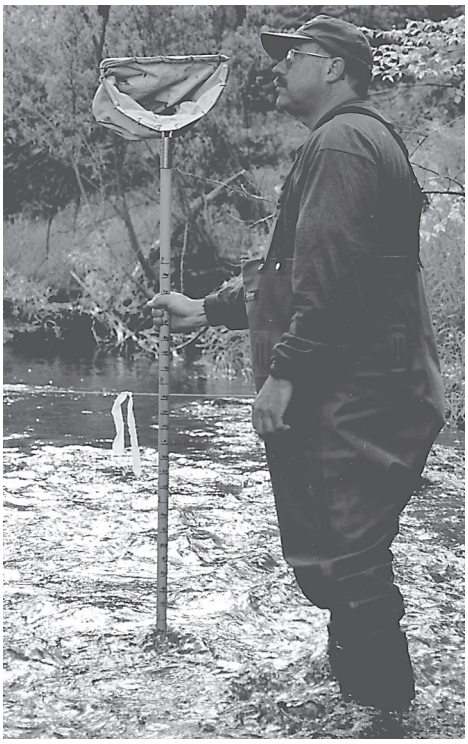
Watershed: An area of land that drains to a main water body.

Safety Considerations

You will need to enter the stream channel to make width and depth measurements and to calculate velocity. Be aware of stream velocity, water depth, and bottom conditions at your monitoring site. Do not attempt to measure streamflow if water velocity appears to be fast enough to knock you down when you are working in the stream. If you are unsure of water depth across the width of the stream, be sure to proceed with caution as you move across the stream, or choose an alternate point from which to measure streamflow.

Determining Streamflow (Area x Velocity = Flow)

The method you are going to use in determining streamflow is known as a velocity-area approach. The task is to find out the volume of water in a 20-ft. (at least) section of stream by determining both the stream's velocity and the area of the stream section. You will first measure the width of the stream, and then measure water depth at a number of locations across the width to find the average depth at your monitoring site. Then by multiplying the average depth by the width, you can determine the average cross-sectional area (ft^2) of the stream. Water velocity (ft/sec) is determined simply by measuring the number of seconds it takes a float to travel along the length of stream you are studying. Since water velocity varies at different depths, (surface water moves more quickly than subsurface water because water moving against rough bottom surfaces is slowed down by friction) you will need to multiply velocity by a correction factor to adjust your measurement to account for the effect of friction. The actual equation you will use to determine flow is this: Flow=Area x Corrected Velocity. This method was developed and adapted from several sources (see bibliography).



Measuring and Calculating Streamflow

Site location

1. At your monitoring site, locate a straight section of stream that is at least 20 feet in length and has a uniform width. The water should be at least 6 inches deep, and have some movement. Unobstructed runs or riffles are ideal sites to choose.
2. Measure 20 feet along the length of your chosen stream segment with your measuring tape and mark both the up and downstream ends of the section with flagging.

Width and depth measurements

3. Working with a partner, measure stream width (wetted edge to wetted edge) by extending a measuring tape across the stream at the midway point of your marked stream segment. Record the width in feet on your recording form. (Using a tape measure graduated in tenths of feet will make calculations easier.)
4. Secure the measuring tape to both shores so that the tape is taut and above the surface of the water. You might choose to attach the tape to two stakes secured on opposite banks to create a transect line across the stream if it is impractical to secure the tape using shoreline vegetation (Figure 1).
5. Using your yardstick or pre-marked D-frame net pole, measure the water depth (ft) at one-foot intervals across the stream where you measured width (and secured the measuring tape). Be sure to measure depth in tenths of feet, not in inches (see conversion chart from inches to tenths of feet on data recording form). Record depth measurements (ft) on the datasheet. If your stream is greater than 20 feet wide, measure depth in 20 equal intervals across the stream.

