Field Manual

University of Florida
Gainesville, Florida

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If found, please return this document to Martin Group –
Kangerlussuaq International Science Support
Kangerlussuaq, Qeqqata, Greenland

Contents

Introduction

This document provides information for activities in two linked projects, one supported by the University of Florida, Water Institute Graduate Fellows: “High Latitude Hydrology” (WIGF), and the other supported by NSF: “Significance of Ice-loss on Landscapes in the Arctic” (SILA). The first part of the document includes descriptions of procedures to use for field sampling, observations, and experiments. Although these descriptions are thorough, they are not exhaustive. In most cases, the procedures will have a team leader in the field who will have previously completed the procedures. Consequently, the instructions should be used as reminder of steps to take while carrying out the procedures.

The second part of the document includes descriptions of Guiding Principles and a Code of Conduct for the WIGF and SILA projects. The combined projects include many people (likely 20 to 30 or more) at various career stages ranging from undergraduates to senior faculty. The large group will have many within group dynamics and interactions with people outside the project participants as they undertake the tasks for the projects. The second part of the document thus lay out guidelines for expected behavior both within and external to the group. The end of this section includes contact information if misbehaviors require reporting.

The third part of the document includes maps and digital elevation models of our field areas. These maps also include sites where samples have been collected previously in our earlier work. They should be used to provide an overview of the terrain, stream locations, topography etc. Use proper maps if you plan to go on extended hikes.

Stream gauging and sampling

Overview

Stream gauging stations will be set up during the initial field sampling period and subsequent measurements will simply repeat the initial measurements. The following instructions for how to set up the stations are included just to provide information on how the stations were initially created. This information will also be important in the event a station needs to be re-established.

The idea behind stream gauging is to measure the average velocity within a sequence of rectangular sections of a stream. The product of the measured velocity (L/t) with the area of the rectangle (L^2) provides the average discharge through each rectangle (L^3/t). The sum of discharge from all of the rectangles provides the total discharge through that cross section of the stream.

Setting up the gauging stations

It simplifies the procedure if a stream cross section can be found that has characteristics to allow creation of a set of rectangles that are uniform widths. Once the section of the stream is identified, it is unlikely to change during the course of our project. Some of the sites have been identified and previously measured (Sisimiut 1 – three separate locations - and Sisimiut 3) but other have not (Lake Helen and on the ice). Identification of those sites will be one of our first tasks after arriving in Greenland.

The sites will have a configuration something like what is shown in Figure 1. The stream dimension are: depth (d), which is variable and width (w), which will not vary much and consequently, once the benchmark and locations of rectangles are established, we will consider it to be constant. The benchmark is string or rope that is stretched tightly across the stream at a constant elevation (h) above the stream surface. The string will be leveled by ensuring that h is a constant distance at several locations across the stream. It will also be divided into segments marked by flagging or electrical tape to mark the locations of the velocity measurements – essentially the center of the rectangles. The stream gauging sites will also have a conductivity, temperature, depth logger installed, either at the cross section or at a nearby location, depending on what is available along the stream banks that can be used to fasten a piece of angle iron and the loggers. For the best installation it will be bolted to a rock outcrop, but if no outcrops are available, it may be pounded into the stream bottom sediments.

The procedure for making the velocity measurements is as follows and will be more or less the same in all cases, although with a few minor differences depending on which flow meter is used, as described below. All measurements should be recorded on data sheets that will be provided.

(1) Measure the stream elevation (h), either relative to the string or some other established benchmark such as the mounting post. What makes up the benchmark will depend on the stream segment.

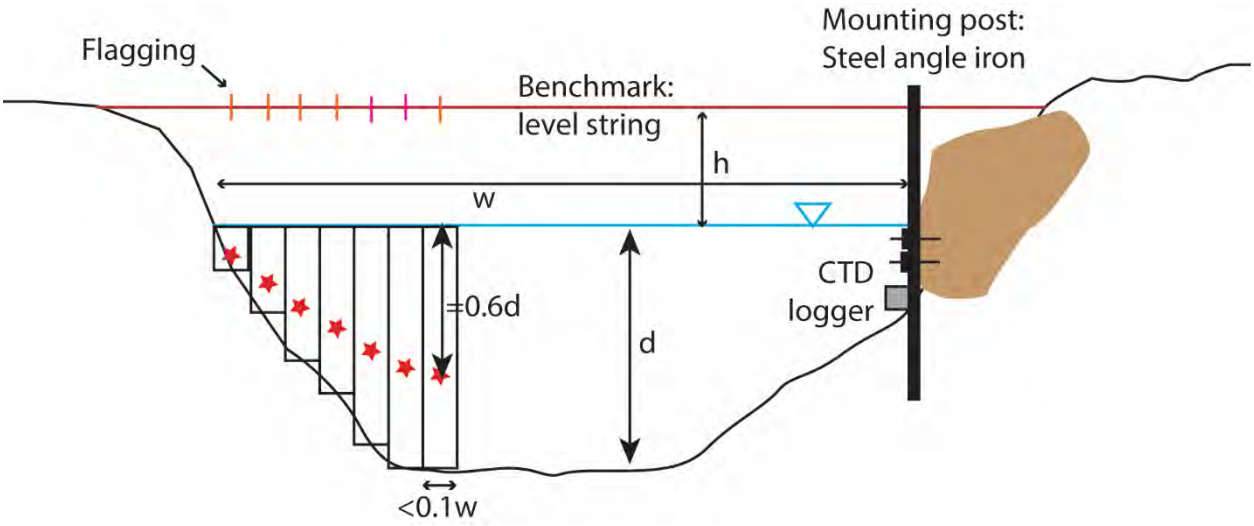


Figure 1. Typical location for a stream gauging cross section for gauging

(2) Ensure that the string benchmark is level across the stream by measuring its elevation above the stream at several locations. If the string is sagging, then tighten it to ensure it is level. Use a truckers hitch to make it as tight as possible.
(3) Initiate the velocity measurements at one side of the stream. Which side to start will be established at each location.
(4) Discharge from each rectangular segment will be measured with the same sequence of events:
    a. Measure the depth of the stream at the center of the rectangle, which is indicated by the flagging tape on the benchmark string
    b. Set the flow meter to a depth of 0.6d below the surface of the stream. The method for setting the depth depends on the type of flow meter.
    c. Follow the protocol for the flow meter to measure the average velocity over the period of measurement at that depth.
(5) Repeat the measurements until velocity at all rectangles have been measured.

Flow Meters

We will use three different types of flow meter. Two are similar – the USGS type AA and USGS Pygmy flow meters. These are the most complicated to use, but are more standard and should be used if possible. They are installed on a wading rod to hold them in place at the proper depth in the stream and will be used to measure stream depth. The third type is a Global Water Flow Probe which can be used to measure stream depth and then set the sensor at the proper depth.

Global Water Flow Probe. This meter will be used for the smaller streams such as S3. It consists of a telescoping hollow tube with depth gradations on the side with an impeller at its based connected to a small computer that plugs into the top (Fig. X). The buttons on the computer are toggle switches that turn on the meter among other functions. The display shows both the instantaneous flow rate as well as an average. To use the meter, plug the computer into the top of the rod. Although the computer is firmly seated into the port, it is just a friction fitting and can fall out if not careful. **Do not drop the computer in the stream.**

Set up a table in the field book following the pattern shown in Table 1. At each rectangular section, measure the distance from the bottom of the stream to the string and the distance from the string to the stream top. The difference is the stream depth. Next calculate the value of 0.6 times the stream depth, add to the distance between the stream top, and set the impeller at that distance away from the string. (This method corrects of change in stream elevation because of the restriction caused by the flow meter). There is an arrow at the impeller housing point in the direction of flow. Be careful that the impeller is pointed parallel with the flow direction and in the correct direction. Stand to the side of the impeller to not block flow. Watch the instantaneous flow rate until it becomes more or less stable, which will happen in just a few seconds. Once that occurs, push the reset button and allow the meter to average the flow for at least 30 seconds. The value for the average flow should not be changing greatly at that point, but if it is still changing wait until it is stable. Report the average flow rate to the note taker.

Table 1. Global Water Meter

Distance from bank (m)	A. Bench to bottom (cm)	B. Bench to Water (cm)	C. Water Depth (cm)	D. 6/10 (cm)	Measurement Depth from Bench (cm)	Ave. Flow speed (ft/sec)
			A-B	C * 0.6	B + D	

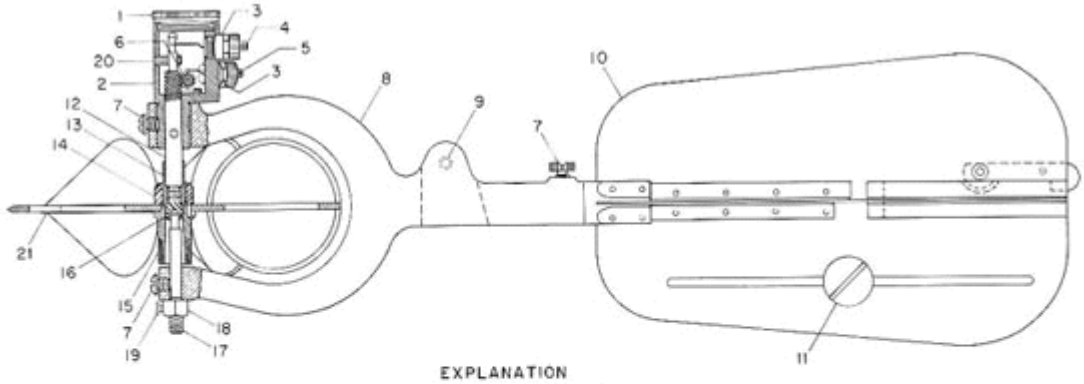
Rickly USGS Type AA Current Meter and Wading Rod. This meter is used with a top-setting wading rod that (1) measures the stream depth, (2) automatically sets the meter at 0.6 the depth of the stream, and transmits the signal from the meter to headphones plugged into the top of the wading rod (Fig. 2). The meter is a set of cups that rotate at speeds proportional to the water velocity (Fig. 3). The number of rotations over a set period of time (usually about 40 seconds) can be converted into the flow velocity using an empirical algebraic formula. **Note: this instrument is delicate and must be treated with care during operation and cleaned, dried, and lubricated at the end of every day it is used.**

Fig. 2. A. The complete assembled wading rod. B. The Vernier scale.

The meter and wading rod will need to be assembled upon reaching the stream gauging site starting with the wading rod. We will have two wading rods with one in metric and the other in English units. Be careful to record units with stream depths. **Do not cross thread any of the screws.** Steps are:Screw together the upper and lower portions of the top and bottom half of the rod. The large hexagonal rod can be screwed together first followed by the smaller round rod. Insert the hexagonal rod through the hexagonal hole in the bottom fitting of the round rod (a torpedo shaped fitting with a wire coming out the top) before screwing the round rod together. The upper hand release (a very hard to push button) will have to be released to rotate the small round rod. Screw the round plate base plate into the threaded section of the bottom of the hexagonal rod. Include the lock washer between the rod and the plate to ensure the plate is securely fastened or the plate may fall off while in the stream and be lost.

(1) Assemble the Type AA flow meter (Fig. 3). Slide the back of the cup assembly on the bottom fitting at the base of the round rod. The cup assembly has a nut on top (7, Fig. 3) that is tightened with a screwdriver to hold the cup assembly on the fitting. Assemble the fins by sliding one over the other. They only go one way so if they don't fit try a different orientation. Flip the lever on the side to hold them together. Attached the wire coming from the rod to either the single contact binding post (4, Fig. 3) if flow is slow, which makes one click per rotation, or the penta contact binding post (5, Fig. 3) if flow

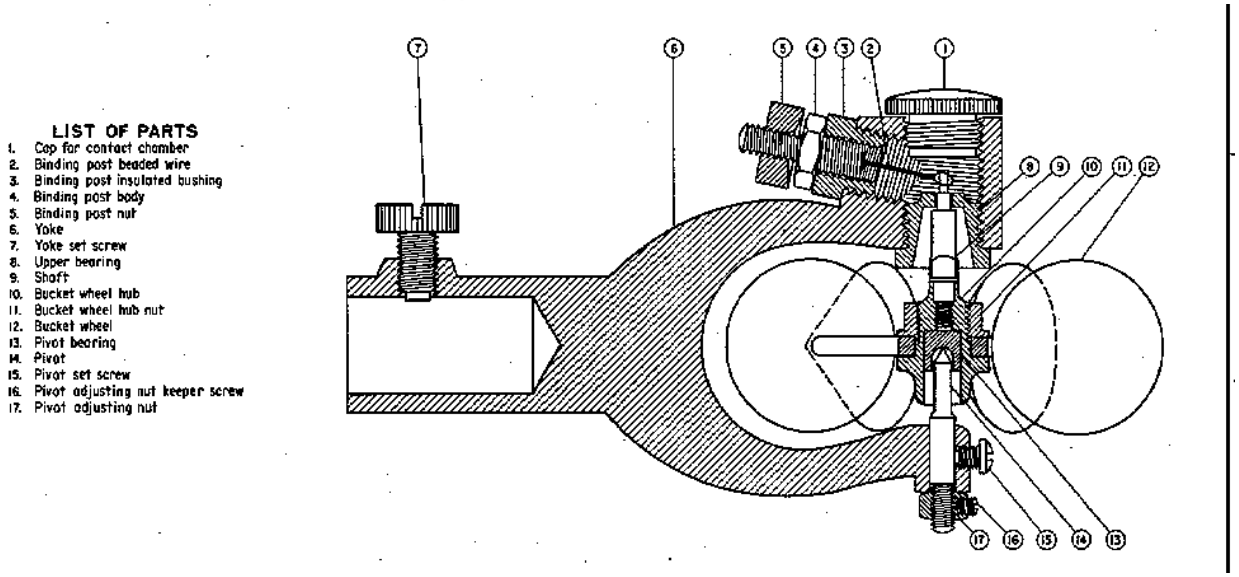
is fast, which makes one click per five rotations. Test that contact is made by spinning the cups and listening for the clicks.



EXPLANATION

1. Cap for contact chamber
2. Contact chamber
3. Insulating bushing for contact binding post
4. Single—contact binding post
5. Penta—contact binding post
6. Penta gear
7. Set screw
8. Yoke
9. Hole for hanger screw
10. Tailpiece
11. Balance weight
12. Shaft
13. Bucket—wheel hub
14. Bucket—wheel hub nut
15. Raising nut
16. Pivot bearing
17. Pivot
18. Pivot-adjusting nut
19. Keeper screw for pivot-adjusting nut
20. Bearing lug
21. Bucket wheel

Figure 3. Type AA current meter



LIST OF PARTS

1. Cap for contact chamber
2. Binding post beaded wire
3. Binding post insulated bushing
4. Binding post body
5. Binding post nut
6. Yoke
7. Yoke set screw
8. Upper bearing
9. Shaft
10. Bucket wheel hub
11. Bucket wheel hub nut
12. Bucket wheel
13. Pivot bearing
14. Pivot
15. Pivot set screw
16. Pivot adjusting nut keeper screw
17. Pivot adjusting nut

(2) Assemble Pygmy meter if using (fig. 4). This meter is similar to the Type AA meter with a couple exception. It has no tail fins so only the cup yoke needs to be attached to the wading rod in the same way that the Type AA meter attaches. It also has only one binding post that measures one click per rotation.

Make measurements

(1) Set up a table in the field book following the pattern shown in Table 2.

(2) Measure the water depth of the first rectangle by reading the depth directly with the scale on the hexagonal rod or on the side of the Global Water meter. On the

wading rod, hash marks occur every 2 cm with 10 cm divisions indicated by double hash marks.

(3) Set the proper depth for the flow meter. The upper handle assembly is a Vernier scale that allows you to automatically set the proper depth (0.6d, figure 1). Line up the proper depth mark for the depth with the tenths of meters of the water depth on the Vernier scale. For example, if the depth is 66 cm, orient the 60 cm mark on the sliding scale with the 6 on the Vernier scale. The meter will automatically be positioned at the correct depth.

Table 2. Price Meter

Rotations per click:

Benchmark: Water level at benchmark

Date/Time:

Starting bank (left or right?). Note, left and right banks are defined looking downstream

Distance from bank (m)	**Water depth (m)**	**# Clicks**	**Seconds**

(4) Be sure to stand to the side and behind the cups to not obstruct flow past the cups. Listen for the click on the head phone to determine the cups are rotating (in most cases you will be able to see them rotate). Signal to the person recording information when you hear a click and start counting clicks, but do not including the click at the start time. That person will then let you know when 40 seconds has passed. After 40 seconds has passed listen for the next click and signal to the recorder when that click occurs. Record the time, number of clicks, and if the clicks represent 1 or 5 rotations of the cups.

(5) Move to the next rectangle and repeat the procedure.

(6) Velocity (V) for each rectangle is related to the number of rotations of the cups by the following relationships

a. For the Price AA meter

$$V=2.2048R + 0.0178$$

b. For the Pygmy meter

$$V = 0.9604R + 0.0312$$

Maintain flow meter

Type AA meter. At the end of the field day, the flow meter needs to be dried and lubricated. Remove the cups from the yoke (8, Fig. 3) by loosening the raising nut (15, Fig. 3; note this nut is reverse threaded) and the set screw (7, Fig. 3). Be sure loosen the set screw and not the keeper screw for the pivot adjusting nut (19, Fig. 3). The keeper screw allows the pivot to be raised and lowered to keep the cups off of the yoke and it should not be moved from its pre-set elevation. After loosening the set screw, the pivot (17, Fig. 3) and cup assembly can be

removed. Make sure the pivot and pivot bearing are clean and dry and put a small drop of the provided oil on the top of the pivot. Reassemble.

Check the contact chamber by unscrewing the cap to see if water got into (1, Fig. 3). If wet, leave open to dry. You may also remove water with Q-tips, but be very careful to not bend or damage the fine wire inside the chamber. If you do not hear clicks the fine wire may have been bent and is not making contact. If that is the case gently bend it to ensure good contact being careful to not break it. The entire cup can be removed by loosening the set screw (7, Fig. 3).

Pygmy meter. A procedure similar to the Type AA meter should be followed at the end of the day, but the pivot can be removed by loosening the Pivot Set screw (4, Fig. 4) and dropping it out of the yoke. The bucket assembly can then be wormed out of the yoke and lubricated. Check for water on the binding post wire by removing the contact chamber cap (1, Fig. 4). Dry as necessary, but again be very careful not to bend or break the binding post wire.

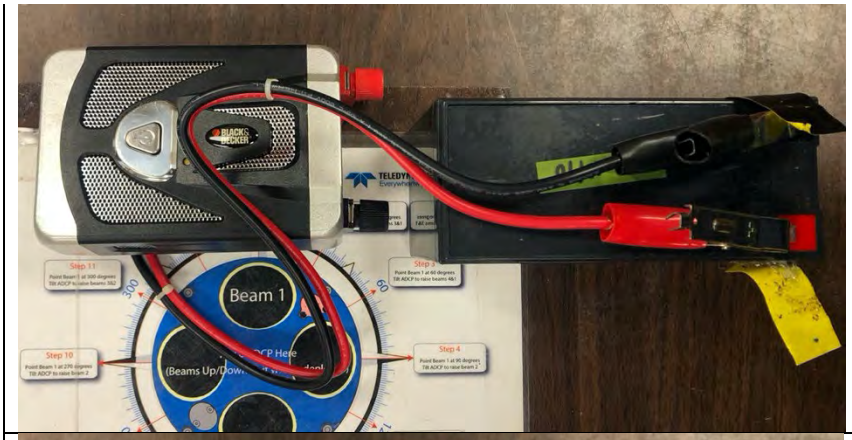
Acoustic Doppler Current Profiler (ADCP) instructions

Check list

• Power sources: • Battery (1) • Power inverter (12V-110V) (2) • Extension cord (3) • Multiconnector (4)	
• Laptop (1) • Laptop power supply (2) • Mouse (3) • Usb-serial adapter (4)	
• ADCP (1) • ADCP communication and power cable (2) • GPS (3) • ADPC Power source (4) •	

Making Connection

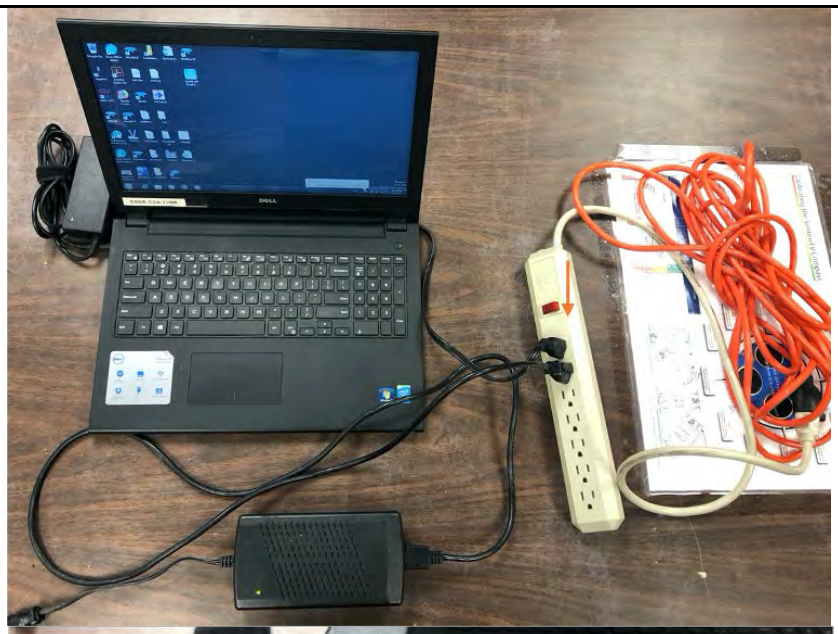
- Connect the battery to the power inverter (red with red, black with black)



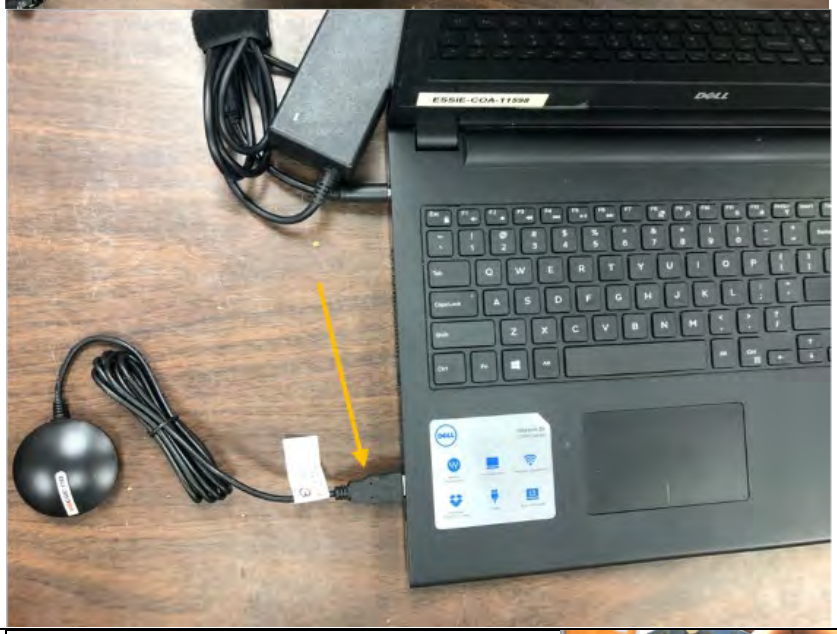
- …and check it is running

- Connect the extension cord to the power inverter (1) and multiconnector to the extension cord. (2)

- Connect computer and ADCP power source to the multiconector (orange arrow)



- Connect the GPS to the computer
- (yellow arrow)

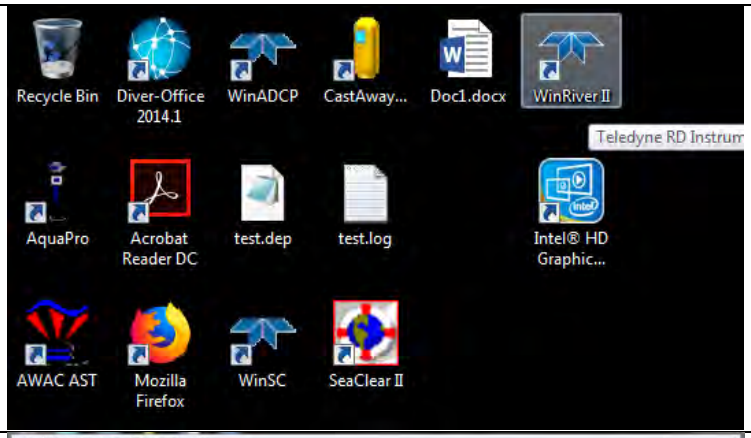


- Take the protector out of the ADCP (at the bottom)