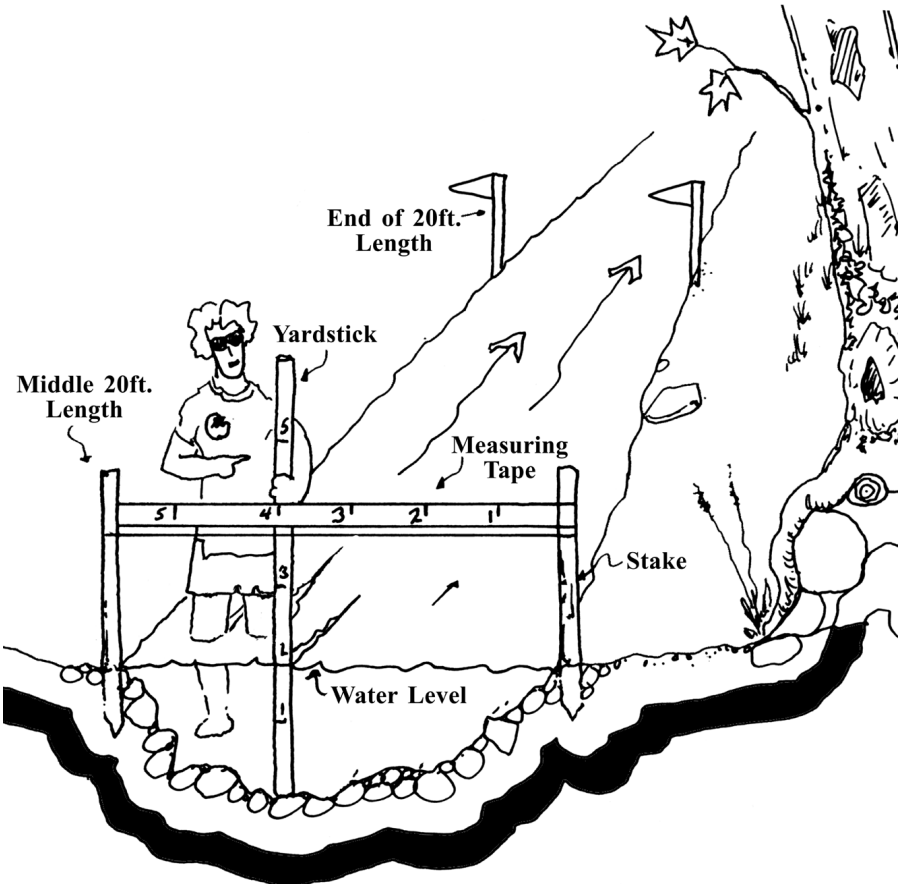


Figure 1. Credit: Chris Padick, Malibu Creek Stream Team

Velocity measurements

Velocity will be measured by tracking the time it takes a floating tennis ball to move the marked 20-foot length of stream. You will time the floating tennis ball (in seconds) a total of four times, at different locations across the stream. Repeating your measurements across the stream, in both slower and faster areas, will help to ensure the closest approximation to the stream's true velocity. This in turn will make your flow calculations more accurate. However, be sure your tennis ball travels freely downstream (during every float trial) without catching in slack water areas of the stream.

6. Position the person who will release the tennis ball upstream from the upper flag. Position the time-keeper on the stream bank (or out of the main flow path) at the downstream flag with the stopwatch. Position the person who will catch the floating tennis ball downstream from the timekeeper (Note: Unless velocity is very fast, the timekeeper should be able to catch the tennis ball float with a net after they have finished timing its run down the stream).
7. The float-releaser will gently drop the float into the stream a few feet upstream from the upper flag, and will alert the timekeeper to begin timing as the float passes the upstream flag (the float should have time to get up to speed by the time it passes the upper flag into the marked length of stream). If the float gets stuck on a log, rock or other obstruction, it should be released from the starting point again.
8. The timekeeper should stop the stopwatch as the float passes the down-stream flag and retrieve the float using the net.
9. Record the float time for the first trial on the recording form.
10. Repeat steps 7-9 for each of the remaining float time trials in different sections of the stream. Record the float time (seconds) for each trial on the recording form.

Determine the Correction Factor

To account for the effects of friction with the stream bottom, select the correction factor that best describes the bottom of your stream:

a. Correction factor for rough or loose rocks, coarse gravel or weeds: 0.8
b. Correction factor for smooth mud, sand or bedrock: 0.9

Determining streamflow

The DNR SWIMS online database will calculate streamflow for you when you enter your measured depths, width, assessed length, velocity float times, and chosen correction factor. If you are curious about the answer while in the field, follow steps on the next page to calculate streamflow on your own.

Overestimation of the float method

In 2011-2012 a number of WAV monitors assisted with a WAV study to compare this method of determining streamflow to results obtained using a flow meter. The great news is that a consistent relationship was found between the two methods from very small (<1 cfs) to large (about 125 cfs) streams. Unfortunately, results suggested that this float method overestimates by about 24% on average. The SWIMS online database will automatically correct results using an equation derived from the study, but those carrying out field calculations should remember to reduce their final result by about a quarter.

Calculating streamflow

11. To determine the average depth at the site, first find the sum of your depth measurements. Then divide the sum of the depths by the number of depth measurements (intervals) you made.
12. Next, multiply your average depth by the stream width. This is the average cross-sectional area (ft^2) of the stream.
13. Determine the average float time by first determining the sum of float times measured. Then divide the sum of the times by the number of float time measurements taken.
14. Divide the length of your stream segment (e.g., 20 feet) by the average float time (seconds) to determine the average surface velocity at the site.
15. Multiply your correction factor by the average velocity measurement.
16. Multiply the average cross-sectional area (ft^2) by the corrected average surface velocity (ft/sec) to determine streamflow.

What is a Staff Gage?

A staff gage is a tool that is often used in conjunction with other methods to determine streamflow. It looks like a large ruler placed vertically within a stream in a position least likely to catch floating debris, and that will be stable during high water flows and the winter freeze. Staff gages are calibrated in tenths of feet and allow a monitor to read and record the stage height (the height of water in the stream at a certain level) any time a monitor has the opportunity to visit the stream site. Staff gages are often placed at the stream's edge on a bridge abutment. WAV monitors may choose to place a staff gage at their monitoring site. You may need a permit to do this, however. Contact your local DNR Service Center for more information on permits.

If a staff gage is installed, monitors can simply record the water level on the staff gage without measuring flow. This method will provide added detail when assessing other parameters. However, scores cannot be compared between sites because each reading is germane only to that site.

Monitors may also choose to install a staff gage at their monitoring site and then, at a number of different water levels, record the stage height and determine the flow in the stream by following methods provided in this fact sheet. This type of monitoring is similar to what professionals do to determine a rating curve for a stream discharge monitoring station. The rating curve will reveal the stream's unique relationship between flow and stage height. Eventually, a monitor could determine streamflow simply by reading the stage height on the staff gage and looking at the site's rating curve to see what the flow is at that stage height. Caution must be used with this method since weeds, ice, or other factors can cause ponding of the stream water or movement of the staff gage over time, thus affecting rating curve results.

Bibliography:
We reviewed and adapted information and methods from Missouri Stream Team Program, the WI DNR, the EPA Volunteer Stream Monitoring Methods Manual (EPA 841-B-97-003), the Nohr Network of Monitors, the Washington Co. (WI) Waterways Program, Hoosier Riverwatch, Project SEARCH, and California's Nonpoint Source Pollution Control Program as well as other technical information.

Water Action Volunteers is a cooperative program between the University of Wisconsin-Madison Division of Extension and the Wisconsin Department of Natural Resources. For more information, go to https://wateractionvolunteers.org/

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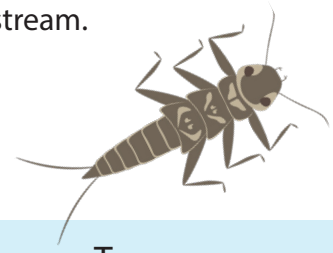
Macroinvertebrate Biotic Index

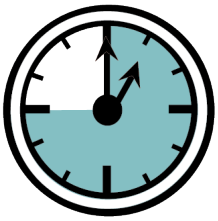
Volunteer Monitoring Factsheet Series 2023

Why are we concerned?

- Aquatic macroinvertebrates are small animals without backbones. Their presence or absence can reflect a stream's general condition.
- Certain macroinvertebrates respond differently to the physical, chemical, and biological conditions within a stream.
- Aquatic macroinvertebrates are relatively immobile so they can't escape either short or long-term pollution exposure. This is important when assessing longterm pollution events within the stream.



Time Needed:
Up to 45 minutes



When:
Twice a year (once in spring, once in fall)

Equipment Needed:
- Hip boots
- D-frame kick net
- White basin (important to have white background-helps to see the critters)
- White ice cube tray
- Key to Life in the River or other identification tools
- Datasheet
- Pen/pencil
- Magnifying glass
- Picking tools (spoons, tweezers, pipettes)

Definition of Terms

Aquatic Macroinvertebrates: Small animals without backbones that live in water and are visible to the human eye.

WAV Biotic Index: Water Quality Index for Wisconsin wadable streams using aquatic macroinvertebrates.

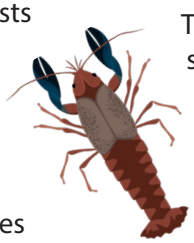
Genus: The category of organisms ranking below the family category, but broader than the species category.

Leaf Pack: Bundles of old leaves sticking together in the water.

Riffle: Shallow area in stream where water flows swiftly over rocks.