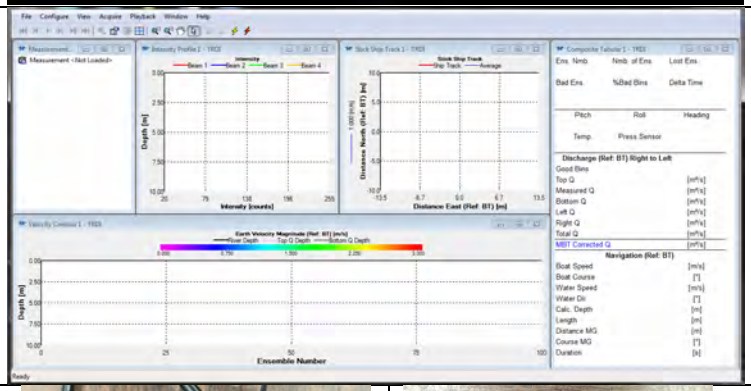
- Connect ADCP serial to USB adapter

ADCP programming with WINRIVER II

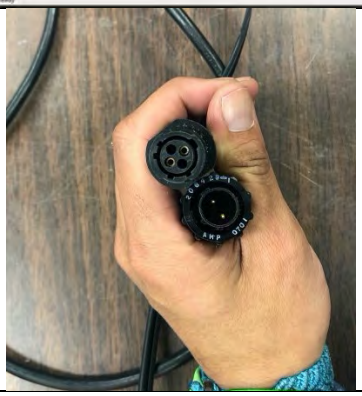
- Open WinRiver II
- (selected icon)

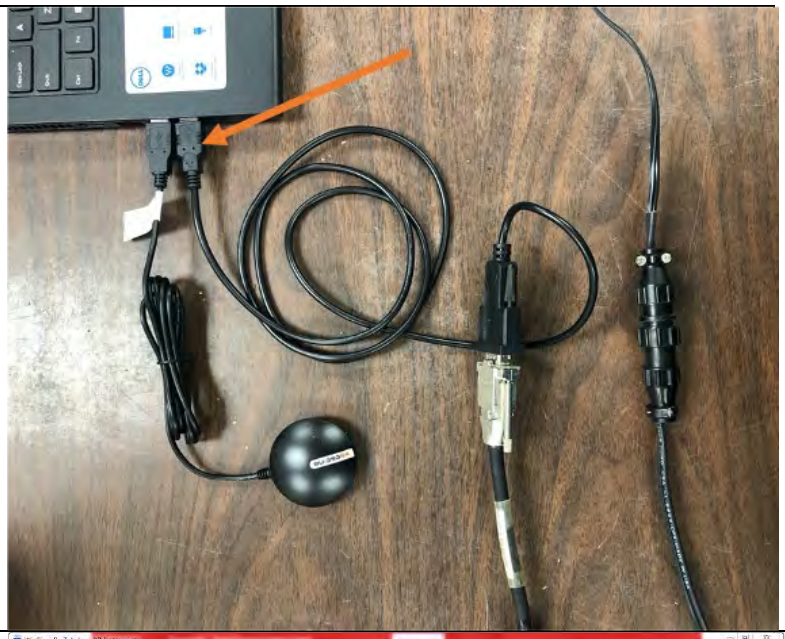
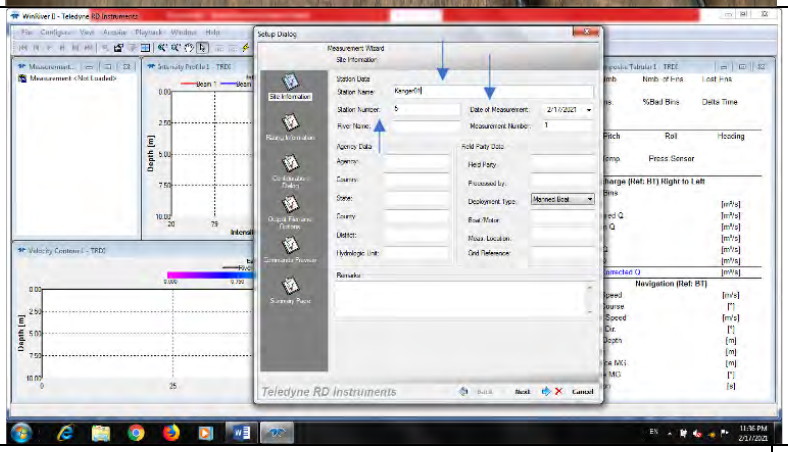
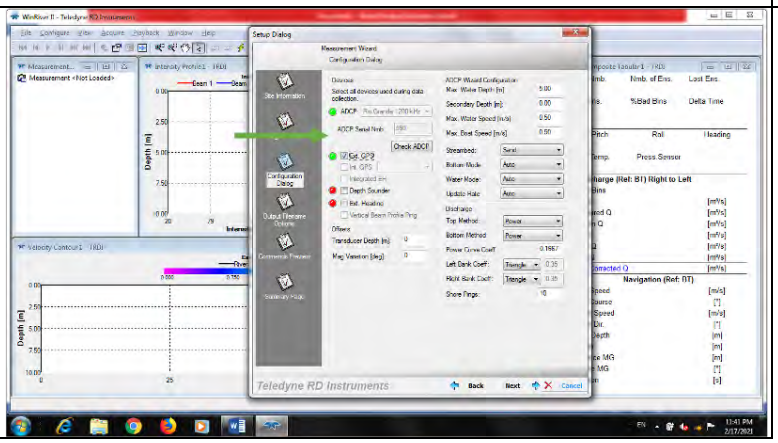


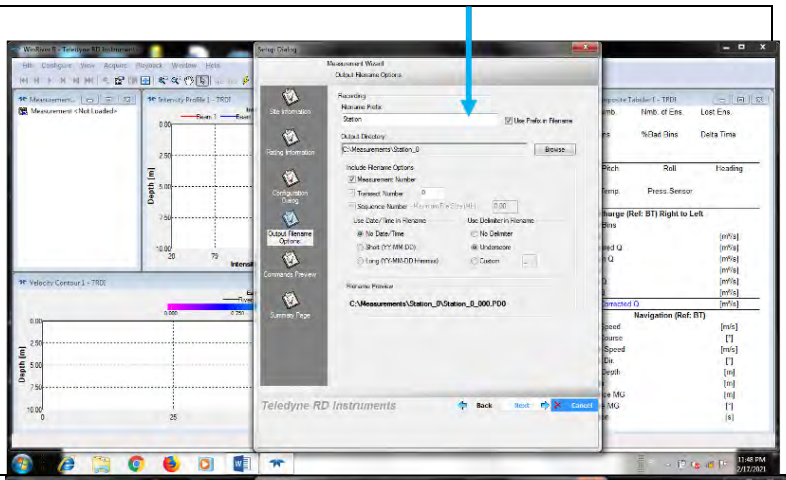
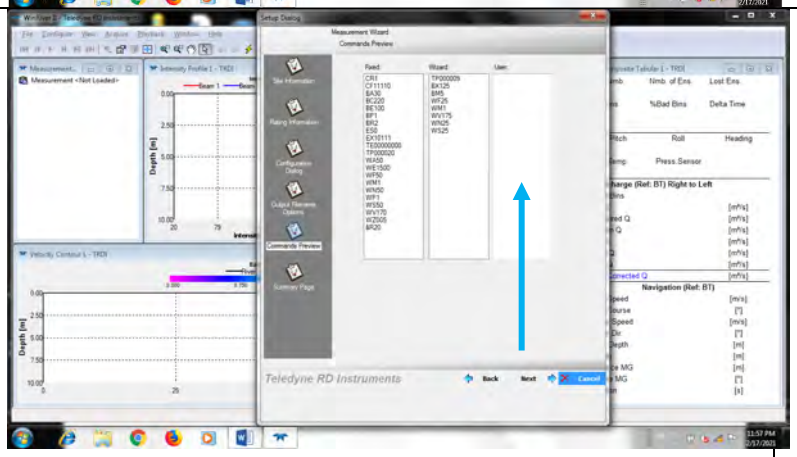
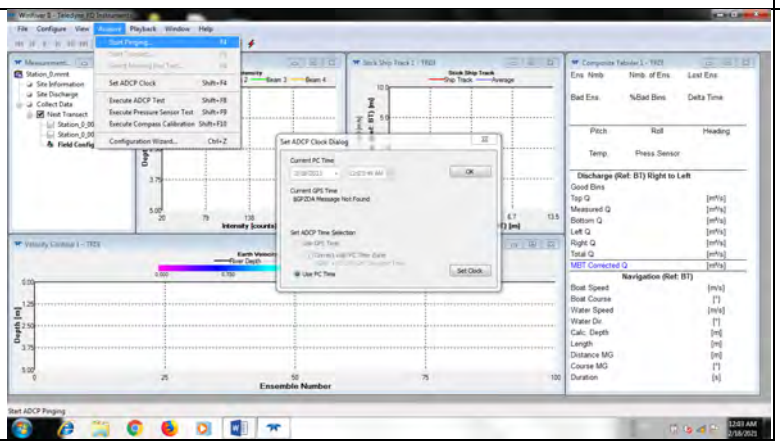
- WinRiver mainscreen:



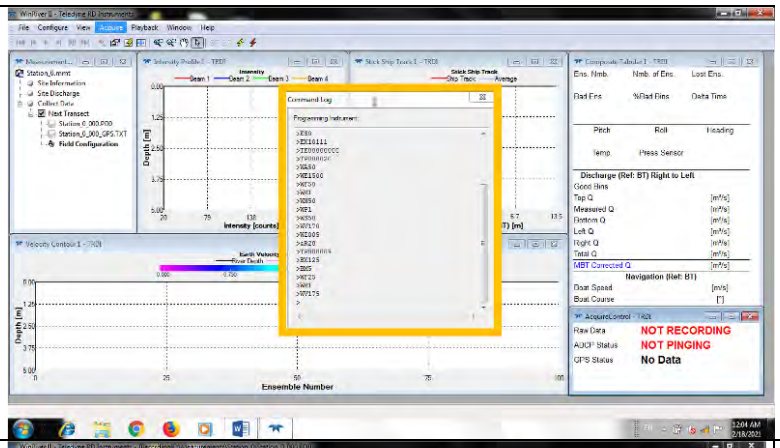
- Connect ADCP power source to ADCP cable



• Connect USB adapter with ADCP serial to the computer	
File→ New measurement → input your data: Station Name, Station Number and Date of Measurement (blue arrows):	
Click Next, you will go to Rating Information (no changes needed) and click Next again	
On the configuration Dialog screen wait until the ADCP and GPS sign turn to green (green arrow):… And click next,	

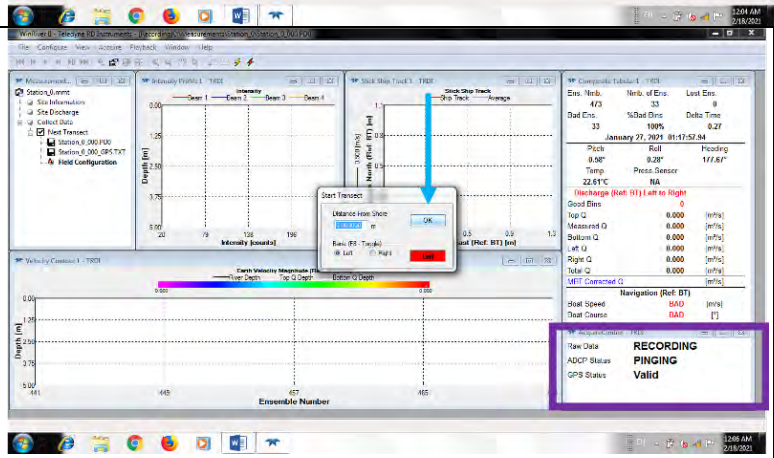
Find the folder where you want to save your measurements and… click next	
On the commands preview window you will need to set up your commands on the right hand side of the screen (where it says USER)* • Example: • WM11 • WS5 • WN25 • WF5 • BX15 • ES0 GO TO APENDIX	
• Then you will need to press next and finish… • In order to collect data go to acquire at the top menu and press start pinning or just press F4. • Make sure you have the same time as the computer and press ok	

you should see the following screen :

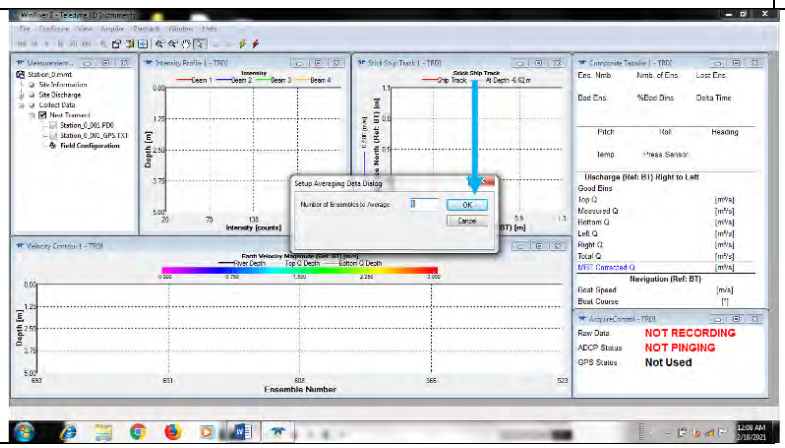


- Afterward the ADCP status should say Pinging in black instead of not pinging in red.
- Press ok (blue arrow)

- Finally, to start recording the new measurement you will need to press f5
  - And it should change again, from red to black and would say recording (purple square)



To finish recording you will need to press f5 again and it should show the following screen. And then press ok or enter on the keyboard

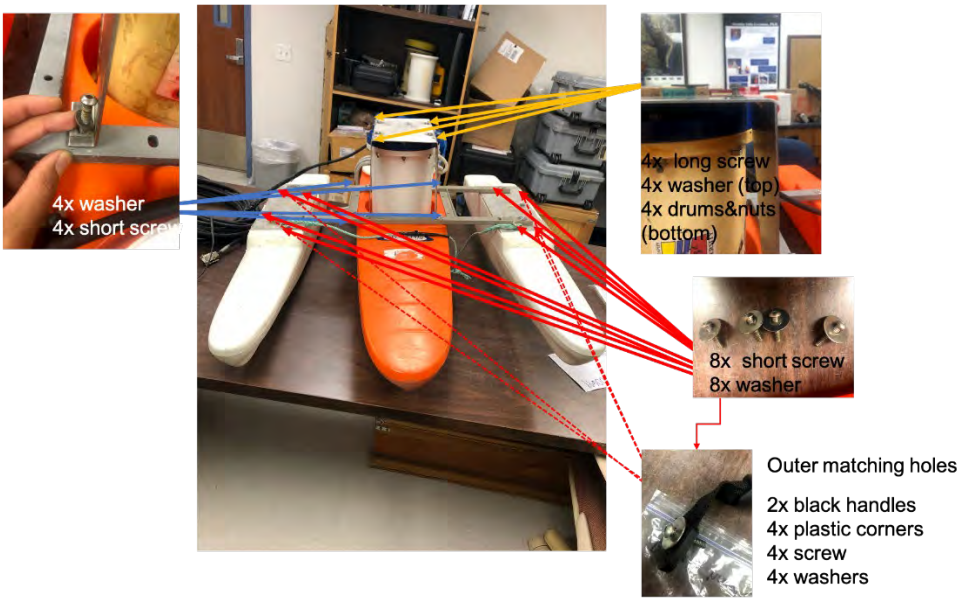


Appendix

- The parameters you would change according to the stream you want to sample are:
  - WM11 water mode (5 or 11 for slow shallow waters, 1 fast water all depths)
  - WS5 (5 cm cell size) you can try WS10 or WS15 for different depths
  - WF5 blanking distance WF=WS for WM 5 and 11 (remember to change this when you change WS)
  - WN25 (number of depth cell)
  - ES0 Salinity: 0 for freshwater or 35 for 35 ppt
  - BX15 (Maximum Bottom Search) how far the ADCP will reach the bottom (example 15for 1.5 m)

ADCP+ trimaran set up

Once the ACDP has been connected it needs a boat to cross the streams!



1. Set up the metal frame (upper fig)
2. Place ADCP (with power cable) from below in the hole in the center
3. Fix the metal box on top with the screws

Water & greenhouse gas sampling

Overview of water and gas sampling

Water and gas sampling is a simple procedure, but with many different small steps. For water sampling, the primary consideration is cleanliness. Even a minor amount of solid (dust, bits of skin, plant material) small enough that you can't see, will change the composition of the water. To maintain the best cleanliness, always work off of a clean tarp and wear gloves. Do not put the outlet end of the tubing or filters on the ground. Avoid touching the rims of the bottles even with the tubing or filter outlets. Close the bottles immediately after filling and/or adding preservatives, and keep them off of the ground – preferably placing them as soon as possible into clean plastic bags – usually gallon size baggies.

For gas sampling the primary consideration is to prevent contamination from atmosphere. There are many valves (2-way and 3-way) to open and close, and you must be careful to not accidently allow the sample or the filler gas (in gas bags) to come in contact with air. Before opening valves, think about which way they are oriented.

Samples will be collected after field parameters are measured using a YSI pro-plus multi-parameter meter. The meter should be calibrated every day it is used. two complete sets of bottles should be collected as field duplicates every tenth sample site,.

YSI calibration

The YSI must be calibrated the morning of the day it is used. Calibration will occur in the field laboratory and the results should recorded in the field book in the YSI case. Calibration steps are as follows:

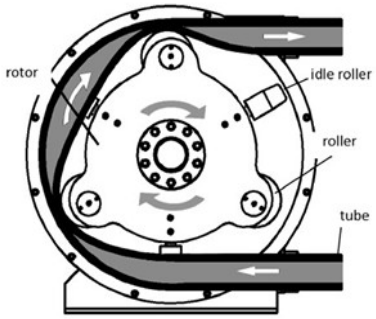
1) **Calibrate the DO probe.** Open the clear plastic protective cover and add ~ 1 inch of water. Loosely thread the cover back onto the probes. Using the YSI navigate to the calibration page for DO calibration. Allow to equilibrate for 10 minutes (this is a good time to go make coffee!). Once DO has stabilized, accept this value by pressing the "Cal" button. If DO is reading less than 90% or does not stabilize, consider replacing the membrane cap on the DO sonde. The YSI will accept this value as 100% DO.
2) **Calibrate pH using 4, 7, and 10 buffers.** Navigate to the pH calibration menu in the YSI. Rinse the electrode with DI water, shake off excess, rinse with pH 4 buffer remaining from the previous day's calibration, and shake off excess. Fill the calibration bottle with fresh clean pH 4 buffer until it covers the bulb at the end of the electrode – about 60 mL. Allow to stabilize, which may take a few minutes. A value of pH 4 may appear but if not, then change to 4, and press **accept**. Store the used calibration solution in a Nalgene bottle. Repeat this procedure with pH 7 and 10 buffers in that order. Once pH 10 has been accepted, press the "Cal" button for the YSI to accept all 3 pH points. The YSI should now be calibrated appropriately for pH.
3) **Calibrate the specific conductivity.** Rinse the electrode with DI water, shake off excess, then rinse the electrode with conductivity standard remaining from the previous calibration. Navigate to the calibration menu and select conductivity, then select specific conductivity. Submerge the sondes in a calibration bottle with enough water to cover the electrode up to the small dimple about half way up the electrode ~60 mL. Gently agitate the electrode to ensure there are no air bubbles trapped in the electrode. Allow the value to stabilize, ensure the correct calibration value is displayed at the top of the menu and if not change the value. Press accept to calibrate.

4) Check the calibration of the redox electrode using the Zobell's solution (ORP range ~ 218-238 mV). Navigate to the calibration menu and select ORP. Rinse with leftover solution then submerge the sondes in ~60 mL of new Zobell's solution. Always store this solution in the fridge but let it warm to room temperature prior to the check. If the ORP value is out of the range for the Zobell's solution, accept the value as 238 mV.

Once the electrodes are calibrated, store them in the solid plastic case that contains a small amount (a few milliliters) of tap water (NOT DI).

Geotech pump assembly

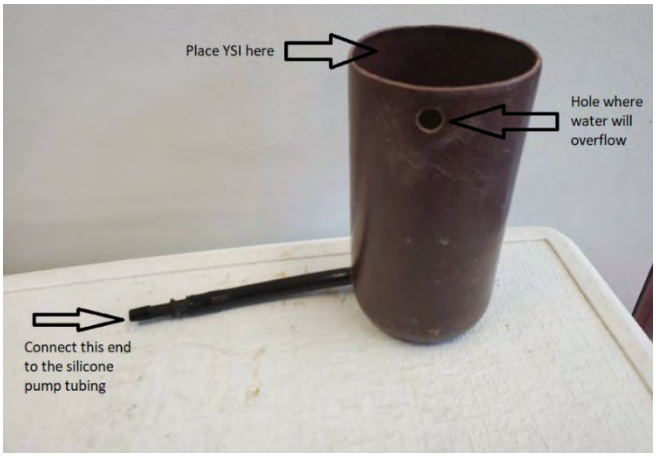
Water and gas samples will be collected via a peristaltic Geotech pump. To assemble, raise the upper portion of the roller housing with the lever, thread the clear silicone tubing into the pumping mechanism (only silicon tubing will work – the black PVC tubing will not), close the pump lever. Connect one end of the silicon tubing to black PVC tubing using double ended barb fittings. Deploy the end of the black tubing in the water body to be sampled, making sure that as much as possible of the sample tubing is in the water to ensure accurate temperature measurements (e.g., no warming if in the sun). The end of the tubing in the water will need to have a weight attached to hold it below the water surface, for example a small piece of metal or rock taped to the tubing. The weight should be attached a few inches back from the end of the tubing so that the end of the tubing points up from the bottom and the tubing should be deployed pointed downstream to keep sediment from clogging the end.



Connect the power cord to the battery and the pump. If the pump will not come on, the most likely problem is bad connections of the power cord, or the power cord has been broken by mishandling. **Do not ever kink the cord or bend it sharply especially near the fittings at the end.** The internal wires may break. The direction of flow does not matter – forward and backward are the same but be sure the flow switch is oriented to suck water from the stream. The knob on the pump controls the speed of pumping. Turn the knob all the way clockwise and the pump will be at its maximum speed – turn the knob counterclockwise to slow down the speed. Note there are two fittings for the pump head - 300 rpm and 600 rpm – which can be switched by unfastening a few bolts holding the head in place. The 600 rpm setting should be used to get maximum flow.

YSI Parameter

Prior to sample collection measure field parameters with the YSI, including temperature, dissolved oxygen (DO both as % and in mg/L), specific conductivity (set to units of µS/cm), pH, and oxidation-reduction potential (ORP – mV). Replace the solid clear plastic electrode covering with the guard cover - a black plastic cover with holes. **Do not cross thread or overtighten the guard or protective covers.** Once protected, the electrodes can be deployed directly in the flow path of water if possible or placed in an overflow cup. If deploying in



streams, place the sensor within the current – not in stagnant areas – and orientation it so the guard cover does not fill with sediment. Where flow is stagnant, such as a lake, measure the field parameters from an overflow cup. Connect the silicone tubing extending out of the Geotech pump to the connector at the bottom of the overflow cup. Place the YSI sondes in the overflow cup and pump water through the cup as fast as possible. Record the values for the field parameters every two minutes along with the time of measurement until the values stabilize along with the date and sample location (GPS coordinates).

Water chemistry sampling

If the YSI sonde is deployed in the overflow cup, sampling begins once YSI parameters are stabilized. If the sonde is deployed in a stream, sampling can begin after the tubing has been completely flushed – typically after pumping for 2 to 3 minutes depending on the length of the tubing. Detach the silicone tubing from the overflow cup and attach a 0.45 um high capacity filter to the end of the silicone tubing. Ensure the tubing is connected to the appropriate end of the filter, so that the flow of water follows the arrow on the filter indicating the proper direction of flow.

Begin pumping sample water at a moderate flow rate, while keeping the filter vertical to ensure all air is removed from the filter. Let the sample water flow for 1-2 minutes before commencing sample collection, depending on flow rate. This should be equivalent to ~3 volumes of the filter and is especially important to do so when re-using filters between different sample locations.

Prepare enough sample bags that contain all the necessary bottles (Table 1) for the planned number of sample collections plus a couple spares in case anything is missing, or you decide to collect additional samples.

Gloves should be worn during sampling collection and all sample bottles and equipment should be placed on a clean tarp to prevent contamination from dirt, plants, and bugs. All sample vials and caps should also be rinsed three times with sample water before filling the vials. **Do not mix up the acid washed and non-acid washed bottles when sampling**. For all samples other than DIC and $\delta^{13}C_{DIC}$, fill bottles by having an arc of the water flow into the mouth of each bottle without touching the rim of the bottle with the tubing or filter outlet. Leave a small (~5 to 10 ml) headspace at the top of the bottles. Immediately preserve samples according to the table and once preservatives are added, briefly shake these vials to ensure mixing. Nutrient sample vials should be frozen as soon as possible after collection.