Background on WAV Biotic Index

From the crayfish burrowing in the stream bed to the tiny aquatic insects skirting the water's surface, streams and rivers swarm with life. The inhabitants of this living place are affected by poor water quality just like humans are affected by an unhealthy environment. However, scientists have found that not all aquatic organisms react the same to poor water quality. Some species are pollutant-tolerant while some are very pollutant-sensitive. From this knowledge, a scale was developed to determine water quality based on the types of life found in the water. For example, streams with primarily pollutant-tolerant organisms generally have poorer water quality than those streams with many pollutant-sensitive animals. This is because poor-quality streams gradually lose pollutant-sensitive animals until only the pollutant-tolerant species are left.



How the Biotic Index was Developed

A number of years ago, a highly respected researcher developed what is known as the Hilsenhoff Biotic Index (HBI). This index identified organisms down to the genus or species. Only experts in aquatic biology use this index to determine water quality. Although very accurate, the HBI is difficult to use outside of a lab setting, so a less complex index called the Family Biotic Index (FBI) was developed. With this index, aquatic animals are identified to the family level, which is a less specific level than genus or species. Training is necessary for scientists to use the FBI accurately.

A third index was developed so volunteers could be more involved with identifying stream health based on biotic indices. A group of Wisconsin scientists from the DNR, University of Wisconsin-Madison Division of Extension, and the University designed the WAV Biotic Index that correlates closely to the Hilsenhoff Biotic Index, but with less scientific detail. This index was created specifically for streams in Wisconsin. Monitoring groups are strongly encouraged to use this educationally focused biotic index.

A healthy stream will have many different organisms, both pollutant-tolerant and those sensitive to pollution.

Although relatively accurate in assessing stream conditions, the biotic index does have its limitations. The biotic index can indicate a problem, but it cannot specify what that problem might be. For example, manure, sewage, fertilizers, sediment and organic materials all negatively impact water quality. In order to pinpoint these possible pollutant sources, monitoring for other parameters such as habitat assessment, dissolved oxygen and temperature needs to be done. The biotic index is useful for identifying long-term pollution problems, since these organisms carry out a portion or all of their life cycle in streams. Other parameters monitored in the WAV program (except habitat) only indicate the water quality conditions at the time of testing.

Selecting a Sample Site

You will collect a total of three biotic index samples within a 300' stream section. Rocky bottom and soft bottom streams support different kinds of organisms, so be sure to choose sites based on your stream type. Your goal is to collect as many different kinds of aquatic macroinvertebrates from as many different habitats as necessary to ensure an accurate site assessment. Be aware that each habitat type has different sampling protocols and some have a greater diversity of organisms than others. If you have many habitats from which to choose, consider sampling from those with the most diversity (i.e. riffles). If your stream has a rocky bottom, sample at two separate riffle areas and at one other habitat. If your stream has a soft bottom or does not have riffles, collect samples at submerged logs, snags or undercut banks.

Habitat Type	Stream Type	Habitat
Riffles	Rocky Bottom	Most Diverse
Undercut Banks	Rocky, soft bottoms	
Snag areas, tree roots	Rocky, soft bottoms	
Leaf packs	Rocky, soft bottoms	Least Diverse

How to Collect Samples at The Different Sample Sites

You will collect three total Biotic Index samples. However, before you begin rinse the net and check to make certain it doesn't contain any debris from the last time it was used. Fill your white basin with about one inch of clean stream water. When sampling, if you find you have too much water or if the water is too muddy, pour the excess/muddy water through your net. If necessary, add some clean water to the original sample.

Riffle Sampling

1. You will collect one sample in the upstream portion and one in the downstream portion of the riffle. The two samples constitute ONE BIOTIC INDEX SAMPLE.
2. Start at the downstream section of the riffle.
3. Place the net firmly on the bottom of the stream standing in front of the net so the water passes you first, then flows through the net. If a second person is with you, this person should act as a time keeper and kicker.
4. When the net is in place use your feet to kick the rocks for two minutes to dis-lodge aquatic macroinvertebrates. Alternatively, pick up each rock within an 18 inch-square area immediately in front of the net and rub thoroughly to remove all organisma clinging to it. Gently replace the rocks in the stream outside of the sample location. Continue to pick up, rub, remove rocks for two minutes.
5. Carry the net to shore and dump the contents into one basin or bucket with about one inch of water in it.
6. All organisms clinging to the net should be removed and placed in the basin.
7. Repeat steps 3-6 for the upstream portion of the riffle. Combine contents of the second sample with the first.
8. Examine the sample and check the debris for any macroinvertebrates that might be hiding.
9. Remove large leaves, sticks, rocks, plants and other debris and place them in another container to check later for organisms that crawl out.
10. You now have a biotic index sample.
11. Combine this sample with the other samples taken from riffles or other habitats.
12. When you have three biotic index samples, follow the steps on the next page to categorize your sample and determine water quality.