Table 1. Water Sampling bottle types

- **Unfiltered**

# bottles	For	Bottle material	Size (ml)	preservative
2	TOC	Amber glass	20	HNO_3

- **0.45 um filter**

# bottles	For	Bottle material	Size (ml)	preservative
2	Rads	Plastic	250	HNO_3
1	Cations and colloid	Acid washed HDPE	20	1 drop HNO_3
1	DOC	Amber glass	20	1 drop HCl
1	Nuts	HDPE, **not** acid washed	30	
2	Alk	HDPE **not** acid washed	60	
1	anion	Opaque cap, **not** acid washed	20	
1	CDOM	Amber glass	20	
2	DIC and $\delta^{13}C_{DIC}$	Qorpak clear glass	20	1 drop (BAC)

- **0.45 + 0.2 um filters**

# bottles	For	Size (ml)	Bottle type
2	Colloids (to squeeze)	60	syringes
2	Stable isotopes	2	Glass black plastic top

The DIC and $\delta^{13}C_{DIC}$ samples are collected to limit atmospheric exchange of CO_2. To sample, turn the pumping rate down to minimal flow and place the tip of the filter on the edge of the vial and slowly fill until a meniscus forms at the top of the vial. Allow water to overflow the bottle for10 to 15 seconds prior to preserving the sample. After removing the filter, add 200 µl of 1% Benzalkonium Chloride (BAC) solution by submerging a pipette tip in the vial and dispensing the BAC at least halfway down in the sample vial. Fill up the cap with sample water, and rapidly cap the sample vial. Screw the lid on tightly – there should be a point where the seal reaches the lip of the vial and then an additional turn will compress the seal. Once twisted tight, invert the sample vials to ensure no bubble are present in either vial. If bubbles are present, dump out the sample and repeat the process.

For colloid samples, pull plunger completely out of a 60 ml syringe, fit the wrapped 0.02 µm Anotop filters on the outlet end, and fill with filtered water. The 0.02 µm filter will act as a

stopper. Keep the filter tip covered in saran wrap to avoid contamination. In the lab, place the syringes in the colloid squeezers and place an acid washed sample bottle in the slot under the syringe. Turn the threaded rod at the top of the colloid squeezer water drips from 0.02 µm filter. Rinse bottle with the first 1 mL by capping and shaking the bottle then discarding the water. Replace the bottle under the syringe and fill at a rate of < 1 drop per second by occasionally tightening the threaded rod at the top. It may take more than half an hour to fill the bottle. Do not overtighten the squeezer which will twist the syringe, make squeezing inefficient, and possibly burst the filter. A burst filter will be observed as a flow stream rather than drops and that sample must be discarded.

Gas headspace extraction

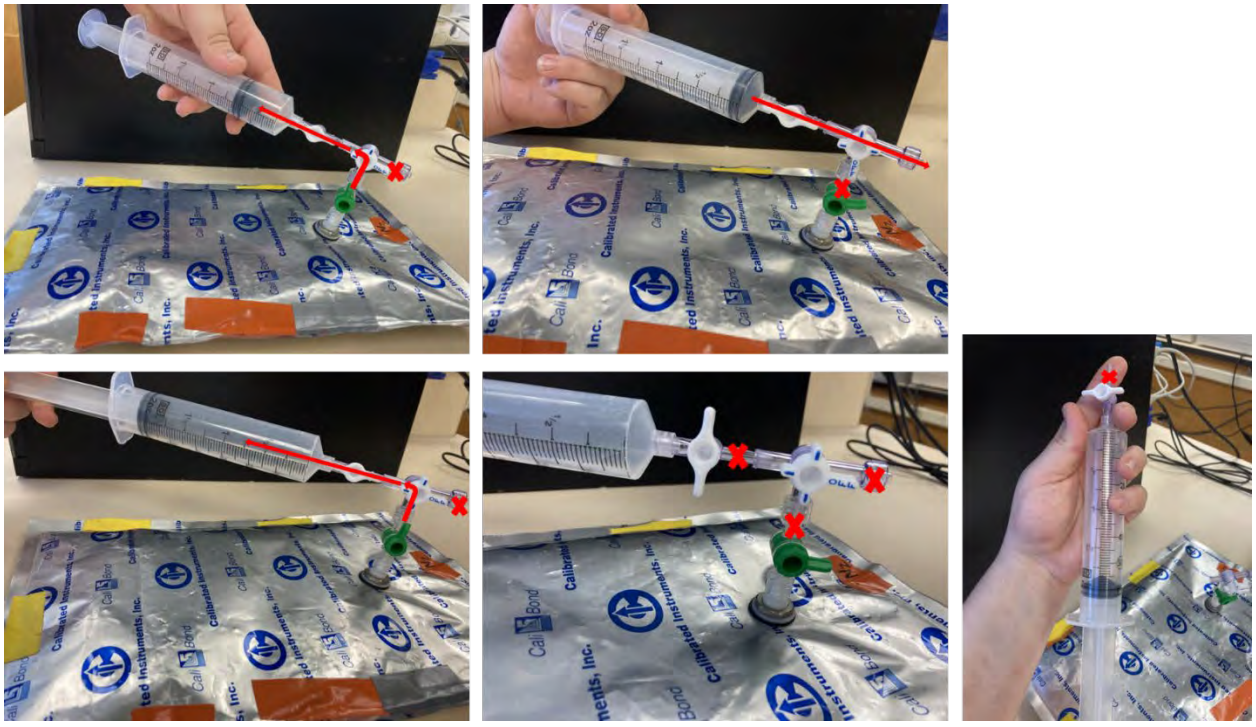
Disconnect all filters from the end of the silicone tubing for headspace extraction samples and insert to the bottom of the ~600 ml plastic bottles. Pump sample water as fast as possible to completely fill the bottle and continue pumping from the bottom for about 30 seconds to displace the water in the upper portion of the bottle that was in contact with the atmosphere. While overflowing loosely place the black rubber stopper over the top with the long plastic tubing in the bottle. While continuing to pump rapidly and overflowing the bottle, slowly remove the silicone tubing so the meniscus at the top of the bottle is preserved. Both stopcock valves should be open (parallel with the length of the tubes). As soon as the silicon tubing is removed, push the stopper into the neck of the bottle with a twisting motion and close the valves as water squirts from the tubing. No bubbles should be in the bottle – if there are bubbles dump the sample and repeat.

Attach a 60 ml syringe to the valve connected to the long tubing. All connections describe below are made with twist Luer lock fittings. Extract 60 mL of water by simultaneously pulling the syringe plunger out and squeezing the bottles, which can be done by placing the bottle between your knees. In turbid samples it may be difficult to see the tubing, so prior to filling, be sure to identify which valves are attached to the long and short tubes. The bottle will be under negative pressure (e.g., slightly collapsed with no gas in it) if the stopper is sealed tightly.

Fill a syringe with N_2, by first purging the connections by extracting approximately 10 mL of N_2 from the bag into a syringe through a three-way stop cock attached to a two way master valve (green in figure) on the gas bag. Once extracted, close the master valve, orient the three-way valve to displace this purged gas from the syringe to the surroundings. Yyou will not have to remove the syringe but make sure the tick marks on three way valve are oriented as shown in the picture. Re-orient the three way valve to your syringe, open the master valve, and extract 60 mL gas. Be careful to not allow the three-way valve to become loose or twist off of the fittings. Make sure the valve on the tubing in the sample bottle is closed and replace the water-

filled syringe with a syringe filled with N_2 gas taken from a gas pillow. Open valves so that you can inject the gas into the sample bottle. Make sure all valves are closed and remove the syringe. Vigorously shake the samples for 3 minutes.

Evacuate the 75 mL serum vial that will store the sample by connecting a hypodermic needle to a 60 mL syringe, closing the valve, and inserting the needle through the septa of the serum bottles. Open the valve between the bottle and syringe (which automatically closes the value to the atmosphere), pulling 60 ml from the bottle, switching to open the syringe to the atmosphere, and pushing out the gas by depressing the plunger. Repeat this process at least five times – as the vacuum increases, it will not be possible to pull a full 60 ml. Close the valves and remove the syringe from the three-way valve leaving the needle inserted in the serum bottle with the three way valve closed to the atmosphere.



Connect a Luer valve to a 60 mL syringe and attach the syringe Luer valve to the valve attached to the SHORT tubing in stopper on the headspace bottle. Open the valves on the stopper and syringe, making sure neither of them is open to the atmosphere. Extract the 60 mL headspace in the sample bottle while simultaneously squeezing the bottle between your knees to ensure a complete transfer of the gas. A small amount of water may enter the syringe, but that is ok. Immediately, close the valve on the syringe, disconnect from the sample bottle and attach to the valve to the needle on the serum bottle. Open all values to transfer gas from the syringe to the serum bottle. If the serum bottle has a good vacuum, it will draw most of the sample into the serum bottle although friction of the syringe may require pushing the plunger to the end, especially for the last few tens of ml of gas.

Suspended Sediment Sampling

Overview

The sediment samplers will be placed in Greenland streams on the first deployment of the Summer 2021 SILA field season and removed following the second deployment in September 2021. The goal of the sampler is to trap the suspended sediment load being actively transported in streams to study minerology and magnetic, geochemical, and isotopic properties. The following sections go over how to set-up the sediment samplers, how to maintain them through the field season, and the post-collection process to isolate the suspended sediment load from the water for sample packing and transport.

_____. The sampler is composed of three main pieces: 1) the front 'nose- cone' end that streamlines the flow around the sampler body, 2) the main sampler body (1 m length x 10 cm diameter poly tube), and 3) a twist-off end cap. The nose-cone and sampler body are permanently fixed in place using PVC glue. The twist-off end cap unscrews and can be removed entirely, allowing access to the sampler body. The sampler is attached using zip-ties to two fixed points that are hammered into the stream bed.

_____ When the nose-cone of the sampler is pointed upstream, water and its associated suspended load enters the sampler at the ambient velocity of the stream through the small millimeter-scale inlet (Figure 1). As the stream water encounters the sampler body its velocity drops due to the greater cross-sectional area of the sampler body. This velocity reduction causes the sediment load to fall out of suspension inside the sampler body; water then exits out through a small diameter tube drilled through the twist-off end cap.

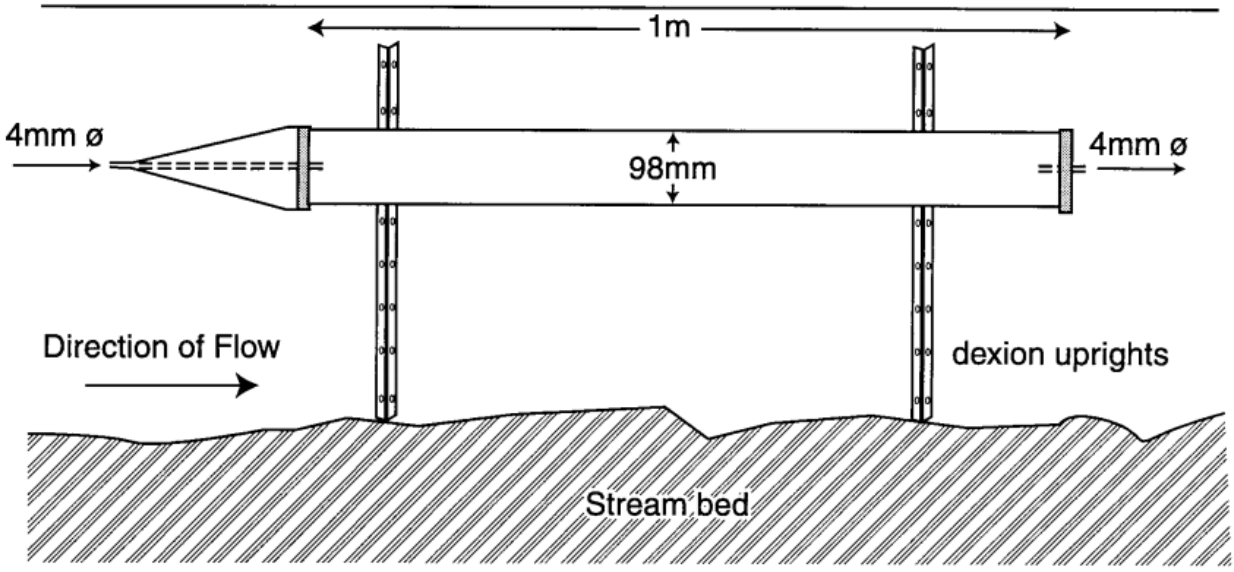


Figure 1: Cross-section of the suspended sediment sampler. Reproduced from Phillips et al. (2000) **14**, 2589-2602.

Sampler Installation

Samplers will be placed in the Watson River at the sandflugtdalen braided river system and at a proglacial location close to the Greenland Ice Sheet. Samplers will also be installed in the Orkendalen River, in the Lake Helen watershed, and in location(s) near to the town of Sisimiut. Materials needed to deploy a sediment sampler include; one sampler body with attached nose-cone, one twist-off end cap, two large zip-ties, two metal stakes, and the mini sledgehammer.

1. Pick a spot at the desired location. For optimal positioning, choose a place with a relatively flat stream bed and reach with steady flow that is deep enough to be able to fully submerge the sampler body. Positioning may not always be able to fulfil these criteria (boulders, low flow, etc), but do your best.
2. Using the mini sledgehammer, drive the two metal stakes into the streambed parallel to the direction of flow, about 60-80 cm apart.
3. Unscrew the twist-off end cap and rinse the sampler body in ambient stream water.
4. Submerge the sampler body so it entirely fills with river water.
5. While the sampler body is submerged, reattach the twist-off end cap to the end of the sampler body and tighten all the way.
6. Orient the sampler nose-cone end upstream and connect the sampler body to the stakes using one zip-tie for each stake. Use the lip as a natural catch-point for the zip-tie and tighten all the way so the front end cannot slip through the zip-tie. Note: It is optimal to try to place the sampler body at 60% of the mean water depth, but this is not always possible.
7. Zip-tie the back of the sampler body to secure its position parallel to the streamflow.
8. Check for stability by gently wiggling the stakes. If there is significant movement, either hammer in the stakes further or tighten the zip-ties.

Sample Collection

Over time, the sampler bodies will fill up with water/sediment and will need to be changed out. We imagine that this will occur every 2-3 days at all locations excluding Lake Helen and the sampler(s) in Sisimiut, where collections may occur on a monthly regimen; however, this maintenance interval will be determined and refined during deployment 1. In order to collect the material in the sample body, you will need a 5G (5 gallon) plastic container, the large funnel, and the wire cutters.

1. Cut the zip-ties one at a time starting at the back of the sampler to free the sampler from the stakes.
2. Position your index finger over the nose-cone inlet and lift the sampler from the river. Keep a slight incline on the sampler body with the nose-cone end (plugged by your finger) at the lowest point to prevent water escaping through the exit hole in the twist-off end cap. Using your third hand (or a friend), remove the twist-off end cap, and very slowly pour the sampler body contents through the funnel into the 5G plastic container. The key here is to do this pouring step very slowly as water and sediment can (and likely will) splash out of the funnel, or worse, the rush of water from the sampler body means you can miss the funnel entirely.

3. Carefully introduce a small amount of ambient river water into the open sampler end to mobilize any sediment stuck on the sidewalls of the body and carefully pour that water into the plastic container as well. Repeat if necessary.
4. Rinse out the sampler body using water from the river and discard.
5. Reinstall the sampler following steps 4-8 in the **Sampler Installation** instructions.
6. Label the 5G plastic container with agreed upon sample names using tape and a sharpie.

Sampler Maintenance

We tried to build these samplers as robust as possible, but due to the harsh proglacial environment in Greenland, some of the components may get damaged in the field and require maintenance. We have shipped extra sampler bodies, nose-cones, and twist-off end caps for these instances. The following steps detail how to build a sediment sampler.

1. What you will need: one main sampler body, one nose-cone-shaped end cap, one twist-off end cap, and PVC cement.
2. On one end of the main sampler body, place a few dabs of PVC cement a few centimeters from the edge on the outer surface of the sampler body.
3. Slide the nose-cone end cap over the sampler body and PVC cement. If possible, twist the nose-cone end cap through a few degrees while sliding it onto the main sampler body to distribute the PVC cement. Let the sampler cure overnight.

Sample Treatment (@KISS)

1. Let the 5G plastic container sit undisturbed on a bench or table to allow the contents to settle. At a minimum this should be 24-48 hrs. Do not store the 5G containers on the floor to settle as you will have to move them up to a bench for siphoning. Moving them can cause resuspension of settled material.
2. From the bench/table location, carefully siphon off much of the water into a holding container taking care not to siphon up any of the settled sediment. Check that this water is relatively clear and discard.
3. Slowly pour the contents of the 5G plastic container into the 2 liter (2L) plastic beakers using the large funnel. Make sure to rinse the inside of the 5G plastic container with the lab squirt bottles and add this rinse to the 2L plastic beakers. Label the beakers with the agreed upon sample names using masking tape and a sharpie. Allow the sediment in these beakers to settle out a second time for as long as possible.
4. Repeat the siphoning process outlined in step 2 for the 2L plastic beakers, again taking care not to siphon up any of the settled sediment.
5. Scrape/wash/be inventive in coercing the sediment into whirl-paks using as little extra water as possible. We don’t want to have to ship, or have to carry, excess water.
6. Label the bags with agreed upon sample names.
7. Rebag the whirl-pak bagged sample in a second whirl-pak, label this bag for redundancy.

Microbiology Procedure Introduction

This document describes procedures for the following field activities:

1. Sampling water to measure microbial abundance and biomass
2. Filter concentration of microbial biomass for nucleic acid extraction
3. Measuring stream plankton O_2 consumption and DIC production
4. Sampling and collection of stream periphyton (coming soon)

Note: Contamination of samples with microbes of human or extraneous origin will affect our measurements and must be avoided. To prevent this possibility, please wear a fresh pair of gloves when performing microbiological collection procedures. Most materials are provided sterile, but when indicated, some equipment will require you to disinfect it with 3% hydrogen peroxide or 70% ethanol before use.

Sampling water to measure microbial abundance and biomass

Microbial cell enumeration using epifluorescence microscopy

50 mL conical tubes
Gilson P1000 pipette and tips
40 mL syringes
0.22 um syringe filters
sharpie
gloves (note sizes)
sample cooler and chilled blue ice

Sodium borate
Formalin (37% formaldehyde)

1. Put on a fresh pair of gloves.
2. Buffer the formalin with sodium borate.
    a. Add a small amount of sodium borate (a couple of grams; about half a spatula) to 20mL formalin in a 50 mL conical tube. Mix it by shaking.
    b. Remove plunger from a 40 mL syringe. Attach the syringe to a 0.22 um syringe filter. Decant the mixture into the syringe so that the undissolved sodium borate remains in the tube.
    c. Insert the plunger and push the solution through filter, collecting it in a new conical tube.
    Tightly close the conical tube for transport to the field.

1. Put on a fresh pair of gloves.
2. Collect 40 mL of water in a 50 mL conical tube (use the graduations on the tubes for approximate volume).
3. Add 2 mL borate-buffered formalin to each sample.
4. Label each sample with the following information:
    i) Site designation; ii) CC (cell count); iii) date; iv) KC (keep chilled); v) DNF (do not freeze)
5. Transport the samples in the chilled cooler.
6. Upon return from the field, store the samples at 4°C.

Estimating microbial biomass based on ATP quantification

sterile 1 L Nalgene bottle
60 mL syringes
0.22um Millex GS syringe filters
Small whirlpak bags
500mL 3% (v/v) Hydrogen Peroxide
labeling tape
sharpie
powder-free gloves (important!)
sample cooler and chilled blue ice

1. Put on a fresh pair of gloves.
2. Add ~50ml of 3% hydrogen peroxide to the 1L bottle, place the lid on, shake vigorously, and then open and dispose of the material (H_2O_2 degrades to H_2O and O_2 and can be disposed of in the field). Rinse the cleaned bottle with stream water 3 times before collecting a sample.
3. Collect a stream water sample in the 1L bottle.
4. Draw up 50mL of the water with the syringe. Invert the syringe so that any air bubbles can be excluded by advancing the plunger. After bubble exclusion, use the gradations on the syringe to record the actual volume.
5. Carefully open the packaging for a 0.22 um Millex GS syringe filters by pulling one corner so that the filter can be removed but the sealing flap remains attached to packaging (the manufacturer's packaging is ideal for sample transport and we'll be returning the filter to the packaging in a subsequent step, so don't discard it!).
6. Attach the syringe filter and push the water through the filter. The filtrate can be discarded on site.
7. Detach the filter from the syringe, fill the syringe with a volume of air, reattach it to the filter, and push the air through the filter to purge the filter of excess water.
   Note: this last plunge will take a little more effort
8. Remove the purged filter from the syringe and place it into the packaging provided by the manufacture. Use a length of lab tape to reseal the filter in the packaging and provide a fresh surface for labeling.
9. Label the sample with the following information:
   i) Site designation; ii) ATP; iii) date; iv) volume filtered; v) KF (keep frozen)
10. Insert the labeled sample into a small whirlpak bag, fold over, and twist the ties to seal.
11. Transport the samples in the chilled cooler.
12. Upon return from the field, store the samples at -20°C.

Chlorophyll a

Materials

Gloves
1 Liter Amber Bottles- 1 per location
labeling tape
sharpie
sample cooler and chilled blue ice

Collection procedure

1. Put on a fresh pair of gloves.
2. Rinse a clean 1L amber bottle with stream water 3 times before collecting a sample.
3. Collect a stream water sample in the 1L bottle. Fill to the top and screw on lid.
4. Label each sample with the following information:
   i) Site designation; ii) CHL (chlorophyll); iii) date; iv) KC (keep chilled); v) DNF (do not freeze)
5. Transport the samples in the chilled cooler.
6. Upon return from the field, store the samples at 4°C.

Filtering water to concentrate microbial biomass for nucleic acid extraction

Materials

gloves
Geotech peristaltic pump
Two **Charged** Batteries for the Geotech Pump
Two short (~40 cm) lengths of Master Flex tubing
Two 1 m and one 10 m length of Fisherbrand 5/16" tubing
Two tubing connectors
Weight for sample intake end of tubing
Supor®-200 142 mm membrane filters (**one per sample**)
142 mm filtration unit (with spare parts bag and Teflon tape)
1 L plastic beaker
150 mm petri dish (**one per sample**)
Sharpies
Labeling tape
2 pairs of forceps (flat)
500 mL 3% (v/v) hydrogen peroxide
25 mL 70% (v/v) ethanol
lighter
spray bottle (for hydrogen peroxide)

parafilm
tarp
sample cooler and chilled blue ice

Collection procedure:

1. Put on a fresh pair of gloves.
2. Set up filtration unit (see schematic and photo in Figure 3 for guidance):
    a. Screw the three legs into the bottom plate.
    b. Loosen the six Swing-A-Way bolts and remove the top plate
    c. Place an O ring on bottom plate and then the sample disbursement disc, which should snap into place. (Note: these parts may already be in place)
    d. Use the spray bottle to generously spray saturate these components with two or three squirts of 3% H_2O_2.
    e. While holding them with gloved hands, use the squirt bottle to generously spray both filter support screens, then place them on top of the disbursement disc.
    f. Ensure that the sample disbursement disc and it's O ring are snapped into top plate (Note: it may already be snapped in). Use the spray bottle to generously spray saturate the components on the top plate with two or three squirts of 3% H_2O_2.
    g. Mate the top and bottom plate and tighten the fasteners progressively using a star pattern. Repeat this pattern until each is snug. DO NOT OVERTIGHTEN AS THE THREADED NYLON COMPONENTS CAN BE DAMAGED BY THIS.
    h. Make sure that the threaded t valve and hose barbs are screwed in securely.

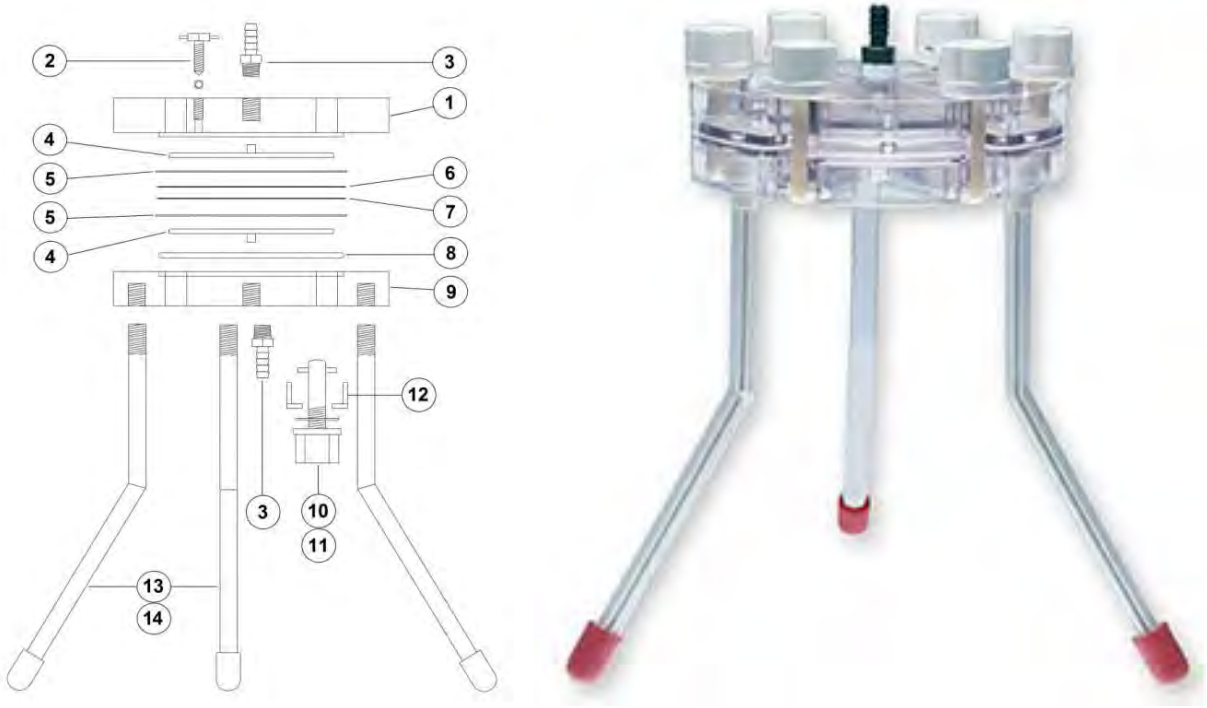
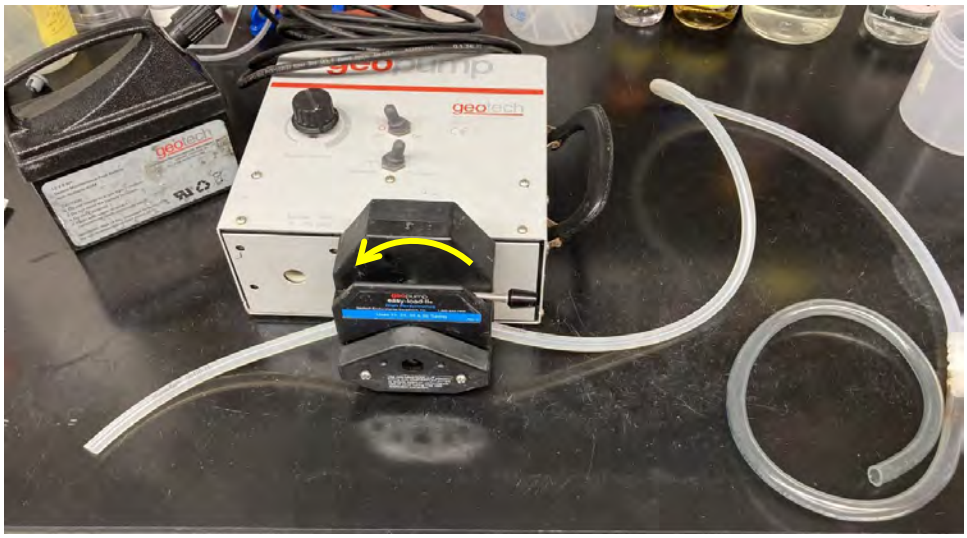


Figure 3. 142 mm filtration unit

1	Top plate
2	Threaded T valve
3	3/8 x 3/8 MPT Hose Barb
4	Sample Disbursement Disc
5	Filter support screen
6	N/A for this protocol
7	Filter
8	O-ring
9	Bottom plate
10	Nylon Swing-A-Way bolt
14	Nylon Tripod Leg
Note: top and bottom plate are the same.	

Table 1. Filter parts



3. Lift the lever on the Geotech pump head to open it (see arrow), insert one of the ~40 cm lengths of Master Flex tubing, and reclose the pump head. Use a tubing connector to link the intake with the ~10 m section of Fisherbrand tubing that will be inserted into the stream.
4. Connect one of the 1 m lengths of Fisherbrand tubing to the other end of the masterflex tubing (outflow) to the top plate of the 142 mm filter holder. Place the other length of 1 m Fisherbrand on the barbed fitting at the outflow on the bottom plate.
5. Pour ~100 mL of 3% hydrogen peroxide into the 1 L beaker.
6. Turn on the pump and insert the end of the 10 m of Fisherbrand tubing into the beaker and allow all the peroxide to be pumped through the entire length of tubing and the filtration unit. If the tubing is blowing bubbles into the periodixe solution, then the pump if flowing in the wrong direction and needs to be switched.

7. When all the hydrogen peroxide solution has been purged, insert the end of the tubing into the stream, set the pump on its maximum speed, and allow the pump to purge stream water through the system for 5 minutes.
   NOTE: Steps 1-7 are assembly and cleaning preparation for the actual filtration steps that follow.
8. Turn the pump off, unscrew the six Swing-A-Way bolts, carefully remove the top plate, and place in a location on the tarp so the cleaned internal surface are facing upwards and not in contact with any unclean surfaces.
9. Use a clean pair of flat forceps (dip into 70% ethanol and burn excess with lighter) to remove the uppermost filter support screen (a good place to put this temporarily is on the cleaned surface of the top plate you just removed and inverted on the tarp).
10. Using clean flat forceps (important!), remove a single Supor-200 142 mm 0.2 um membrane filter and place on top of the filter support screen. Then place the second filter support screen (currently resting on top plate) on top of the filter.
11. Reassemble and tighten the filtration unit as described in 2g.
12. Place the end of tubing with the weight into the stream and place the end of the 1 m of tubing attached to the filtration unit outflow in an empty 1 L beaker to measure the approximate volume sampled.
13. Turn on the pump and set the pump speed on the highest setting. Water from the stream should enter the tubing, filtration unit, and begin flowing from the outflow into the beaker.
14. To vent air from the system, partially loosen the threaded T valve, tilt the filtration unit so the valve is elevated, allowing any trapped bubbles to rise to the value and escape. During this process, don't worry about the small amount of liquid that will also escape with the air. After purging the air, retighten the T value until it is sealed. During the filtration process, air will continually enter the system, so occasionally check for air bubbles and periodic purge them, which reduces filtration time.
15. When the 1 L beaker is filled with filtrate, record that a liter was been filtered, dump it out, and continue collecting filtrate in the emptied beaker. Counting the number of times the beaker fills is the way we will keep track of the approximate volume filtered per sample.
16. The objective is to filter at least 10 L of water through each filter, but if water is still flowing well through the filter after filtering 10 L, please continue filtering until the flow rate is reduced to a trickle, indicating that the filter is loaded and clogging with particulate material.
    Note: Sediment-laden waters sourced from the ice sheet will likely clog the filter after passing ~10 L, but up to 20 L should be filterable in waters with lower sediment content. As the filter accumulates material, some amount of water may start leaking from the filtration unit or its fittings. The Swing-A-Way bolts can be snugged, but please do not overtighten them. And if a little water is leaking out, it is not a big deal.
17. Turn off the pump, loosen the six Swing-A-Way bolts, and **carefully** lift up the top plate of filtration unit to access the filter. When doing this, make sure the filter and filter support screen have stayed on the bottom plate and did not stuck to the top plate, which

could cause them to fall when moving and drop onto the ground as the top plate is removed.

18. Using two pair (one in each hand) of clean flat forceps (dip into 70% ethanol and burn excess with lighter), transfer the filter into a clean 150 mm petri dish and seal with parafilm (can also be sealed with lab tape for transport).
19. Label the top of the petri plate with the following information:
    i) Site designation; ii) DNA; iii) date; iv) volume filtered; v) KF (keep frozen)
20. Transport the sample in the chilled cooler.
21. Upon return from the field, store cold and alert a member of the microbiology team (the samples need to be transferred into a storage buffer ASAP and before they are stored frozen at -20°C.)

Procedure upon return to the field lab:

22. In front of the laminar flow hood in the lab, remove the parafilm and lid from the petri dish containing the sample.
23. Use a cleaned scalpel to cut filter in into quarters and clean forceps to fold up the filter pieces and place them in a 10 mL cryovial.
24. Add 5 mL of DNA storage buffer or RNAlater (only for specific samples).
25. Label the top of the petri plate with the following information:
    i) Site designation; ii) DNA or RNA; iii) A or B or C or D (each letter representing a subsample); iv) date; v) volume filtered; vi) KF (keep frozen)
26. Transfer the samples to a -20°C freezer for storage (Note: may want to store samples cold in RNAlater for ~1h before freezing)
27. Ship samples to Gainesville frozen and store at -20 or -80°C.

Note: when overnight camping prevents daily return to the lab, may need to bring some of the storage buffers into the field.

Measuring O_2 consumption and DIC production

Materials

gloves
9 – 40 mL French square bottles and caps (6 with wire loops and 3 without, per site)
9 – 20 mL French square bottles and caps (6 with wire loops and 3 without, per site)
P1000 pipette and tips
alkaline-iodide-azide solution (250 mL)
manganese chloride solution (250 mL)
1% Benzalkonium Chloride (BAC; 50 mL)
P1000 pipette
20 ml or 40 ml French square vials

~2 m of 550 paracord (per site)
labeling tape
aluminum foil
1 roll of thin craft wire
12 – 1” small nonclimbing carabiners (per site)
2 – 3.5” nonclimbing carabiners (per site)
sample cooler and chilled blue ice
tarp

Dissolved Oxygen by the Winkler Method

Prior to field:

1. Put on a fresh pair of gloves.
2. Make alkaline-iodide-azide solution by adding 2.5 g sodium azide (NaN_3), 25 g potassium iodide (KI), and 80 g sodium hydroxide (NaOH) to 250mL water. Store in the dark.

Note: sodium azide is toxic. It should only be handled with gloves and any overflow can be collected by placement of the serum bottle on a paper towel to absorb the excess.

3. Make manganese chloride solution by adding 100 g of $MnCl_2$ to 250 mL deionized water.

Collection procedure:

4. Put on a fresh pair of gloves.
5. Fill each of the 9 – 20 mL bottles with sample water and insert the closure on the 6 bottles that have wire loops attached to their necks while avoiding inclusion of any headspace gases (these 6 samples will be incubated for 24 h in situ; t=24). After capping, invert the bottles to make sure no air bubbles are present. If bubbles exist, then open, empty, refill, and cap the bottle to exclude air.
6. Place a piece of labeling tape on each closure and label three with a “L” for light incubation and three with a “D” for dark incubation.
7. To the 3 uncapped 20 mL bottles without wire loops attached to their necks, fix each sample by addition of 900 ul of the manganese chloride solution and 900 ul of the alkali-iodide-azide solution. Insert the pipet tip about halfway into the solution when dispensing so that the additions displace liquid from the surface (these 3 samples will be t=0 concentration values).

8. Add cap and invert the bottles to make sure no air bubbles are present.
9. Label each of these bottles using a piece of tape and the following information: i) Site designation; ii) DO (dissolved oxygen); iii) date and time; iv) rep A or B or C (each letter representing a replicate); v) t=0 (initial [DO])); vi) DNF (do not freeze)
10. Store the t=0 samples at room temperature until titration.
11. Using the small carabiners, attach the 6 sealed 20 mL bottles with wire loops (t=24) to the paracord rigging provided. Use a piece of aluminum foil to completely cover the 3 bottles for dark incubation (D).
12. Place the bottle string into the stream and secure the free end of the paracord to any support on the stream bank (a 3.5" carabiner and extra paracord are provided to use for this if needed). Record the time that the samples were placed in the stream.
13. Allow the bottles to incubate in situ incubation for 24 h.
14. After incubation for 24 h, remove the bottles from the stream, and carefully remove the closures.
15. To each of the 6 bottles, fix each sample by addition of 900 ul of the manganese chloride solution and 900 ul of the alkali-iodide-azide solution. Insert the pipet tip about halfway into the solution when dispensing so that the additions displace liquid from the surface (these 6 samples will be t=24 concentration values for the dark and light).
16. Add cap and invert the bottles to make sure no air bubbles are present.
17. Label each of these bottles using a piece of tape and the following information: i) Site designation; ii) DO (dissolved oxygen); iii) date and time; iv) Dark/Light A or B or C (each letter representing a replicate in the dark or light); v) t=24 (final [DO])); vi) DNF (do not freeze)
18. Transport the sample at ambient temperature.
19. Upon return from the field, store all samples at room temperature until titration.

Measurement of dissolved inorganic carbon

Prior to field:

1. Put on a fresh pair of gloves.
2. Prepare a 50 mL solution of 1% (w/v) benzalkonium chloride (BAC)

Collection procedure:

Put on a fresh pair of gloves.

3. Fill each of the 9 – 40 mL bottles with sample water and insert the closure on the 6 bottles that have wire loops attached to their necks while avoiding inclusion of any headspace gases (these 6 samples will be incubated for 24 h in situ; t=24). After capping, invert the bottles to make sure no air bubbles are present. If bubbles exist, then open, empty, refill, and cap the bottle to exclude air.
4. Place a piece of labeling tape on each closure and label three with a "L" for light incubation and three with a "D" for dark incubation.

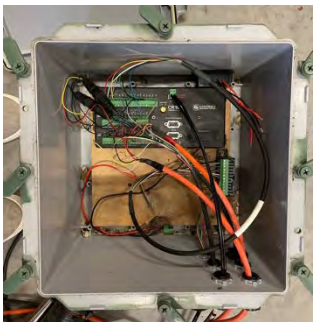
5. To the 3 uncapped 40 mL bottles without wire loops attached to their necks, fix each sample by addition of 400 uL of 1% BAC to each bottle. Insert the pipet tip about halfway into the solution when dispensing so that the additions displace liquid from the surface (these 3 samples will be t=0 concentration values).
6. Add cap and invert the bottles to make sure no air bubbles are present.
7. Label each of these bottles using a piece of tape and the following information: i) Site designation; ii) DIC (dissolved inorganic carbon); iii) date and time; iv) rep A or B or C (each letter representing a replicate); v) t=0 (initial [DIC]); vi) DNF (do not freeze); vii) KC (keep chilled)
8. Store the t=0 samples in the chilled cooler.
9. Using the small carabiners, attach the 6 sealed 40 mL bottles with wire loops (t=24) to the paracord rigging provided. Use a piece of aluminum foil to completely cover the 3 bottles for dark incubation (D).
10. Place the bottle string into the stream and secure the free end of the paracord to any support on the stream bank (a 3.5” carabiner and extra paracord are provided to use for this if needed). Record the time that the samples were placed in the stream.
11. Allow the bottles to incubate in situ incubation for 24 h.
12. After incubation for 24 h, remove the bottles from the stream, and carefully remove the closures.
13. To each of the 6 bottles, fix each sample by addition of 400 uL of 1% BAC to each bottle. Insert the pipet tip about halfway into the solution when dispensing so that the additions displace liquid from the surface (these 6 samples will be t=24 concentration values for the dark and light).
14. Add cap and invert the bottles to make sure no air bubbles are present.
15. Label each of these bottles using a piece of tape and the following information: i) Site designation; ii) DIC (dissolved inorganic carbon); iii) date and time; iv) Dark/Light A or B or C (each letter representing a replicate in the dark or light); v) t=24 (final [DIC])); vi) DNF (do not freeze); vii) KC (keep chilled)
16. Transport t=24 samples in the chilled cooler.
17. Upon return from the field, store all samples at 4^{o}C.

Stream monitoring and experiments

Lily Boxes

Introduction

The Lily Box set-up is an efficient means to gather a broad range of water quality data. The box itself is weatherproof and contains a CR1000 datalogger and battery, which connect via external cables to a suite of water quality sensors and a solar panel. The lily box and solar panel remain in a discrete location on a stream bank, with the cables feeding into the water. Sensors are secured in a perforated PVC pipe and deployed into the stream.



Components (per box)

- Box
  - CR1000 Datalogger (Campbell Scientific)
  - A547 Interface (Campbell Scientific)
  - 35 amp hour battery
  - Charge controller (CZH-Labs)
  - Low voltage disconnect (Binen)
- Sensors
  - Fluorescent dissolved organic matter (Cyclops, Turner Designs)
  - Turbidity (Cyclops, Turner Designs)
  - Conductivity (Campbell Scientific)
  - pH (ISFET, Campbell Scientific)
  - CO2 (Eosense)
  - Dissolved Oxygen (Onset Corp.) – not connected to battery
  - Stage (Onset Corp.) – not connected to battery
  - Pendant (Onset corp.) – not connected to battery
  - Sensor housing: brown perforated PVC pipe
- 35 Amp Hour Battery
- 1x 20 watt Solar Panel (or 2x 10 watt solar panels)

Preparation in Greenland

1. Connect loose sensors to respective cables
   a. CO2, turbidity, FDOM – sensors in black pelican case
2. Wire sensors into CR1000
   a. Follow wiring diagram
   b. Wire in tightly
3. Calibrate sensors within a few days of deployment
   a. Calibration instructions in separate document
4. New DO caps on DO sensors; batteries into pendants

Gather for field

- Laptop
- Sensors and respective cables (CO2, turbidity, FDOM, pH, conductivity)
- Wireless sensors (DO, stage, pendant)
- Lily box
- 35 ah battery
- Brown PVC pipe sensor housing
- Copper wire to ground
- 1x 20 watt solar panel (or 2x 10 watt) – prewired, just connect to charge controller
- Cables
    - CO2 (Eosense): inside each lily box
    - Turbidity and CDOM: some in black pelican case, some shipped separately
- Cable bag (in boxes 10 (Kanger) and 2 (Sisimuit))
    - Cable bag: shuttle, DO and Stage couplers, macro/USB cable
- Tool backpack (screwdrivers, hammer, etc.)
    - Pack: nuts, washers, bolts, anchor bolts, tools, zip ties(action packer 14)
- Hammer drill (action packer 14)
- Hack saw (action packer 14)
- Steel channel (weather station crates)

Stream Deployment

1. Select site and position box sufficiently above water level
2. Launch DO, stage, and pendant on laptop (Hoboware Pro)
    a. Use: Onset Shuttle, DO coupler, Stage coupler, pendant coupler, macro-USB
    b. Above are located in “Cable bags” in boxes 10 (Kanger) and 2 (Sisimuit)
3. Secure sensors into brown perforated PVC pipe, tie with zip ties
4. Secure sensors into stream
5. Open box
6. Ground datalogger – ground before connecting power
7. Set up power system (see below)
8. On laptop, open Loggernet Pro
9. Connect CR1000 to laptop
    a. Serial/USB cord to connect
    b. Unplug and plug-in cord 2x to CR1000 to establish a connection
10. Create new datalogger -> name it -> save
11. Select “connect”
12. Send new program -> “Greenland 15 min”
    a. All stream chemistry Lily boxes receive the same **“Greenland 15 min” program**
13. Check “test” variables to ensure all running smoothly
14. Hit “disconnect,” unplug, secure box lid

Deployment notes

- Ensure the correct program is sent to the CR1000
    - Double check sampling frequency
    - Sending a new program deletes all data on CR1000
- Ensure good connections at all joints between solar panel, battery, CR1000
- Ensure box and battery are secured on stream bank significantly above water level
- Disconnecting from battery for periods of time will not delete data, but will result in data collection gaps

Sensors

Fluorescent dissolved organic matter (FDOM)	 "U" on sensor
Turbidity	 "T" on sensor
Conductivity	
pH	
CO2	
Dissolved Oxygen	
Water Level Logger	

Stream deployment hardware

Item	Use	Location	Photo
Angle iron	Secure PVC housing to rock	Weather station crates	
½ nuts, washers, and bolts	Secure angle iron to PVC housing	Action packer 14	
Hack saw	Cut angle iron to size	Action packer 14	
Hammer drill	Drill into rock	Action packer 14	
Wedge anchors	Secure angle iron to rock	Action packer 14	

Power system

- 35 ah battery
- 1x 20-watt solar panel or 2x 10-watt solar panels
- Charge controller (Binen)
- Low voltage disconnect (CZH-Labs)
- Black (negative) & red (positive) cables
- Copper wire to ground

Solar Panel Charge controller battery Low volt disconnect Datalogger

Connect devices

Ground before connecting power.
All that needs to be done is connecting + connections and – connections at the battery. And solar panel needs to be plugged into the charge controller.

Nutrient dosing experiment

Introduction

The purpose of nutrient dosing experiment is examining biological availability of nutrients in flowing waters. To test nutrient regime, nutrients are added along with a conservative tracer (Cl^-). When injected, only nutrients are biologically consumed longitudinally (Fig. 1). Based on the idea above, nutrient state can be indicated by the ratio between conservative tracer and target nutrient (Nutrient:Cl). After dosing nutrients, streamwater will be sampled until increased nutrient level returns to its initial level. Then the concentrations of nutrients and chloride will be measured and calculate Nutrient:Cl of each sample. Nutrient:Cl over time creates breakthrough curve (Fig. 2). The height of this curve is different among systems depending on nutrient states. Nutrient depleted systems would exhibit lower Nutrient:Cl than nutrient rice systems because nutrient uptake is higher.

This experiment will be simultaneous conducted with dilution gauging to estimate discharge. Dilution gauging can be achieved by dosing only conservative tracer. It will also test the possibility of salt extraction, which can lead to overestimated nutrient concentration.

In Greenland, there will be four dosing at each study site. First dosing will be dilution gauging, which will provide information about discharge and travel time of solutes. Following three dosing will add each nutrient (nitrogen, phosphorous, and iron) with conservative tracer.

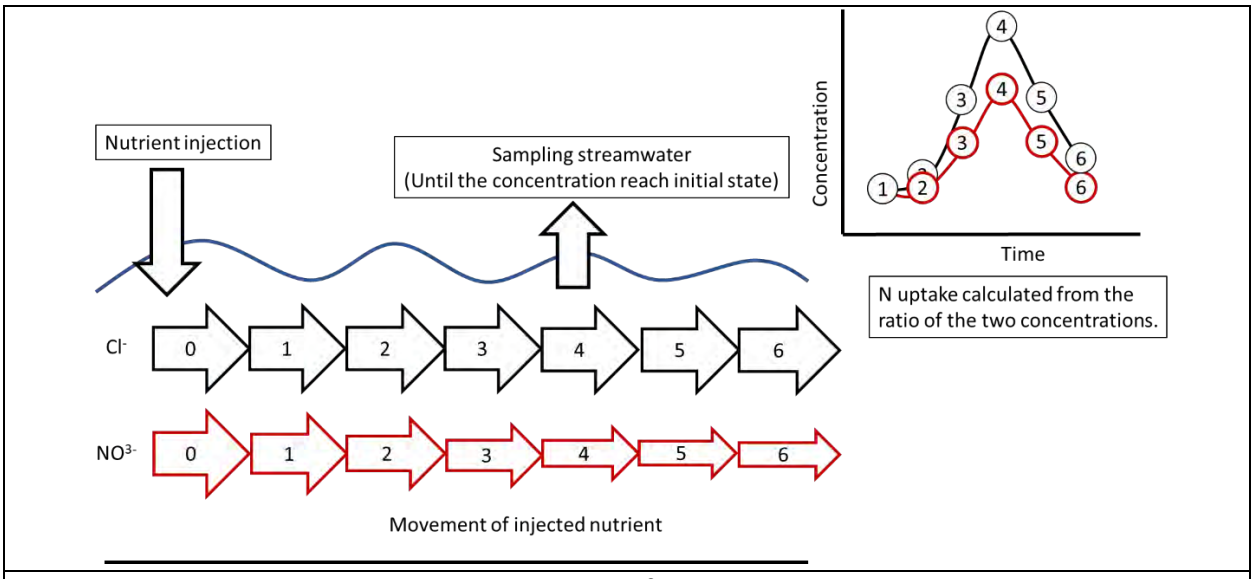


Figure 1. Visualized longitudinal movement of injected tracers and potential breakthrough curves. The arrows indicate the amount of tracers. The size of black arrows (Cl^-) remain the same throughout the reach while the sizes of red arrows (nutrient) decrease as moving toward downstream. The numbers indicate the sequential timing of samples.

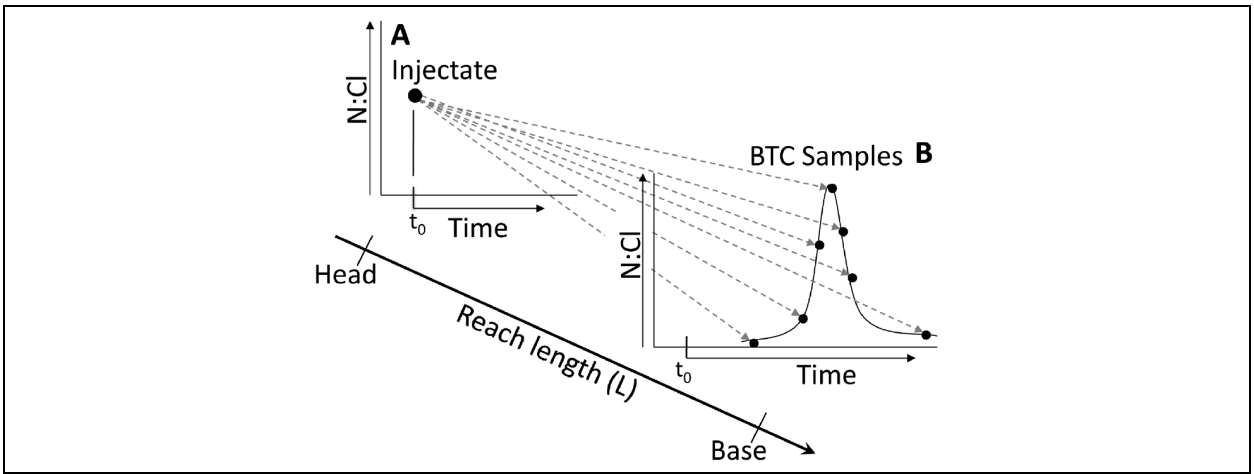


Figure 2. Conceptual diagram describing how nutrient dosing creates breakthrough curve of Nutrient:Cl at the reach base (Covino et al., 2010).

Preparation

1. The following equipment and materials are needed.
   - Measuring tape (or GPS)
   - Portable electroconductivity (EC) meter
   - 200 ml sampling bottles (n=30, label #1-30)
   - 20 ml vials (n=30 per nutrient type, label site-nutrient type-#)
   - Syringes

- End-cap filters
- Salts (Preweighed)
    - Conservative: NaCl
    - Nutrients: NH_4Cl, H_2KPO_4, EDTA-Fe

1-1. Preweigh salts following dosing ratios below.