Think Like a Scientist!

Follow the directions VERY CAREFULLY! Accuracy is a must for valid data comparisons.

Sampling Undercut Banks

1. Undercut banks have scooped out areas just below the surface of the water. This creates a bank that slightly overhangs on the surface of the water and habitat for many kinds of organisms.
2. Facing the bank, move the net in a bottom-to-surface motion, jabbing at the bank vegetation to loosen organisms. Jabbing the net about 20 times should provide you enough organisms for your sample.
3. Carry the net to shore and dump the contents into a basin or bucket with about one inch of water in it.
4. All organisms clinging to the net should be removed and placed in the basin.
5. Examine the sample and remove any large leaves, sticks, rocks, plants and other debris. Check the debris for any macroinvertebrates that might be hiding in it.
6. Place the debris in another container to check for organisms that may crawl out later.
7. You now have a biotic index sample.
8. Combine this sample with the other samples taken from undercut banks or other habitats.
9. When you have three samples, follow the steps on the next page to categorize your sample and determine water quality.

Sampling Snag Areas, Tree Roots, and Submerged Logs

1. Snag areas are accumulations of debris caught behind logs, stumps or boulders in the water.
2. Select a three-foot by three-foot area (for uniform comparisons) around the snag, tree roots, logs or other debris.
3. Scrape the surface of the tree roots, logs or other debris with your net. You can also disturb the surfaces by scraping them with a stick, hands or your foot, or you can pull off some of the bark to get at organisms hiding underneath. Like with undercut bank sampling, 20 jabs equals one sample.
4. To remove sediment, swirl the net in the stream, being careful to keep the opening out of the water so you don't lose any organisms.
5. Carry the net to shore and dump the contents into a basin or bucket with about one inch of water in it.
6. All organisms clinging to the net should be removed and placed in the basin.
7. Examine the sample and remove any large leaves, sticks, rocks, plants and other debris. Check the debris for any macroinvertebrates that might be hiding in it.
8. Place the debris in another container to check for organisms that may crawl out later.
9. You now have a biotic index sample.
10. Combine this sample with the other samples taken from snags or other habitats.
11. When you have three samples, follow the steps below to categorize your sample and determine water quality.

Leaf Pack Sampling

1. Look for old leaf packs that are about four to six months old. Old leaf packs are dark brown, slimy and slightly decomposed.
2. Position the dip net downstream from the leaf pack. Use your feet or hands to gently move the leaf pack into the net.
3. Swirl the leaf pack in the net, knocking off some of the aquatic macroinvertebrates.
4. Carry the net to shore.
5. Hold the net near the basin and take out the leaves one at a time to inspect for organisms. Remove any macro-invertebrates that you find, and place them in the basin.
6. Once you've finished with the leaves, remove any organisms that are clinging to the net.
7. Place the leaves in another container to check for organisms that may crawl out later.
8. You now have a biotic index sample.
9. Combine this sample with the other samples taken from leaf packs or other habitats.
10. When you have three samples, follow the steps below to categorize your sample and determine water quality.

How healthy is the stream?

	Score
Good	2.6 - 3.5
Fair	2.1 - 2.5
Poor	1.0 - 2.0

After Collecting Your Samples:

- Check the basin with the debris to see if any aquatic macroinvertebrates crawled out. Add these animals to your prepared sample.
- Fill the ice cube tray half-full with stream water.
- Using the picking tools, sort out the macroinvertebrates and place ones that look alike together in their own ice cube tray compartments. Sorting and placing similar looking macroinvertebrates together will help insure that you find all varieties of species in the sample.
- Refer to the Key to Macroinvertebrate Life in the River or other tools to identify the aquatic macroinvertebrates.
- On your datasheet, circle the animals that you found in your sample. Then use the online database or follow steps on the Biotic Index datasheet to calculate a water quality score.
- Safely return all macoinvertebrates to the stream and **thoroughly clean, drain and dry your equipment to avoid transporting aquatic invasive species to other waters**.

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Water Action Volunteers is a cooperative program between the University of Wisconsin-Madison Division of Extension and the Wisconsin Department of Natural Resources. For more information, go to https://wateractionvolunteers.org/