Nutrient type	Salt	Dosing ratio (kg-salt per m^3/s)	Target concentration (mg-nutrient/L)
Cl (conservative)	NaCl	3	7.5
N	KNO3	2.9	1
P	KH2PO4	0.43	0.25
Fe	EDTA-Fe	0.49	0.2

Site information

2. Measure the width of streams at 5-10 cross sections over a reach where nutrients will be dosed.
    - The length of the reach is at least ~15 times of the width.

3. Measure the depth 5 times in each cross section.

Nutrient injection & Dilution gauging (first dosing)

4. Separate working group into two. Each group respectively goes to the top and the bottom of the reach.
    - At least three people are required: one at the reach top and other two at the reach bottom
    - Top: Salts, a bottle (or a bucket), walkie-talkie, one sampling bottle
    - Bottom: Sampling bottles, syringes, end-cap filters, vials, EC meter, notebook

5. Deploy an EC meter at the bottom of the reach. One consistently monitors EC while another one gets ready for sampling streamwater.
    - Set the sampling frequency as 30 seconds or faster.

6. At the reach top, Mix the salt with streamwater in a bottle or bucket.
7. Sample the mixed saltwater. It will be later filtered with syringe and end-cap filter and collected in a vial.
    - The concentration will be measured to make sure about the ratio between conversative tracer and nutrients of dosing.

8. Pour the mixed saltwater at the top of the reach. Rinse the bottle or bucket with streamwater to make sure that all added salt are injected. Start counting time.
    - Set the time of the injection prior to the injection and share it with the group at the bottom.

9. Sample streamwater with a regular interval at the bottom of the reach. Distribute samples to catch pre-arrival, rising limb, peak, recession limb, and tail. Double the sampling frequency around the peak (Fig. 3).

- The person monitoring EC counts time after the injection. Let another one know when to sample streamwater. Record the time of sampling and EC at that moment. Another person samples when told and places the samples on a safe ground.
- During the first dosing (dilution gauging), sampling frequency is set by the one monitoring EC. Before the rising limb, sample streamwater every 10 minutes. From rising limb to decreasing limb, sample every 1 minute.
- For other nutrient dosing, sampling frequency is set based on the EC curve monitored during the first dosing.

10. After collecting 30 samples, filter them using syringes and end-cap filters. Collect filtered water directly in vials.
11. Wait long enough before next dosing to make sure that the injected salts are washed out from the reach. Waiting time is usually three times longer than peak timing. For example, if the concentration peaks 20 minutes after dosing, waiting time would be 60 minutes (1 hour).
12. Repeat the processes 6-10 for each dosing.

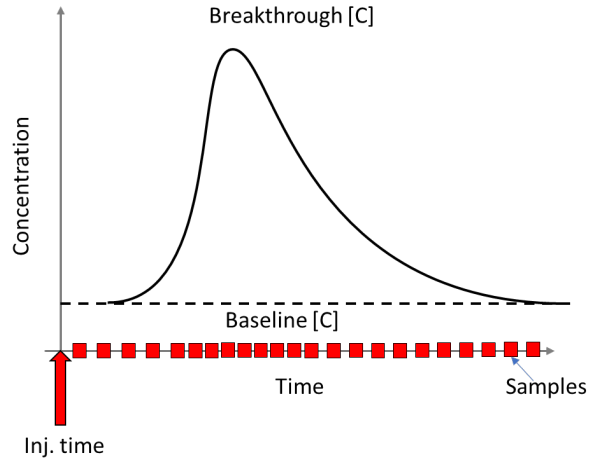


Figure 3. Distribution of sampling throughout the breakthrough curve. Red squares below are the timing of sampling. Sampling is more frequent around the peak.

Nutrient diffusing substrate

Introduction

Nutrient diffusing substrate (NDS) tests how nutrient addition alters microbial growth. NDS is made of solidified agar solution mixed with target nutrient. Solid agar will be stored in a small polycon cup, whose lid has a hole. On the top of the agar, glass filter will be placed, where nutrients diffuse and microbes grow. NDS will be deployed for ~2 weeks under streamwater. After retrieved, microbial biomass on glass filters will be measured. If nutrients are limited, microbial accumulation on the glass filters will be significantly higher at nutrient added NDS than at control NDS. If there is no nutrient limitation, no significant difference between NDS will be found. In Greenland, effects of three nutrients (nitrogen, phosphorous, and iron) and colimitation of those nutrients will be studied.

NDS preparation

1. The following equipment and materials are needed.
    - 1 oz polycon containers with screw-lid
    - 2.7 cm glass crucible covers
    - Granulate agar
    - Ammonium chloride (NH_4Cl)
    - Potassium phosphate (KH_2PO_4)
    - Ferric EDTA
    - Scale
    - Hot plate
    - Stirrer
    - Volumetric flasks
    - Beakers
    - Plastic L-bars
    - Zip ties

2. Drill a hole at the container lid to fit the glass crucible covers.
3. Label each container with a corresponding nutrient type.
    - Control / N / P / Fe / N+P / N+Fe / P+Fe / N+P+Fe

4. Make agar solutions.
    - Boil 200 ml of distilled water and continuously stir it.
    - Add granulated agar and nutrients as mentioned in the Table 1.
    - Be cautious after adding agar. The solution can easily boil over.

Table 1. Salt types and required amount of each salt to make NDSs.					
Type	Salt	Molar weight (g)	Desired molarity	Salt added (g)	Agar added (g)
Control	-	-	-	-	4
N	NH_4Cl	53.5	0.5	5.35	4
P	KH_2PO_4	136.1	0.5	13.61	4
Fe	$C_{10}H_{12}FeN_2O_8$	344.1	0.2	13.76	4
N+P	NH_4Cl KH_2PO_4	53.5 136.1	0.5	5.35 13.61	6
N+Fe	NH_4Cl $C_{10}H_{12}FeN_2O_8$	53.5 344.1	0.5 0.2	5.35 13.76	6
P+Fe	KH_2PO_4 $C_{10}H_{12}FeN_2O_8$	136.1 344.1	0.5 0.2	13.61 13.76	6
N+P+Fe	NH_4Cl KH_2PO_4 $C_{10}H_{12}FeN_2O_8$	53.5 136.1 344.1	0.5 0.5 0.2	5.35 13.61 13.76	6

5. Fill the containers with the solution.
    - Solution is ready when it becomes transparent.
    - Pour until the containers are fully filled.

6. Allow 10-20 minutes to cool and solidify.
7. Place a glass crucible on agar surface and shut the lid.
    - Be sure that the crucibles are firmly held.

8. Fix the containers to the L-bars with zip ties and silicon glue.
    - NDS types should be randomly arranged on the L-bars.

9. Wrap the containers in aluminum foil and keep them in the refrigerator until the deployment.
    - Don’t expose the crucible to air more than an hour, or the solution will dry.

Figure 1. The cups filled with the agar solution. Crucible glass filters are on the solidified agar solution (left). After close the lid, the top of the NDSs are covered by aluminum foil to block light (right).

NDS deployment & retrieval

1. The following equipment and materials are needed.
    - NDS sets
    - Cooler
    - Stakes
    - Aluminum foil

2. Place the L-bars in a riffle next to each other.
    - The containers should be entirely submerged, but close enough to the stream surface for sunlight.

3. Position the bars parallel to flow preventing sedimentation (Fig. 2).

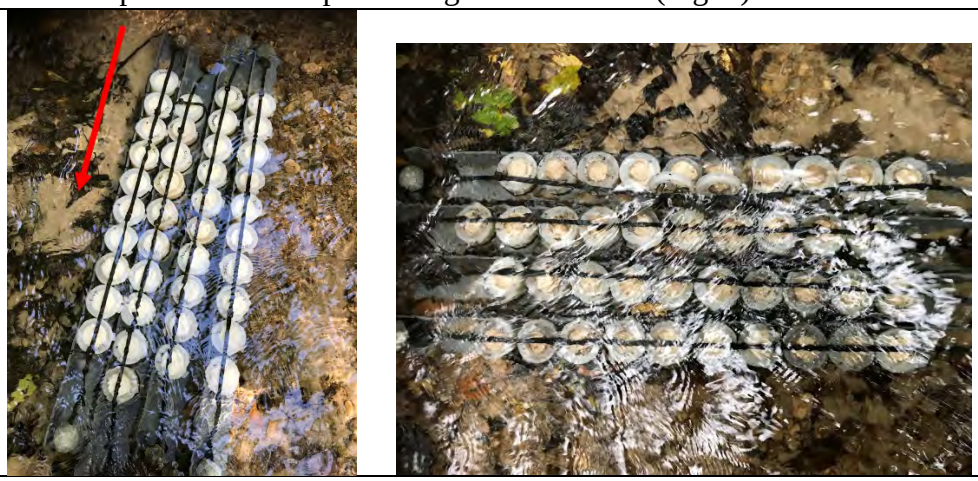


Figure 2. Deployed NDS. The red arrow indicates the direction of flow.

4. Secure the bars into stream bottom with stakes.
5. Deploy NDS ~14 days.
6. After ~14 days, retrieve NDS sets from the stream bottom. After remove the bars from stream, immediately wrap the containers with aluminum foil or immediately remove the disks, put them in separate Ziplocks, and store on the ice.
    - If the crucibles expose to sunlight, chlorophyll will be degraded.

7. Before further analysis, keep the containers (or disks) in a freezing state.

Weather Station

Introduction

Three weather stations will be established in Greenland to collect climate data throughout the years of the project. Two stations will be in Kanger, with the third in Sisimiut. The data will include photosynthetically available radiation, wind speed, wind direction, relative humidity, precipitation accumulation, and temperature.

Components (per station)

- Campbell scientific WxPRO weather station: https://www.campbellsci.com/wxpro
  - Tripod
  - Main tower with lightning rod
  - Crossbar
  - Misc. hardware
  - Stand-alone post for snow accumulation and rain gauge
- Sensors
  - CS215-L Air Temperature and Relative Humidity
  - 03002-L Wind Sentry Set
  - TE525-L Rain Gauge with 6 in. Orifice
  - CS300-L Pyranometer
  - Game camera
- Box
  - CR800 Datalogger (Campbell Scientific)
  - Charge controller (CZH-Labs)
  - Low voltage disconnect (Binen)
- 100 amp hour battery + black housing
- 50 watt solar panel

Gather for field

- Laptop
- Weather station crate
- Sensors (wind set, relative humidity + temperature, pyranometer, rain gauge)
- Game camera
- Datalogger box
- 100 ah battery + black housing
- 50 watt solar panel
- Cable ziplock bag (in boxes 10 (Kanger) and 2 (Sisimuit))
  - Cable bag: serial/USB cable
- Tool backpack (screwdrivers, hammer, etc.)
  - Pack: zip ties, nuts, washers, bolts, anchor bolts, wrenches, etc. (action packer 14)
- Hammer drill (action packer 14)

Deploying in field

1. Hardware
    a. Expand tripod
    b. Insert main tower into the tripod
    c. Attach crossbar - should be 1 meter above ground
    d. Adjust and tighten bolts
    e. Secure station:
        i. Can use hammer drill to drill into rock, secure with angle irons
2. Sensors
    a. Attach with hardware in crates - see photos below
    b. Zip tie cables neatly to crossbar
    c. Feed cables through hole in datalogger box
    d. Secure the game camera to tripod with zip ties, check batteries and setting
3. Rain gauge + precipitation post
    a. Secure to ground: hammer drill and anchor bolts or simply using the lighter posts
    b. Secure rain gauge to post – ensure in frame of game camera
    c. Run rain gauge wire to datalogger box
4. Datalogger box
    a. Attach with zip ties or wire to the tripod - hole for cable entry must face down
    b. Feed cables through hole of box
    c. Wire sensors into CR8000 – see diagram
    d. Calibrate pyranometer and wind sensor – see wiring diagram
5. Power
    a. Ground station and datalogger to the physical ground with copper wire in crate
        i. Ground first, then connect to power
    b. Attach 50 watt solar panel (prewired) to blue charge controller
        i. Red electrical tape signifies positive (+)
6. Datalogger
    a. Indicator light will **blink every 3 seconds** to indicate **power on**
    b. Wire sensors in according to wiring diagram
    c. Connect laptop to datalogger - serial to USB cord
    d. Open Loggernet
    e. Connect datalogger
    f. Send program (in Greenland folder)
        i. When searching for program, be sure “all files” option is on.
    g. Light will **blink every 15 seconds** to indicate **program is successfully running**
    h. Disconnect from the datalogger, close lid, leave.

Reference photos

Crossbar attachment

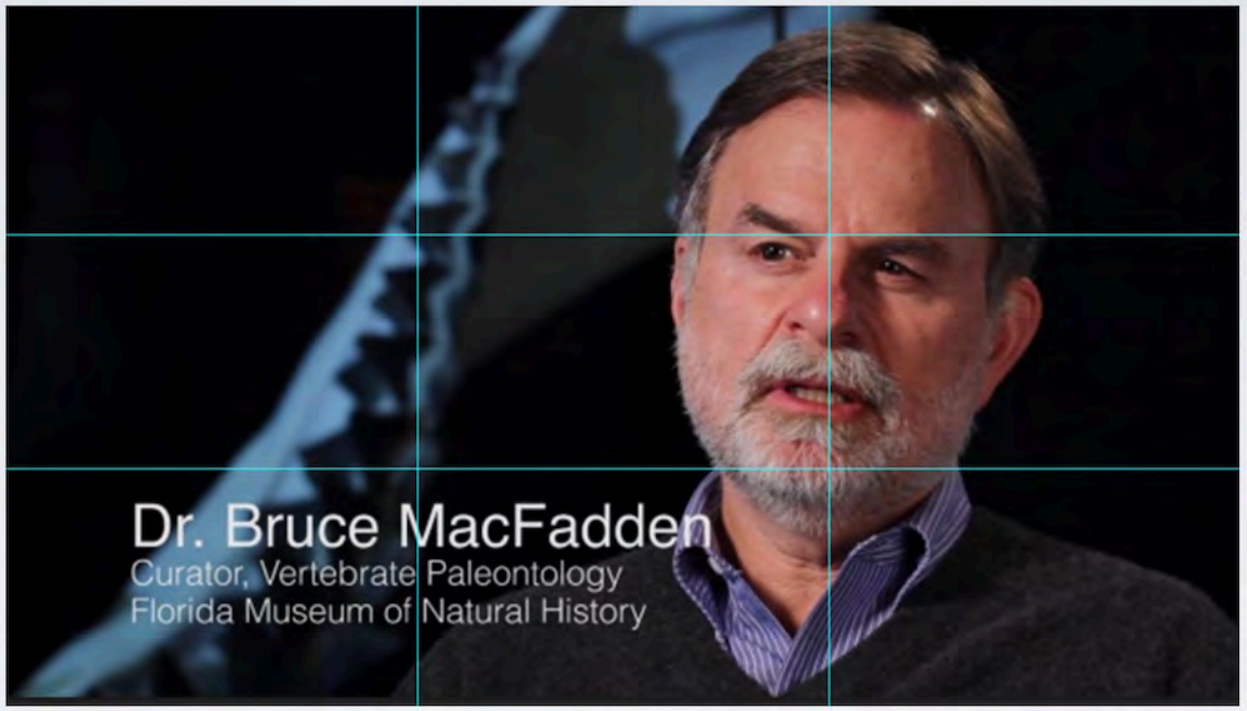

Sensor positioning

Wind sensor


Video and Still Imagery for WIGF Narrative Stories

Framing the Image - The Camera as the Eye of the Audience

- Landscape as compared to Portrait orientation
  - Landscape - Horizontal rectangle - Television and computer screens
  - Portrait - Vertical rectangle - Most cell phone photos - Tik-Tok, etc
  - Landscape is the norm for professional projects
- Composition within Frame - Where the eye is drawn



  - Rule of Thirds - Lines on vertical and horizontal thirds form 4 intersections
  - Looking Room - Subject on one vertical thirds with space on the other
- Subject Angle to Camera
  - Generally off screen - addressing an interviewer or another person in scene
  - Directly to camera - intentionally addressing the audience - newscast
- Camera Angle to Subject
  - Eye level - emphasis on subject or main activities in the scene
  - High level - emphasis on background - diminished relevance of subject
  - Low level - emphasis on foreground - increased authority of subject
- Basic Progression for sequence of images in each scenario
  - Wide or Establishing shot - provides overall perspective of the setting
  - Long shot - includes all the essential components of the setting

Medium shot - emphasis on two elements within the setting

Close-up - details of one of the elements

Return to Medium or long shot to show progress of action

- Additional shots can include a medium close-up and extreme close-up
- These are used as needed to illustrate details and interactions

Stability of Images - Remember the Camera is the Eye of Audience

A tripod is the most stable - be sure that the image is level with horizon

Gimbals for cell phones - handheld devices with balance features

Cases for tablets with handles - two handed with elbows against chest

Handheld phones - set on horizontal surface or hold with elbows to chest

- Motion with the camera - consider long term stability for viewer

Pan - on tripod rotate camera / image horizontally to left or right

Pan - if handheld rotate hips with elbows against chest

Tilt - on tripod move camera / image vertically up or down

Tilt - if handheld bend at waist with elbows against chest

Zoom - using adjustment on device screen can be difficult

Zoom - can also be accomplished by walking toward or away from scene

Image Exposure and Color Tone

Generally exposure and tone handled automatically by device

Bright background can cause underexposure on foreground subjects, so be aware of device methods to increase sensitivity of darkened areas

These same methods should be able to decrease exposure when subject is too brightly lit, which eliminates details of the subject. Underexposure is usually corrected more easily by software later than overexposure.

Recording plan and techniques

Have a general idea of what you need to capture to convey the story

This would include awareness of various components of events

Be prepared to record aspects of events that you weren't expecting

When possible start recording a few seconds before the action occurs

Allow the recording to continue for several seconds after completion

Have a sense of when to request a do-over of a demonstration

If you don't have the footage, it can't be used or fixed during the editing

B-roll footage are elements that may be reactions from others observing the activity, details of some of the equipment, landscape or watershed views, and other images that are part of the scene that may not seem important at the time.

Record video at 1920 x 1080 at 30 frames per second quality or better.

Be aware of the storage capacity of the recording device.

Have external storage device available to transfer footage onto each day.

Use and name individual folders for storage of each day's recordings

Audio - another aspect of recording

The microphones built into devices work pretty well at 6 - 10 foot range

Record important dialogue as close as is practical for the camera placement

Consider a separate recording where the "talent" describes the process that has just been recorded.

Make an effort to record B-roll ambiance sounds of the location and activity

It's helpful to have headphones to hear sound quality during recording process.

Software

- Software for Video Editing

  Desktop/:Laptop - Apple iMovie (free) Final Cut Pro ($300) - Adobe Premiere Pro (Student Subscription) for Apple or Windows - Lightworks - Windows, Linux and Mac (free version and purchase) https://www.lwks.com

  UFApps https://apps.ufl.edu/Citrix/UFAppsWeb/ provides 2 video editors: Movie Plus and Movie plus 8. A YouTube tutorial is available here: https://youtu.be/7fBIe09kGXc.

  Training for Final Cut and Premiere Pro available on UF eLearning - Linked In - Lightworks provides tutorials on the website

  Devices - Apple iMovie and others on App Store - Android - FilmoraGo and others referenced here: https://www.androidauthority.com/best-video-editor-apps-android-716248/

  Training for Devices also available on UF eLearning - Linked In

- Software for Photo Editing

  Adobe Photoshop and Lightroom are industry standards in Mac and Windows platforms (Student Subscription)

  Microsoft Windows 10 Photos included with software

  Mac OS X includes Preview - pdf reader with photo editing capabilities.

- Transcription Services

  https://get.otter.ai Automated video transcripts - 600 minutes free per month

https://www.rev.com - Automated and Human Transcripts at different rates

- Reference materials

  Techniques of Visual Persuasion + creating powerful image that motivate (2021) Larry Jordan - covers Photos, Videos and Presentations - www.peachpit.com - Print version - ISBN-13: 978-0-13-676679-7 ebook ISBN-13: 978-0-13-676689-6

  Smart Phone Cinematography - https://moviola.com/courses/smartphone-cinematography/lessons/the-basics/

- Questions?

  email Michael Munroe - munroe@ufl.edu

Principles and Guidelines

Introduction

The combined SILA and WIGF projects include a large and diverse team of collaborators working toward the common goal of understanding the role of glacial cycles in hydrological systems of the Arctic. The diversity of our team is one of its primary strengths. To best use that strength, we must have effective and frequent communications to ensure equitable distribution of project resources and to identify and resolve any conflicts or diverging goals as soon as possible. This document describes some principles to guide our communication and represents a consensus opinion of the most equitable use of available resources, attribution of discoveries made, and rights and responsibilities of all participants.

Participants

Although our project has a universal overall goal, we have a range of experience, various expertise, and an array of individual objectives that connected us to the projects in different ways. Each of us, regardless of seniority, has an equal right to guide project activities so that the activities' outcomes support individual objectives. However, our responsibilities to the project, and by extension to the funding agencies, mean that individual objectives, when supported by the project grants, should address project goals to ensure they are achieved.

The leaders of the project include the Principal Investigators (PIs) and Senior Personnel (SP) on the SILA and WIGF grants, who are formally charged with accomplishing project goals and making certain the outcomes are documented through proper data management, timely project reports, and peer-reviewed publications. The PIs and SP will be supported in those tasks by graduate students and post-doctoral researchers, who are integral members of the team with similar rights and responsibilities as the PIs and SP. The graduate students and post-doctoral researchers may be supported through either or both SILA and WIGF projects. They have immediate goals to complete dissertations and write peer-reviewed publications, which are paramount to advancing their careers as they document research accomplishments. All PIs and SP should support those efforts both for the good of the project as well as advancement of the students and post-docs. An additional group of participants includes faculty and students who are not included as PI or SP on either the WIGF or SILA proposal, but have a scientific interest in the project. Although not formally included in the proposals, this group enriches the overall project through contribution of their expertise. The group composition may change over the time frame of the project but nonetheless members of this group should be treated as participants of the team and supported as much as possible given fiduciary and financial constraints in the grant funds. Undergraduate students may also join the project periodically to help with tasks as needed and to further their educations, for example through writing of senior theses. They should be treated with respect, allowed to explore ideas of their own, and be expected to contribute to decision-making for the project. In return, they will be expected to complete their tasks in a timely manner, including writing of theses.

The diversity of participants dictates that the rewards of the project should be equitably distributed depending on the activities of individuals and needed project outcomes. The junior members of the team will be encouraged to take leadership roles in developing project plans and outcomes, while recognizing that the responsibility of the outcomes remain with the PIs and SP. Therefore, the PIs and SP have the authority over final decisions and if differences arise among the PIs and SP that cannot be resolved, the final authority rests with the lead PI of the proposals (J. Martin). For these reasons, one of our guiding principles must be open, equitable, and effective communication across the strata of expertise and responsibilities. This communication is particularly important for sharing of mutual resources, including financial and infrastructure that support this work.

Finances

The project is currently supported by two funding sources, one from UF (WIGF) and the other from NSF (SILA). Funds available from the WIGF include student stipends and tuition. Use of these funds is fixed by the Graduate School and cannot be changed. An additional small amount of WIGF funds is available to support integrative activities that will aid development of collaborations within the entire cohort, including faculty and students not included in the WIGF proposal. What constitutes an integrative activity has few restrictions. Decisions about which activities should be supported will be reached by consensus of the group, recognizing the small amount that is available and that it is intended to support the group rather than individuals.

Current grant support may be augmented in the future from additional proposals that have been or will be written. If those new proposals are successful, their resources will be directed by the PIs of those proposals and dedicated to supporting activities of the groups submitting the proposals. Because new proposals are linked to this project, they may supplement mutual efforts through blending of activities of the different proposals. If new proposals require support of either the WIGF or SILA proposal, that support needs to be stated explicitly in the new proposals.

The financial resources available from NSF have two primary funding streams. One is administered by Polar Field Services and provides logistical support for some, but not all, travel expenses to Greenland and while in the field. Although those funds are external to UF, we will have to make decisions prior to the field season and while in the field about how our activities impact those budgets. The second funding stream is provided directly to UF. For internal accounting purposes, this part of the budget is divided among the departments that house each PI and SP according to the budget that was approved by NSF. These internal budgets cover a variety of activities, including costs that will be charged to individual laboratories and costs that are required to support communal project activities. Costs that could be considered communal include:

- **Permits.** Several different permits may be needed for our field work and some may have fees associated with them. Permit costs are communal because they will allow all field activities that are the central activity of the project.
- **Field equipment.** Much equipment is already available from many laboratory inventories, but that equipment may need refurbishing or have costs associated with expendables. In addition, new equipment may need to be purchased or constructed.

Some new equipment is included as line items if it was known to be needed when the proposal was written.

- **Field Travel.** Although costs for US-Greenland flights, within Greenland flights, and lodging are supported by the Polar Field Services budget, additional travel costs not supported by the Polar Services budget include travel to and from Scotia, NY, food and lodging at Scotia, and food expenses while in Greenland. These costs should be divided among the UF budgets according to the Budget Justification.
- **Analytical costs.** Specialized equipment that may be needed for analyses exists in many of our laboratories. Those instruments may have formalized recharge costs. If no formal recharge costs have been established, costs related to running the instruments, including taxes levied by the UF Research Office for laboratory space, should be supported by funds of those involved in the analyses.

We should be aware of these costs and be willing to share the expenses associated with activities that benefit the communal project. For those units that allow the designation of overhead return to campus centers, the Water Institute should be the designated center.

Data

The ambitious program set out in the SILA proposal will generate an enormous amount of data based on both field observations and laboratory analyses. A list of data to be generated can be found in the Data Management Plan in the SILA proposal (appended below). Because these data will be the foundation for all project outcomes, it is critical that we take the utmost care in generation, documentation, and storage of the data. Because these data may be used for multiple purposes, and to enhance the interdisciplinary nature of the project, we will need to share data among ourselves. However, data sharing should support distinctly different interpretations and not be used to address duplicative questions, hypotheses, or ideas. We may develop a data management committee if issues arise during the sharing of data.

In addition to sharing data internally within the project, NSF has specific rules governing access to those data by external researchers, which we are obligated to follow. As described more fully in the Data Management Plan, data must be made publicly available within two years of collection or by the end of the award, whichever comes first. Although many data portals now exist, we are required to submit all of our data to NSF's Arctic Data Center (https://arcticdata.io/) because our support comes from the Office of Polar Programs. We may choose to submit to other sites as well, such as the Greenland Ecosystem Monitoring (GEM) database (https://data.g-e-m.dk/) and the Dryad data repository (http://datadryad.org/) when appropriate, but those submissions must be in addition to submissions to the Arctic Data Center. Each of those data repositories has strict formatting rules with which we must become familiar prior to and during data collection to ensure that data formats are appropriate for the repositories and internal data sharing can be accomplished smoothly. Each laboratory will be responsible for uploading data in the required time frame. Formatting and uploading protocols should be shared between groups.

Products and Authorship

Peer-reviewed publications and presentations at scientific meetings are likely to be the most important outcomes of our research efforts, although other products, such as videos,

interviews etc., may also be important outlets for results of the project. Products, particularly papers, from large collaborative groups such as SILA are often developed in parallel, which may create the potential for redundant or similar products, particularly as data and ideas are shared. To reduce this potential, ideas for products should be provided to the group as soon as they begin to develop. Commonly, peer-reviewed paper development starts with writing and submission of abstracts to conferences. Prior to submission, the ideas contained in the abstracts should be presented to the group, ideally both in oral and written forms, by the lead authors of the work, at which time authorship of the abstracts can be discussed. Ideas in abstracts are commonly modified as talks are prepared. Practicing conference talks could provide an avenue to enhancing the ideas of the presentation and improve ideas to be included in peer-reviewed papers stemming from the abstract. These early stages of paper development are appropriate times to discuss the author list and authorship order, which should reflect the collaborative research efforts of others in the project who may have or will contribute to the papers in substantial ways.

The lead on conference abstracts and presentations will likely become the primary author of peer-reviewed paper and will take the lead on continued development of the ideas to be presented, outline the scope of the paper, and write most or all of the initial draft. Primary authors are likely to be post-doctoral researchers and graduate students, who should be encouraged to lead the writing effort so that the papers may be included in their dissertations and provide the best enhancements of their careers. These papers would be expected to be developed initially in collaboration with the students' and post-docs' advisors and this sub-group of the project should develop a tentative secondary author list early in the development of the paper. Secondary authors could be included by contributing to the paper in many different ways. However, in all cases, contributions should reflect a meaningful intellectual engagement with the ideas contained in the papers. This engagement may take many forms including:

- Initiated and framed the research questions and hypotheses;
- Designed, led, and helped with field sample collections or observations;
- Analyzed samples in field and institutional based laboratories;
- Provided or helped with data analyses and modeling;
- Provided substantive comments on the paper prior to and during writing;
- Written portions of the paper.

The preliminary author list may change for many reasons as papers are developed, for example, if additional data or modeling are needed to complete the paper, new ideas are contributed to the paper, or writing efforts beyond cursory editorial advice are provided. In addition, the timely submission of papers is paramount to the advancement of the overall project, and consequently, the authorship list and/or order may change if a paper is stalled at the start or during the writing stage.

When the initial author list is shared with the group at the outset of paper development, anyone included as co-authors may opt-out of being included on the list should they feel they have not or will not contributed in a significant way. Similarly, others may wish to opt-in if they feel they have or could contribute in meaningful ways as described above. The way in which they will contribute should be described explicitly and those included in the initial author list will decide if the new author's contribution will substantially improve the paper and if so will include the new author. Alternatively if the contribution is a new direction that will significantly change

the direction or scope of the original paper, a second paper should be considered with its own authorship list.

Acknowledgement of financial support is required for all products. To acknowledge NSF support, publication (including websites) must include the following statement: "**This material is based upon work supported by the National Science Foundation under Grant No. OPP-2000649**". NSF support must be orally acknowledged during interviews and at conference presentations. The following disclaimer must also be included in publications other than scientific, technical, or professional journals: "**Any opinions, findings, and conclusions or recommendations expressed in this material are those of the author(s) and do not necessarily reflect the views of the National Science Foundation**". Acknowledgements must also be made to the Water Institute for support of the WIGF, including in all publications, presentations, and through social media. Should other funding agencies support work of the group, proper and appropriate acknowledgements must be made to those supporting agencies as well. Additional acknowledgements should be made as appropriate, particularly to others in the group who helped with the work but are not included in the authorship list, and to others who helped facilitate the work, such as personnel at Polar Field Services and technical support for individual labs.

Reporting

Annual project reports will be due to NSF prior to the anniversary of the project start date, which is August 1. Because SILA is a four year project, the reports will be due 2021 through 2023. We are likely to request no-cost extensions for additional years, which will add annual reporting requirements. The final year of the project will trigger a final technical report and a statement of general outcomes, both of which will be due 90 days after the end of the project on October 31 of the final year. Each PI managing budgets through SILA will be responsible for reporting their activities and accomplishments using the NSF template, which will be provided as the annual reports become due. These reports will be compiled by the lead PI (J. Martin) and submitted to NSF as a single report. Contributions from individual labs must be provided in sufficient time to compile them and thus they should be completed at least one month prior to the due date for the reports. Late reports have the potential to jeopardize funding for this project and other grants. Additional information may need to be provided to the Water Institute on the progress and outcomes from the project. That information is likely to be requested annually.

Data Management Plan

The work described in this proposal will lead to the production of samples and data that will be of use to other researchers examining hydrology, water chemistry, microbiology, ecology, plant distribution, and weathering in Greenland. The data will be valuable to a range of additional researchers including ecologists working on high latitude cycling of carbon and nutrients, climatologists interested in shifts in ecosystems with polar warming, and paleoceanographers working on marine isotope records of glacial – interglacial cycling.

Expected data:

- Weather data from 4 locations extending from the ice edge to the coast across a gradient of precipitation. Data will include precipitation, snow fall, temperature, relative humidity, solar radiation, daily photographs and wind speed and direction.
- Soil temperature gradients at the weather station locations.
- Stream discharge derived from measured stream stage-discharge relationships and logged (15 minute interval) stream stage at five locations across the ice to coast transect.
- Time integrated solute fluxes from surface-water passive flux meters.
- Stream and meltwater samples in time series at daily and sub-daily frequencies, three times over ~30 days in years 2 and 3.
- Dissolved greenhouse gas concentrations (CO_2, CH_4, N_2O) collect at same time and frequencies as the water samples. All gas samples will be consumed during analyses.
- Active layer water samples from deglaciated watersheds and sandurs along with head gradients between streams and active layer water.
- Solid samples: suspended sediment in the proglacial streams (with daily water sampling), bedload sediment from all streams (once years 2 and 3 each), and dust collected from air and trapped in annual snow fall.
- Plant diversity and abundance along transects that cross gradients of water availability and topography.
- Nutrient diffusing substrate results including 3 replicates and 5 controls.
- Whole reach scale injections of reactive solutes with monitored responses with loggers and water grab sampling.

- N_2O concentrations measured in the laboratory. (CO2 and CH4 will be measured in the field)
- Water chemical (major, trace, and nutrient) concentrations, and isotopic (H, O, C, Sr, and Nd) compositions utilizing in house laboratories (see Facilities and Resources). Most water samples will be consumed during analyses but remaining aliquots of water will be preserved at 4^o C for a minimum of 5 years.
- Aliquots of bedload and suspended sediment samples will be digested before and after sequential leaching for various components (Fe and P contents and Sr and Nd isotope ratios). Residuals of the samples will be archived at the University of Florida.

- Culture-independent assessment of microbial community structures in water, sediments, and soils. Physiological measurements to assess specific activities and rates of microbial metabolism.
- Chemical concentrations, and isotopic compositions (see above), will be measured in selected plant material. Plant samples will be accessioned at FLAS (see Facilities) and digitized to facilitate access from other Arctic researchers.

- Curricula for Greenlandic high school classes will be created at appropriate levels through collaboration with Greenlandic high school teachers.
- Fact sheets about climate and environmental change in the Arctic will be created. The fact sheets will be designed for dissemination through the tourist industry and modified to levels appropriate for use in secondary schools in Greenland.

Period of data retention:

All of the data generated in this study will be made publicly available within two years of data collection or by the end of the award, whichever comes first. Thus, we expect for data collected during the field seasons in years 2 and 3 of the project to be made available by 2023 and 2024, respectively. Data derived from sample analyses in our laboratories will require additional time after the field seasons, but our expectation will be completion of these analyses within a year of return from the field. Curricular products will be made available as they are created.

Data formats and dissemination:

- Quantitative discrete data (e.g., discharge data, chemical and isotopic compositions of gases, water, and solids, results of manipulative experiments) will be tabulated in spread sheet formats in as simple organization as possible with detailed metadata descriptions of the data organization. The metadata will include all general pertinent information including sampling locations, times, durations, and data available in the files. Dissemination will occur through peer-reviewed publications and access through data archiving sites.
- Nucleic acid sequence data: data resulting from nucleic acid analyses will be archived with Genbank, a standard repository for sequence data, and MG-RAST, a second repository for both sequences and metadata. Sequences deposited with MG-RAST and Genbank will be available to the public for use independent of the investigators.
- All plant distribution data will be published in the Dryad data repository (http://datadryad.org/), with a unique DOI where it will be publicly available and easily accessible, following recommended standards.
- Curricular materials will be made available to all Greenlandic high school teachers through the Greenlandic Web Portal, which is restricted to use within Greenland. Additional curricular material will be created through collaborations with U.S. high school teachers and made available through open access websites.
- Base electronic files will be made available through open access websites. Additional limited print copies will provided to distribution centers (e.g., Arctic Circle Business).

Data storage and preservation of access:

All metadata files, full data sets, and derived data products, will be archived at the Arctic Data Center (https://arcticdata.io/) and in peer-reviewed publications. As appropriate, data from this project will also be archived at the Greenland Ecosystem Monitoring (GEM) database (https://data.g-e-m.dk/), and the project description and metadata pointing to the data archive sites will be provided through Isaaffik – the Arctic Gateway (ahttps://www.isaaffik.org/).

Code of Conduct

Goals

This document has multiple goals related to our time in the field in Greenland and interactions within our group on campus. The document describes best behaviors and remedies in the case of inappropriate behaviors. Specific goals are:

- To provide a safe and respectful environment for all SILA and WIGF team members,
- To outline behavior expected of SILA and WIGF team members while in the field and on campus, including interactions with others outside of our group,
- Ensure all team members have access to resources and support for addressing and resolving interpersonal conflicts, including discrimination, harassment, and sexual assault,
- Detail specific reporting procedures and outcomes to enable rapid communication and response in the event that any team member feels unsafe.

Expectations

All team members in the SILA and WIGF projects have the right to be free from discrimination, unlawful harassment, sexual misconduct, and violence. That right can be supported through frequent communication among all team members, including students, post-docs, and faculty as a way to coordinate responses related to issues of concern and head off possible conflicts before they become serious. If members are uncomfortable discussing issues directly with others on the team, they may also contact others outside of the immediate group, including Water Institute staff, an individual's departmental administration, or the UF Title IX office (contact information is provided in Appendix B the end of this document). The Title IX office supports the federal law (Title IX) that gives everyone the right to equal access to education and employment in the absence of sexual harassment and gender discrimination. Note that faculty and staff are required to report any type of sexual or gender-based discrimination, harassment, or misconduct to UF's Title IX Coordinator who cannot provide confidentiality but will maintain your privacy. If you need to talk to someone confidentially, faculty and staff can point you to a counselor or you can reach out to one or more of the confidential contacts listed in Appendix B.

All team members are expected to follow the law, as well as specific guidelines laid out in this and other Codes of Conduct, and behave in a manner that does not infringe upon the rights of others. Violations will result in serious sanctions.

Overview of the Greenland research environment, and SILA, WIGF, NSF, and UF policies:

The success of both SILA and WIGF depends on interactions among many people including those of us directly supported by the project as well as others who will provide logistical support to facilitate our research. In addition, we will be living in towns and will have frequent contacts with local residence and many tourists passing through the regions. Our behavior while in Greenland will reflect on our group, the NSF, and the U.S. We must treat everyone with whom we interact in a humane, gracious, and respectful manner. This applies to all members of the SILA team, including PIs, graduate students, and those fulfilling other roles.

Some specific situations to be aware of, where exemplary behavior is paramount, are listed below:

- We will be a large group while in the field and will be working closely with each other including shared field gear, time, and activities. We must respect each other's privacy and help as much as possible to complete all tasks, scientific and domestic, that will lead to the success of the project.
- We will have support from, and rely on, employees of Polar Field Services, the company that provides logistical support to NSF. They are passionate about their work and are eager to help us complete our science. But they will be busy with many tasks as they support other field parties as well. Have patience in your interactions with them.
- Many local residents will provide support (vehicles, housing, etc.) through contracts with Polar Field Services and there will be additional interactions while in town, at airports, stores, and restaurants. People serving you in those capacities deserve respect and to be treated civilly.
- While in the field, practice the same consideration for the environment you would anywhere else in the world. We have an important responsibility as representatives of the Arctic science and University of Florida communities to demonstrate respect for both the local communities and the surrounding environment.
- Particularly while housed at KISS, we will be in close quarters with many research groups from around the world and will be sharing many facilities – laboratories, meeting space, bathrooms, and kitchens. It is essential that we are good citizens and respectful of other's privacy and use of the shared facilities. Our housing in Sisimiut are private facilities rented through AirBnb. All rules associated with the facilities must be obeyed with consideration to the neighbors.
- All team members must be mindful of the importance of safety and follow procedures that maintain a safe working environment for everyone, both while at our bases of operation and while in the field. Although most field sites will not have extreme hazards that require specialized equipment, field work has inherent dangers. Examples include fast flowing rivers, animals (dogs, foxes, muskox, reindeer), unstable sediments, cold temperatures, unstable rocks, and cliffs. Work under the buddy system, and keep an eye out for teammates.
- We will have access to vehicles in town and if you drive, you must abide by all local laws (e.g., speed limits – note that the speed limit in downtown Kangerlussuaq is 20 km/hr - no riding in the back of trucks, seat belt use required). Be aware of pedestrians, particularly small children, who often are out playing without adult supervision. You must be aware of your surroundings at all times.
- Team members over 21 may consume alcohol on the deployment. Team members who create problems while under the influence of alcohol may be dismissed from the deployment. Furnishing alcoholic beverages to any person under the age of 21 will be grounds for immediate expulsion. UF, and by extension our field sites, is a drug-free and alcohol-free workplace. Alcoholic beverages may not be stored or consumed in work areas. Work areas are defined as shops, labs, or motor vehicles of any type. Use and distribution of illegal drugs on the deployment will not be tolerated. Team members will not be penalized for violations of the alcohol or drug policy that are disclosed as part of a sexual misconduct report.

Our NSF support comes with the mandate that we comply with the NSF Office of Polar Programs Code of Conduct for work in Polar Regions. The Code can be found at https://www.nsf.gov/geo/opp/documents/policy/polar_coc.pdf. Please read the Code in its entirety before leaving. You should also become familiar with the general NSF policy towards harassment (information at https://www.nsf.gov/od/odi/harassment.jsp). Text included in Appendix A is taken verbatim from the Code and lists some, but not all, examples of conduct that violate the fundamental principles and objectives of the Code.

Similarly, as University of Florida personnel, we are obligated to abide by UF rules which prohibit discrimination against any person on the basis of race, color, national origin, religion, age, sex, gender, sexual orientation, gender expression, gender identity, gender transition status, sex- or gender-stereotyping, pregnancy, physical or mental disability, medical condition, genetic information, ancestry, marital status, citizenship, or protected veterans. These prohibitions stem from the scope and definitions outlined by the Department of Education's new Final Rule as a "Title IX violation" (see https://titleix.ufl.edu), with which all team members should become familiar. Examples of prohibited behaviors are described in Appendix A.

What to do if you see or become aware of inappropriate activities

The most effective way we can prevent harm is by looking out for each other. We are all expected to share in creating a safe environment and acting when witnessing dangerous or harmful behavior. Although bystander intervention training programs are available, training is not necessary to be an active bystander, which can be achieved with any of the following activities;

- Be aware of your surroundings and social situations.
- If a situation makes you or others uncomfortable, or it looks like someone is being targeted, recognize that it is a problem and that you can be part of the solution to help.
- Take action to diffuse the situation while staying safe; some ideas include checking in with the targeted individual, telling a senior personnel what is happening, recruiting help from friends, diffusing the situation by distracting those involved.
- If you are uncertain if there is a problem, check in with the individuals involved to see if they are okay or need help.
- Look out for your friends and colleagues, but never put yourself at risk!

Numerous activities can be used to support victims of all inappropriate activities including, but not limited to sexual misconduct, discrimination, and harassment.

- Tell them that you believe them, that you support them, that it is not their fault, that no one deserves to be targeted by such behavior.
- Provide them with the list of contact information and resources included in this document.
- Ask if they want your help in finding out what their options are.
- Ask what else you can do to help.
- Respect their decision not to talk with you if they don't want to.

In the case of sexual assault, survivors commonly do not identify their experience as rape or abuse although they may recognize harmful behavior. Over time, as they feel safer, they may try to understand the experience through talking about it. The support of a friend can be extremely beneficial in the healing process. If put in that situation, remember that the well-being of the person who was harmed must be prioritized. Reporting an incident to others without the support or knowledge of the person who was harmed could be more traumatic than helpful. If you are UF staff or faculty, under current Title IX interpretations you must report any potential incident of sexual based discrimination, harassment or misconduct. After such a report, the office will conduct outreach to the person harmed, who will make the decision about whether an investigation moves forward.

If you have been subject of any type of sexual based discrimination, harassment or misconduct, we want to know that the group as a whole supports you. You have the right to:

- Talk to anyone about your experience;
- Not talk to anyone about your experience (silence can make the healing process more difficult, and we encourage you to reach out to a trusted friend or one of the resources listed below);
- Change your mind about talking to anyone about your experience at any time;
- Report to any option listed in Appendix B;
- Bring someone with you to provide support during reporting or any resulting discussions;
- Seek reasonable accommodations to minimize the impact of the experience on the success of your work in Greenland or on the SILA or WIGF projects;
- Seek mental health and medical assistance, including medical care and a medical forensic exam.

This list is not comprehensive and you may have other right and options.

Reporting options

If you believe that you have experienced discrimination, unlawful harassment, sexual misconduct, or violence, or if you believe you have observed it among others, you have the right, and are encouraged, to disclose this behavior to anyone you chose. You also have the right to not talk with anyone about the behavior; however, that will mean that the behavior is less likely to be addressed and changed and that you are trying to handle a difficult situation alone. You also have the right to change your mind about talking to anyone at any time. Disclosure of any incident can be made through multiple channels including:

- Disclosure to a SILA or WIGF faculty member, in Greenland or at UF, but recognize that they have an obligation to report any potential incident of sexual based discrimination or misconduct to UF's Title IX Coodrinator.
- Disclosure to any member of the SILA or WIGF team,
- Disclosure to any Water Institute staff member
- Disclosure to your or any other departmental administration
- Disclosure to the UF Title IX Office (Office Accessibility and Gender Equality)

- Disclosure to local police or other relevant authority (i.e., Polar Services)
- Disclosure to Office of Victim Services (confidential resource)
- Disclosure to Office Ombuds (confidential resource for some forms of discrimination and harassment)
- Disclosure to Counseling services (confidential resource- see contact information below)

What happens after a report is made

Disclosure of an incident will result in information being provided to relevant faculty and potentially Water Institute staff, departmental administrators, and the UF Title IX office. When alerted, the Title IX office will work with the individual who was harmed to determine a course of action. When alerted about an incident, an investigator from the UF Title IX office will provide resources for support and ask if the individual wants the office to move forward with an investigation. An investigation will move forward only with approval by the individual, unless one of the following conditions is met:

- The incident was part of a larger pattern (one example is if multiple individuals reported the same person for comments that constituted sexual harassment or discrimination);
- The accused individual has a history of violence, sexual violence, arrest, or the incident was committed by multiple perpetrators;
- The incident was perpetrated with a weapon, included physical violence (such as hitting, restraint, pushing, or kicking), or the threat of violence;
- The affected individual is a minor;

The target will be included and informed of the results of their disclosure, any action that is taken, and the results of any investigation. Because we are supported by funds from the National Science Foundation (NSF), we must report information about any findings of sexual or other types of harassment and actions taken to the NSF (for details regarding the procedure, see https://www.nsf.gov/od/odi/docs/Sexual_Harassment_FAQs.pdf).

Potential Disciplinary Actions

Most issues, including harassment, discrimination, and safety, may be resolved through communication with open dialogue; team members are encouraged to report issues early before they become major. All issues, no matter how minor, require immediate action and depending on the issue may trigger long-term consequences. The actions related to the SILA and WIGF projects will be decided by faculty in consultation with other appropriate authorities; other consequences may be taken by other authorities. If faculty are unavailable or otherwise not appropriate for reporting, a report can be made to personnel at UF or elsewhere on the deployment as discussed above.

Certain major infractions will result in removal of the offender from the immediate situation, and potentially dismissal from deployment. These infractions include but are not limited to physical or verbal abuse or assault, hazing of new participants, bullying, verbal or physical intimidation, coercion, threats, sexual harassment and misconduct, or behavior that endangers the health and safety of oneself or others. Violations of gender equity policy that are reported to the Title IX office may be resolved through both Formal and Informal Resolution

Processes in which the Title IX Coordinator acts as a third neutral party to mediate the communication throughout the process. Other actions handled internally could include rearrangement of living spaces, reallocating personnel among field teams, restricted activities on the deployment, or dismissal from the deployment. Repeated infractions of SILA policies may result in expulsion from deployment if these behaviors are not corrected after being brought to the perpetrator's attention. In general, the cost incurred for travel changes will be paid by the offending team member. A person may receive formal warnings – typically via email – for minor infractions before being dismissed; all faculty may issue warnings, typically after consulting with other PIs. Anyone who brings a complaint will be informed of the remediation action as soon as possible, generally by the person who received the complaint. A person who feels they have been dismissed unfairly may contact appropriate University of Florida officials (Water Institute staff, department chairs, or the Title IX office; see Appendix B). Such grievances will be addressed in a timely manner. Retaliatory conduct is prohibited and may result in more severe discipline than the underlying alleged misconduct.

Resources

Development of this policy drew heavily on the Toolik Field Station policy (https://toolik.alaska.edu/user_guide/Sexual_Misconduct_Policy_20180329.pdf)

UF Sexual harassment Policy
https://hr.ufl.edu/forms-policies/policies-managers/sexual-harassment/

UF Gender Equity Policy
https://titleix.ufl.edu/about/laws-policies/

UF Title IX: Get Help
https://titleix.ufl.edu/get-help/

Prohibited conduct

The following activities are prohibited by the NSF Code of Conduct (https://www.nsf.gov/od/odi/harassment.jsp) and prohibitions specific to activities in the Arctic are included in NSF Office of Polar Programs Code of Conduct (https://www.nsf.gov/geo/opp/documents/policy/polar_coc.pdf).

Physical or verbal abuse of any person, including, but not limited to, harassment, stalking, bullying, or hazing of any kind, whether the behavior is carried out verbally, physically, electronically, or in written form.

- Conduct that is offensive, indecent, obscene, or disorderly.
- Possession, use, sale, manufacture, transfer, trafficking in, or being under the influence of illegal drugs, including marijuana, and abuse of legal drugs.
- Violation of applicable policies, including, but not limited, to the NSF Safety and Occupational Health Policy; the U.S. Antarctic Program (USAP) Alcohol Policy, and the USAP Lodging Policy.
- Violation of the USAP Information Technology Enterprise Rules of Behavior.
- Violation of the Principles for the Conduct of Research in the Arctic.
- Solicitation of gifts. In general, Federal ethics laws prohibit the solicitation of gifts (for example, any gratuity, favor, food, or entertainment). See e.g. 5 C.F.R. 2635.202(c)(2). Personnel may not solicit gifts.
- Endorsements, expressed or implied, of products, services, or enterprises. Such endorsements are prohibited. See e.g. 5 C.F.R. § 2635.702. USAP and/or NSF facilities, property, logos, or insignias may not be used for endorsement purposes.

In addition, no form of physical or verbal abuse or assault, intimidation, coercion, threats, gender, race-based, or sexual harassment, sexual misconduct, or behavior that endangers the health and safety of oneself or others is allowed. Some conduct is specifically prohibited under Title IX (see https://titleix.ufl.edu), including:

– A wide range of offensive behaviors (verbal or non-verbal actions of aggression, intimidation, and hostility) based on gender, sex, sexual orientation, and gender identity or gender expression. These behaviors do not need to be of a sexual nature but must place the receiving person in reasonable fear of physical harm, or objectively disrupt or interfere with their employment, education, or other activities related to their role on the University of Florida campus.

– Violence committed by a person who is or has been in a social relationship of a romantic or intimate nature with the victim. The existence of such a relationship shall be determined based on the reporting party's statement and with consideration of the length of the relationship, the type of relationship, and the frequency of interaction between the persons involved in the relationship.

– Unwelcome sexual advances, requests for sexual favors, or other verbal or physical conduct of a sexual nature directed at a person that places another person in reasonable fear of physical harm, or objectively disrupts employment, education, research, living, or other activities.

– Multiple, unwelcome acts directed at a specific person that (by a reasonable person's standard) cause that individual to fear for his/her (or others') safety and cause him/her considerable emotional distress. Specific types of stalking include (but are not limited to): monitoring, following, surveilling, harassing, pursuing, threatening, repeatedly contacting a person without consent, interfering, or damaging personal property.

– Any adverse action or behavior (or attempted adverse action or behavior) imposed against an individual as a result of their participation/ involvement in an investigation. To be retaliatory, the action or behavior must have a materially adverse effect on an individual's employment, academics, living environment, or mental well-being. Allegations of retaliation should be immediately reported to the Title IX Coordinator; any individual responsible for retaliation will be subject to disciplinary action.

Minor infractions

Other disruptive conduct, which may not rise to the level of the conduct listed above, may nonetheless interfere with the ability of other team members to do their jobs, and may erode team esprit de corps. As described above, repeated infractions of deployment rules may also result in more severe disciplinary action if these behaviors are not corrected after being brought to the person's attention. Formal warning(s) for "minor" infractions may be issued, and the faculty are responsible for keeping a record of who has been warned. Examples of minor infractions include:

- Disregard for quiet hours i.e.: loud parties in residential areas
- Smoking in any building
- Abuse of alcohol
- Dangerous driving
- Disregard for the personal property of others
- Loud and obnoxious behavior

Contact information:

The following resources are required to report any alleged or disclosed violations of the universities' sexual-based misconduct & relationship violence policy to the Office for Accessibility and Gender Equity. Other resources can be found here: https://titleix.ufl.edu/get-help/campus-resources/

General contacts

UF Title IX Office
https://titleix.ufl.edu/

Russ Frohman JD., Ed.D., coordinator
Assistant Vice President for Accessibility and Gender Equity
ADA Coordinator and Title IX Coordinator
1908 Stadium Road | 427 Yon Hall
Gainesville, FL 32611
Phone: (352) 275-1242
Fax: (352) 392-5268
inform@titleix.ufl.edu

Jessica C. Baker, GCDF, CCSP, Engagement and Prevention Coordinator
Phone: (352) 294-7015
Fax: (352) 392-5268
jcjess@ufl.edu

UF Water Institute
Dr. Wendy Graham, Director
Phone: (352) 294-7741
wgraham@ufl.edu

Dr. Paloma Carton de Grammont, Research Coordinator III
Phone: (352) 294-7744
Cellphone: (517) 802 8490
palomacgl@ufl.edu

Department of Geological Sciences
Dr. David Foster, Chairman
Ms. Kristin Nichola, Office Manager
Phone (352)-392-2231
knichola@ufl.edu

Department of Biology

Marta Wayne, Professor & Department Chair
Phone: (352) 392-9925
Email: mlwayne@ufl.edu

Department of Microbiology and Cell Science
Eric Triplett, Professor and Chair
Phone: 352-392-5430
Email: ewt@ufl.edu

School of Forest, Fisheries and Geomatic Sciences
Terrell T. "Red" Baker, Director and Professor
Phone" 352-846-0850
Email: ttredbaker@ufl.edu

Department of Soil and Water Sciences
Matt Whiles, Professor and Chair
Phone: (352) 294-3151
Email: mwhiles@ufl.edu

School of Journalism and Communications
Ted Spiker, Professor and Chair
Phone: 352-392-0500
Email: tspiker@jou.ufl.edu

Confidential Resources

These contacts will keep your identity confidential.

The University of Florida Police Department Office of Victim Services
1515 Museum Road
P.O. Box 112150
Gainesville, FL 32611-2150
(352) 392-5648 (Monday – Friday, 8:00 a.m. – 5:00 p.m.)
(352) 392-1111 (after business hours and on weekends).
If you would like to speak to our Victim Advocates, please contact ovs@mail.ufl.edu.
https://police.ufl.edu/about/divisions/office-of-victim-services/

Counseling & Wellness Center
3190 Radio Road,
PO Box 112662
Gainesville, FL 32611-2662
(352)-392-1575

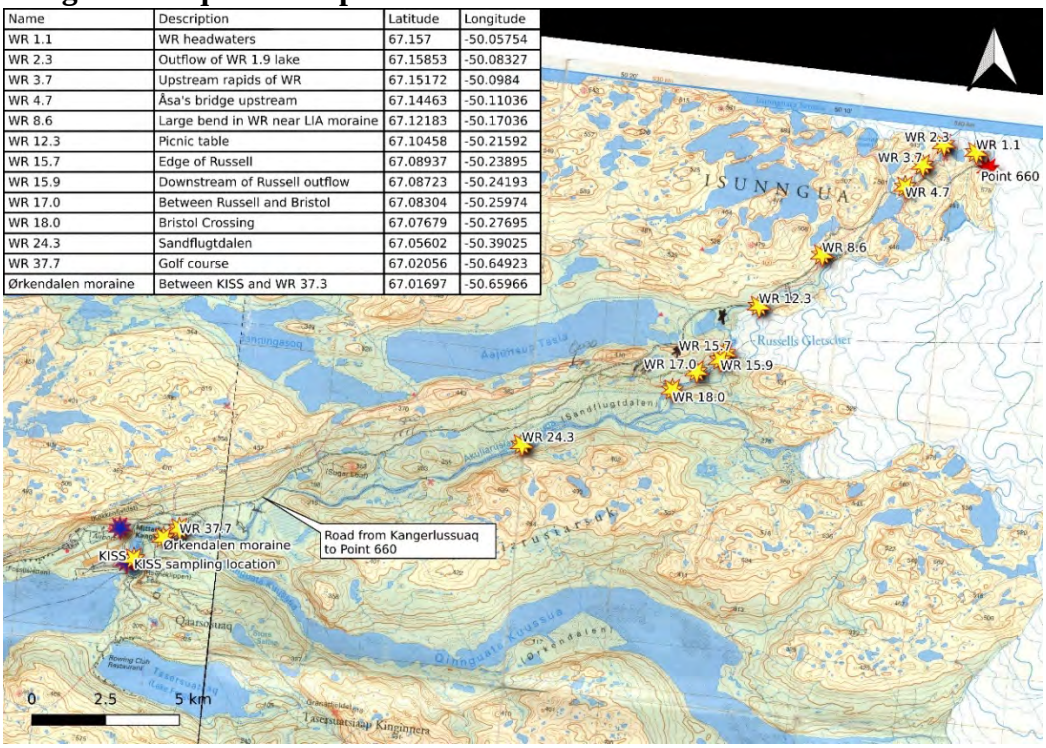
http://www.counseling.ufl.edu/cwc/

Crisis and Emergency Resource Center (CERC)
(352)-392-1575
http://www.counseling.ufl.edu/services/crisis/

Location Maps

Kangerlussuaq Area Maps

Name	Description	Latitude	Longitude
WR 1.1	WR headwaters	67.157	-50.05754
WR 2.3	Outflow of WR 1.9 lake	67.15853	-50.08327
WR 3.7	Upstream rapids of WR	67.15172	-50.0984
WR 4.7	Åsa's bridge upstream	67.14463	-50.11036
WR 8.6	Large bend in WR near LIA moraine	67.12183	-50.17036
WR 12.3	Picnic table	67.10458	-50.21592
WR 15.7	Edge of Russell	67.08937	-50.23895
WR 15.9	Downstream of Russell outflow	67.08723	-50.24193
WR 17.0	Between Russell and Bristol	67.08304	-50.25974
WR 18.0	Bristol Crossing	67.07679	-50.27695
WR 24.3	Sandflugtdalen	67.05602	-50.39025
WR 37.7	Golf course	67.02056	-50.64923
Ørkendalen moraine	Between KISS and WR 37.3	67.01697	-50.65966



Name	Description	Latitude	Longitude
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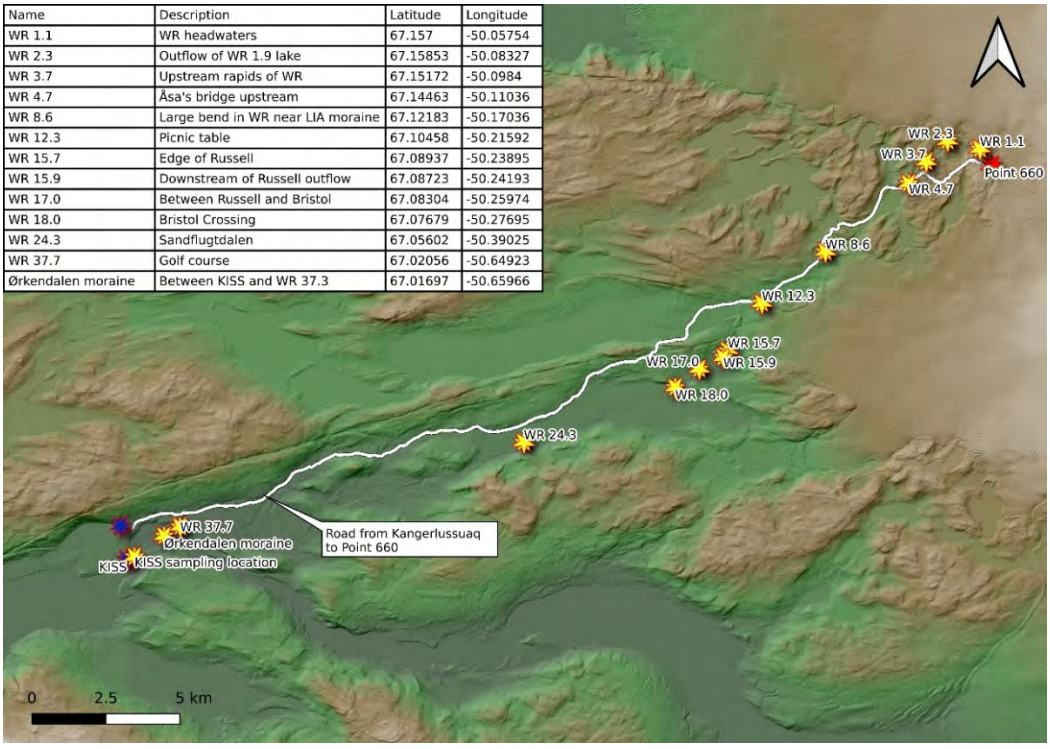
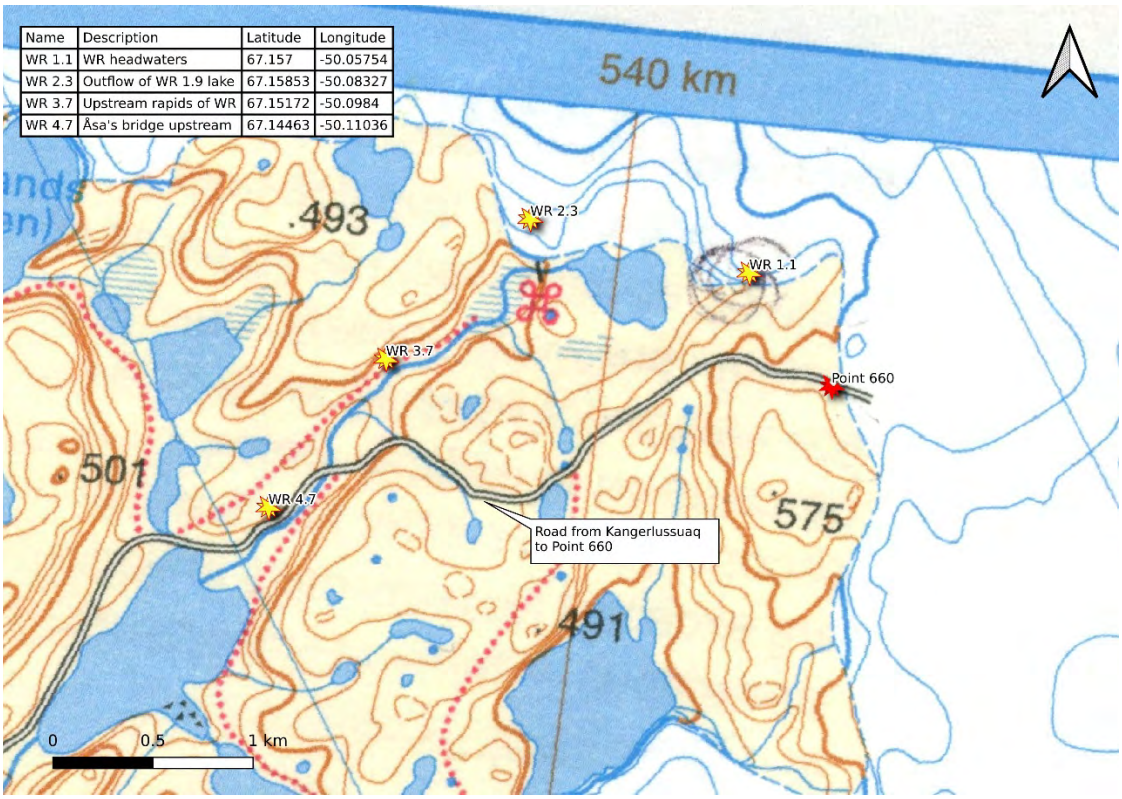
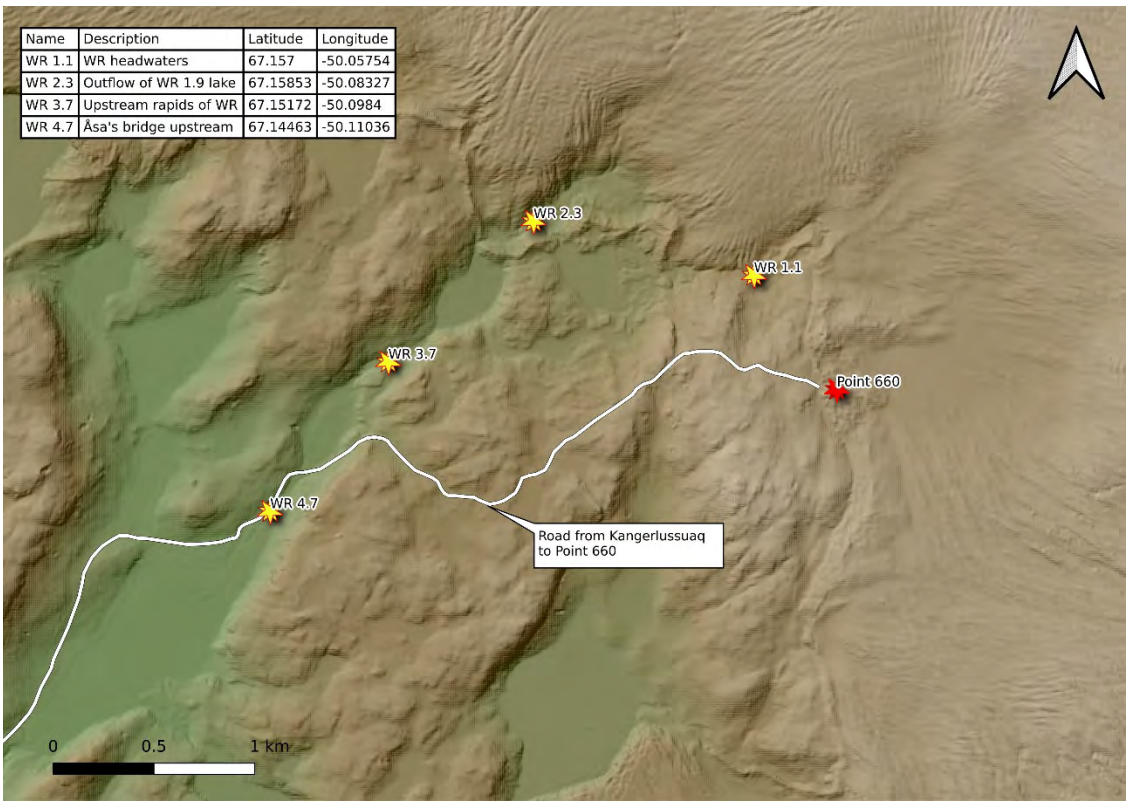
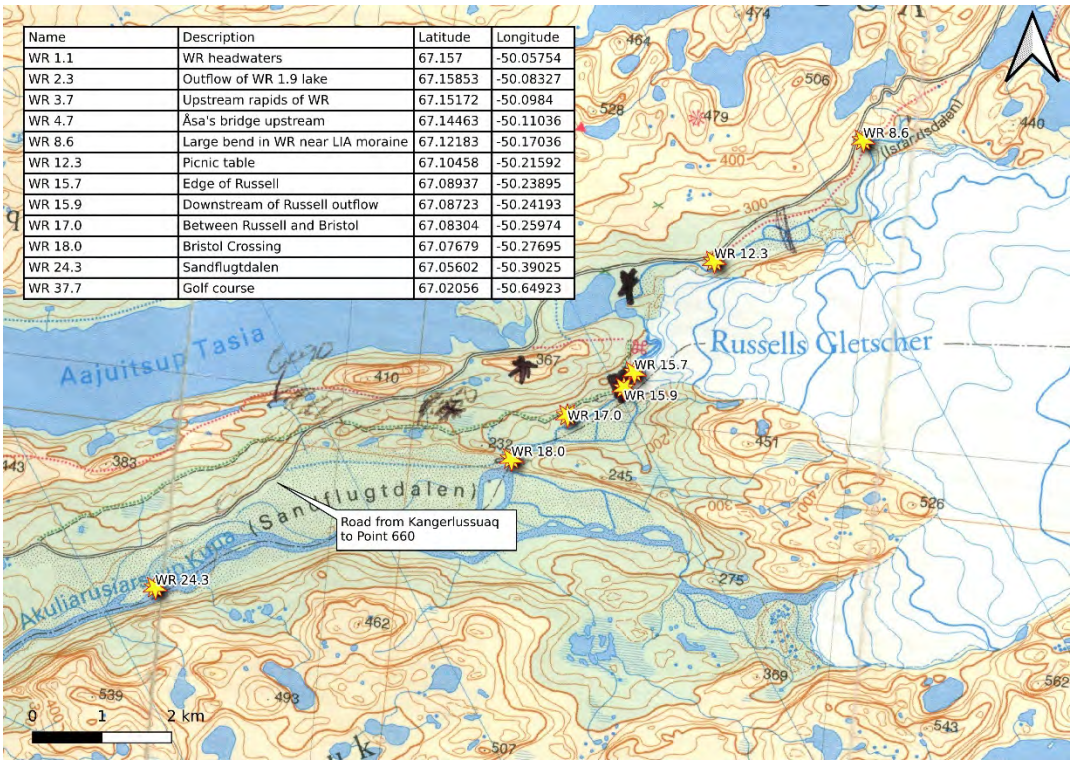


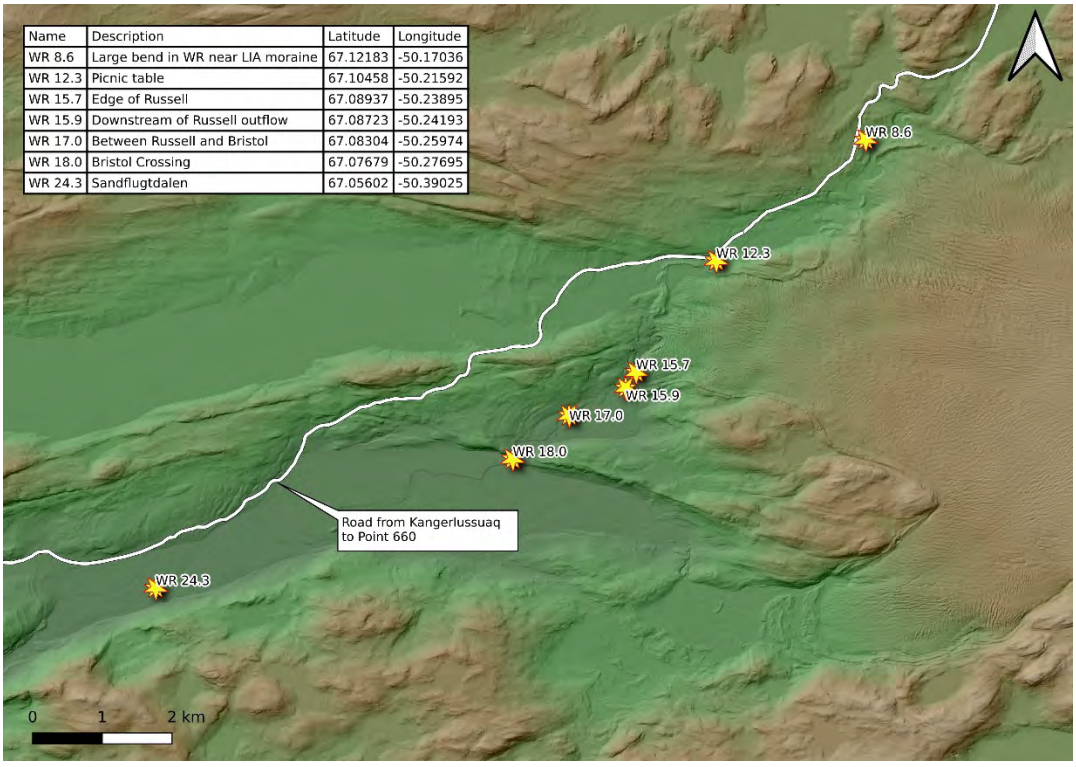
Watson River overview



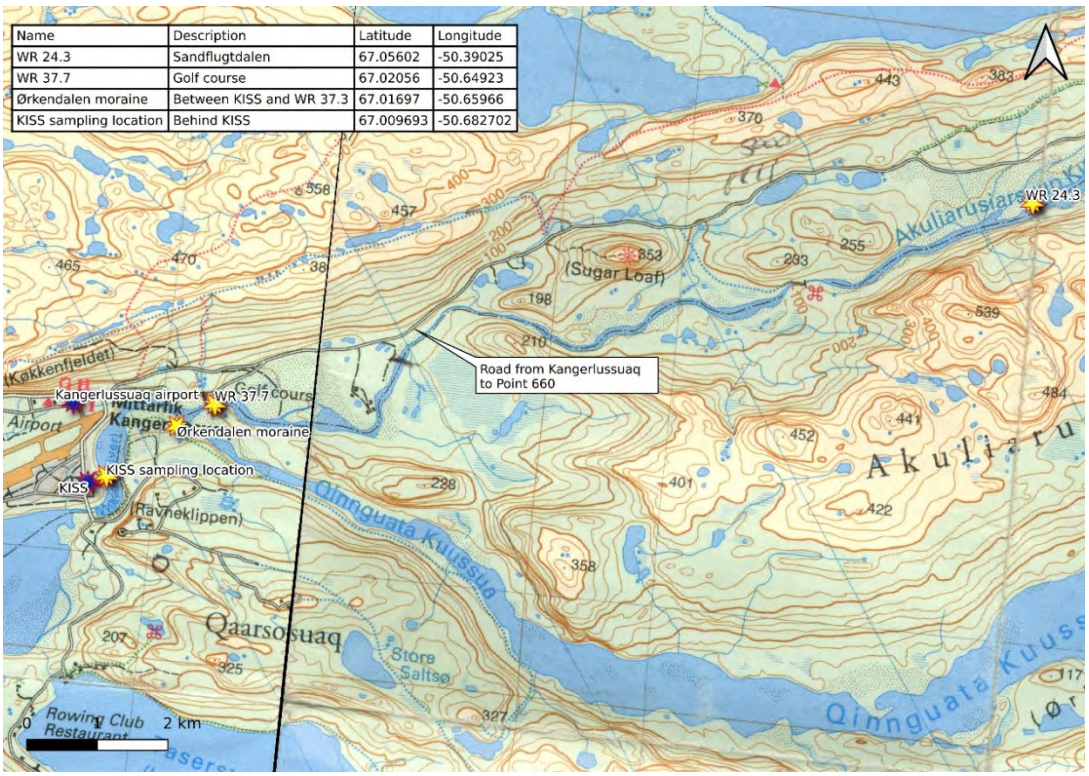


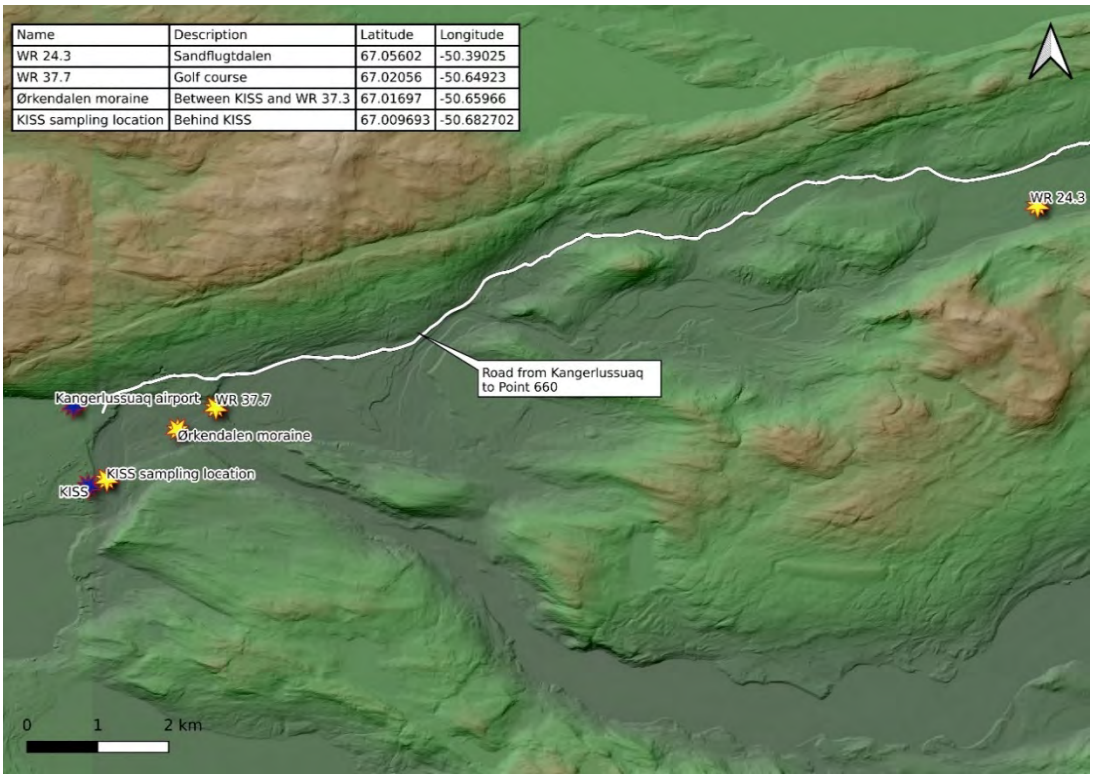
Watson River near ice



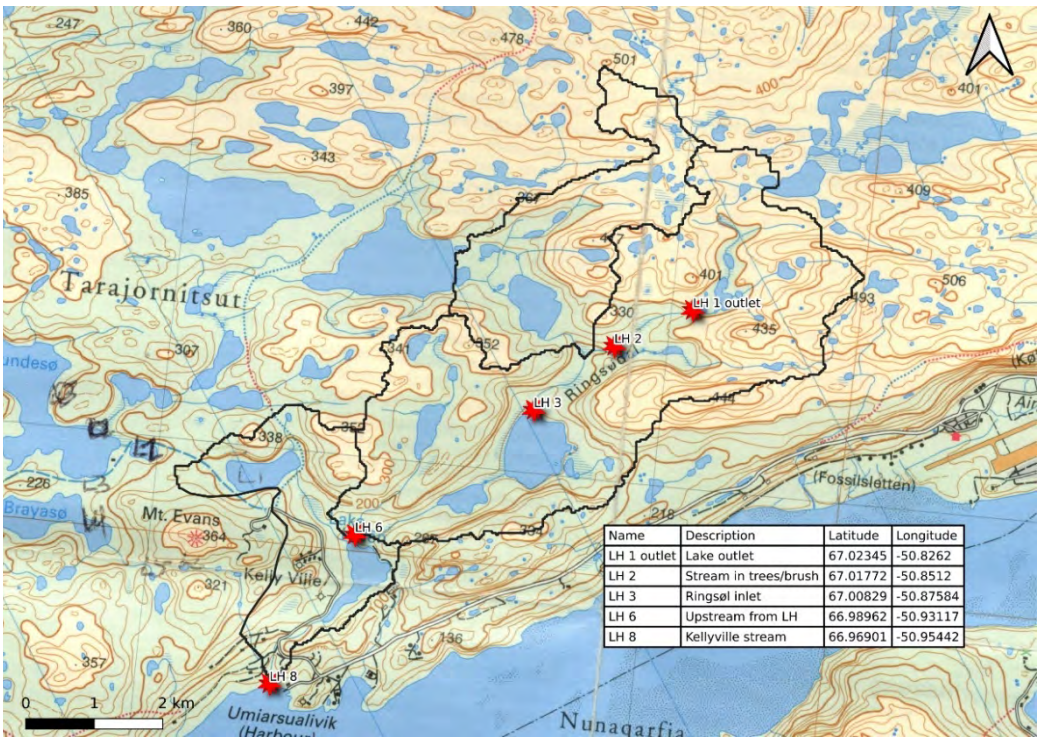


Watson river upper mid section

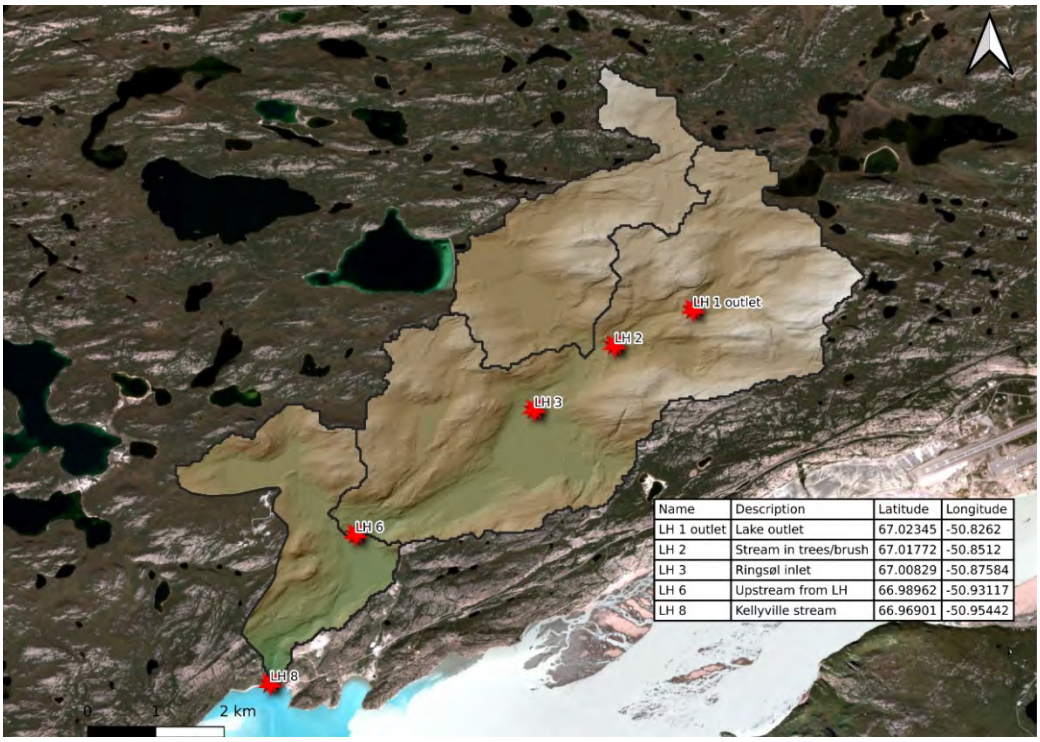




Watson river near KISS



Name	Description	Latitude	Longitude
LH 1 outlet	Lake outlet	67.02345	-50.8262
LH 2	Stream in trees/brush	67.01772	-50.8512
LH 3	Ringsøl inlet	67.00829	-50.87584
LH 6	Upstream from LH	66.98962	-50.93117
LH 8	Kellyville stream	66.96901	-50.95442



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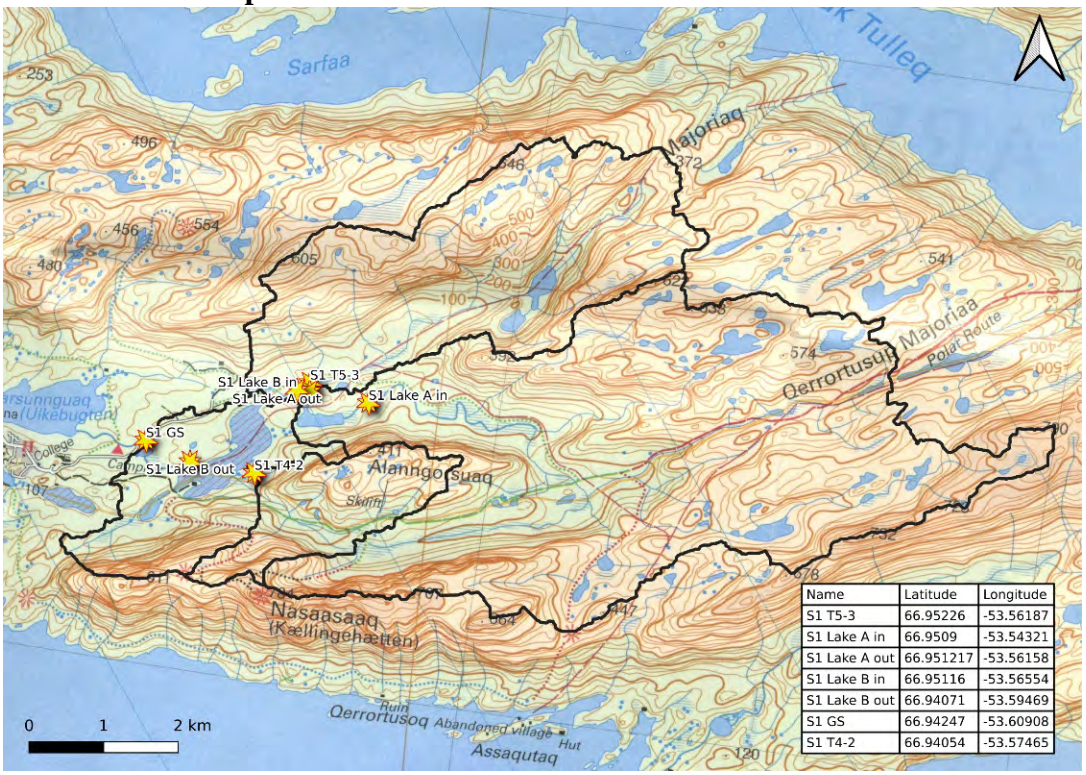
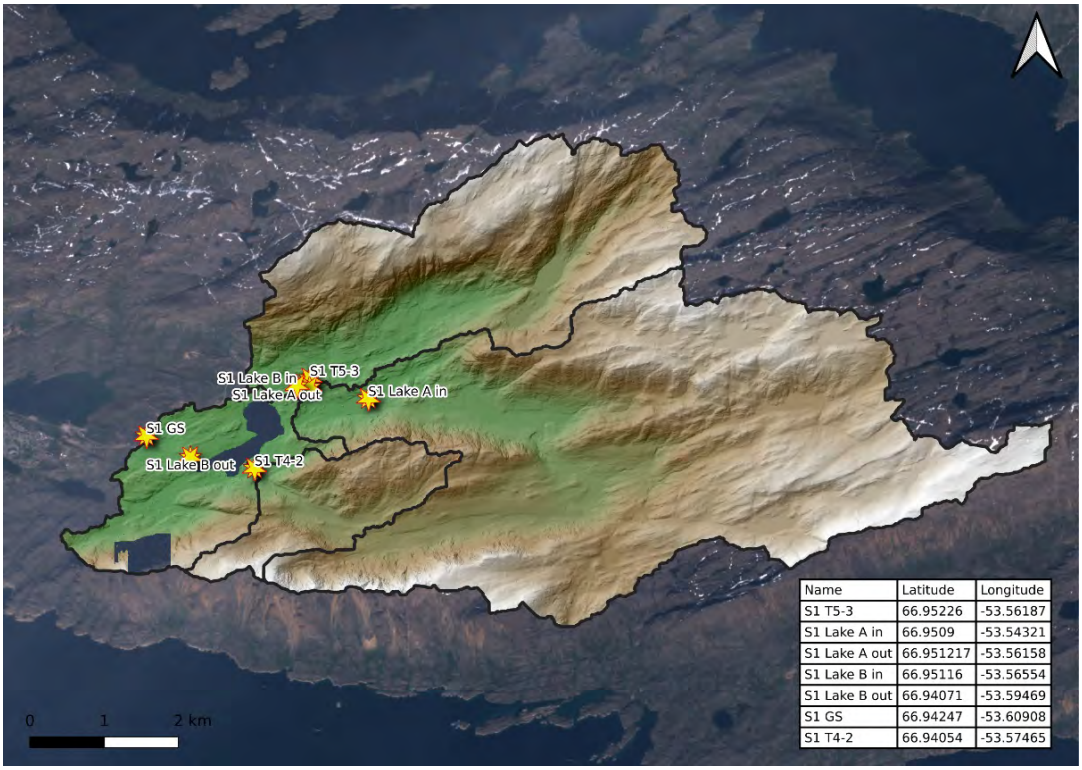
LH 6	Upstream from LH	66.98962	-50.93117
LH 8	Kellyville stream	66.96901	-50.95442

Lake Helen

Sisimiut Area Maps

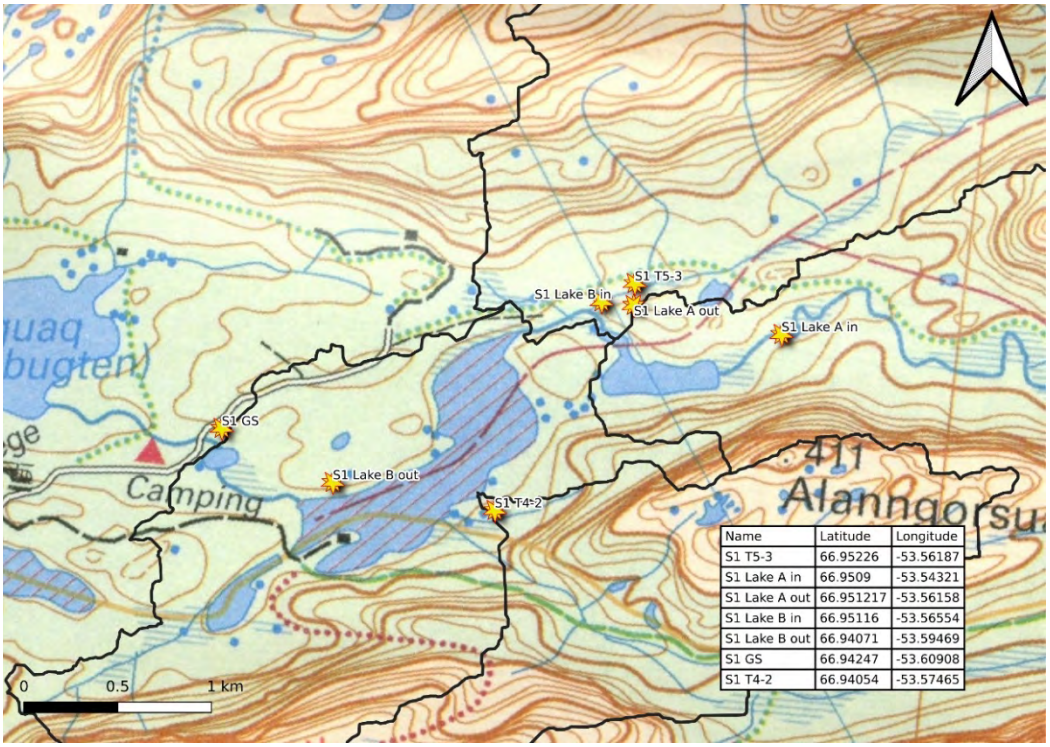


Name	Latitude	Longitude
S1 T5-3	66.95226	-53.56187
S1 Lake A in	66.9509	-53.54321
S1 Lake A out	66.951217	-53.56158
S1 Lake B in	66.95116	-53.56554
S1 Lake B out	66.94071	-53.59469
S1 GS	66.94247	-53.60908
S1 T4-2	66.94054	-53.57465



Name	Latitude	Longitude
S1 T5-3	66.95226	-53.56187
S1 Lake A in	66.9509	-53.54321
S1 Lake A out	66.951217	-53.56158
S1 Lake B in	66.95116	-53.56554
S1 Lake B out	66.94071	-53.59469
S1 GS	66.94247	-53.60908
S1 T4-2	66.94054	-53.57465

S1 all



Name	Latitude	Longitude
S1 T5-3	66.95226	-53.56187
S1 Lake A in	66.9509	-53.54321
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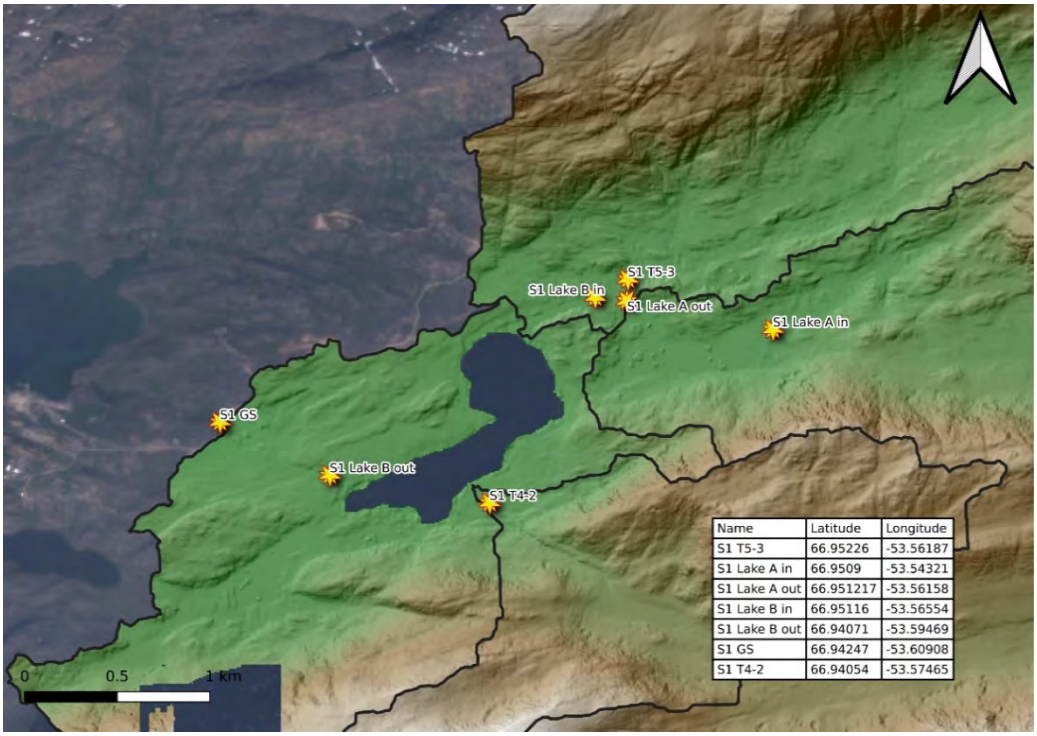

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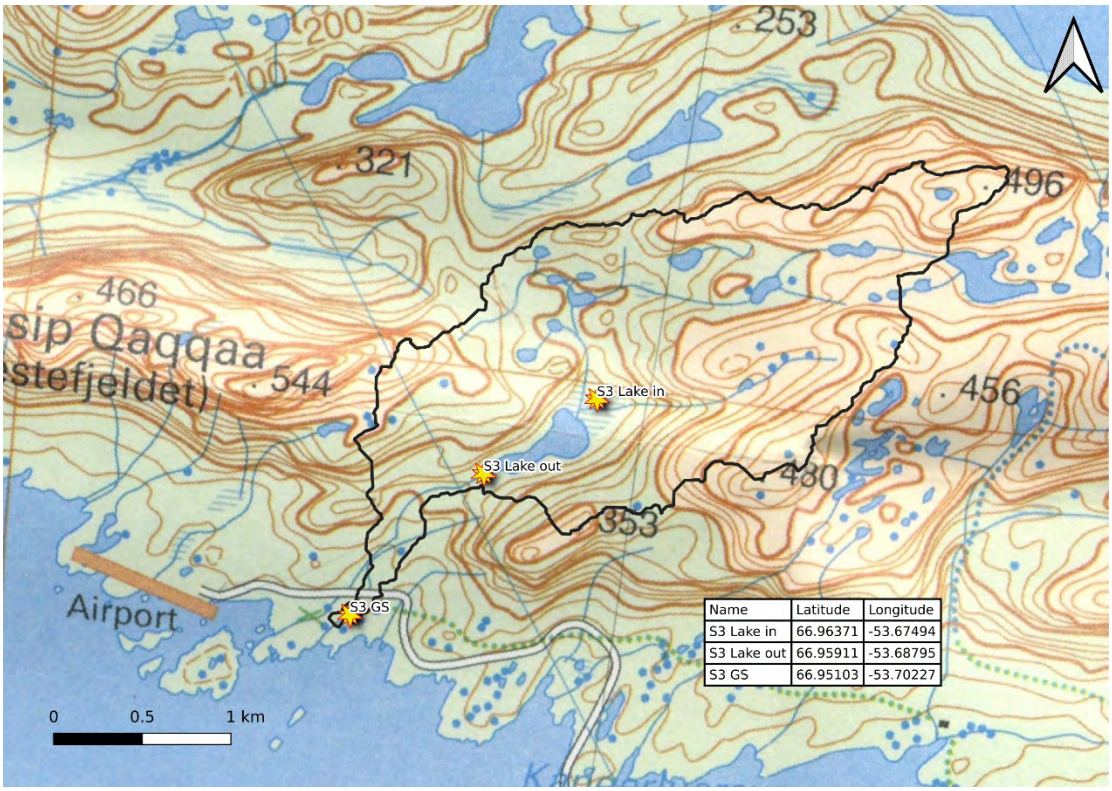
S1 T5-3	66.95226	-53.56187
S1 Lake A in	66.9509	-53.54321
S1 Lake A out	66.951217	-53.56158
S1 Lake B in	66.95116	-53.56554
S1 Lake B out	66.94071	-53.59469
S1 GS	66.94247	-53.60908
S1 T4-2	66.94054	-53.57465

S1 – sample sites only


200
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S3 Lake in
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Airport
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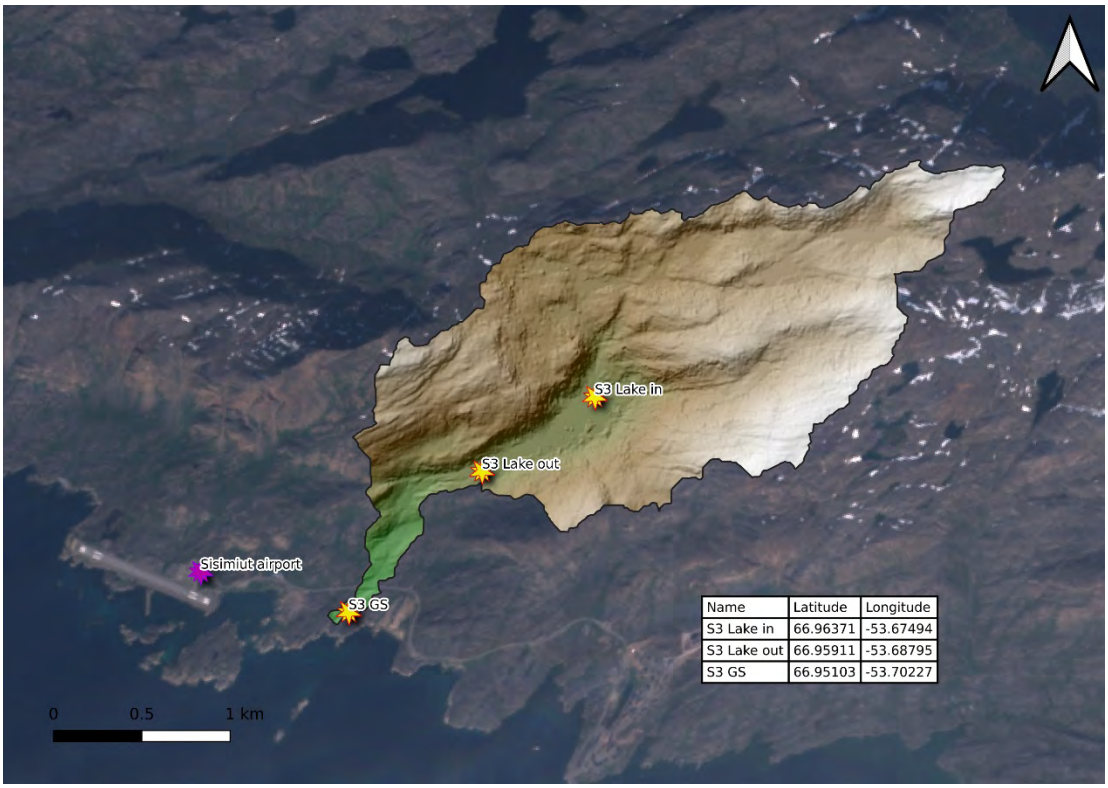
1 km
Name | Latitude | Longitude
S3 Lake in | 66.96371 | -53.67494
S3 Lake out | 66.95911 | -53.68795
S3 GS | 66.95103 | -53.70227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Name | Latitude | Longitude
S3 Lake in | 66.96371 | -53.67494
S3 Lake out | 66.95911 | -53.68795
S3 GS | 66.95103 | -53.70227

S3 - all