Biology 345
Ecology

Field Ecology Workshop Manual 2025

Athabasca University
Faculty of Science and Technology

Course Team

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Materials to bring to the 4-day lab:

Notebook (!), pencils, appropriate clothing, change of clothes, hat and footwear for outdoor activities (hiking boots or rubber boots), (be prepared for rain, mud, high solar radiation, mosquitoes and other biting insects), laptop, textbook (important for students who have not yet completed most parts of the course).

We will work both in the field and in the lab (lab coats are provided).

Be prepared to work outdoors. If you have any physical or medical conditions that require special considerations, the lab instructors must be informed well ahead of time.

Before attending the lab, join the Ecology Landing group: Go to *landing.athabascau.ca* and log in with your AU student ID. In the search field, look for *BIOL 345: ECOLOGY 2025*. Click on the link and *join the group*. All lab results (but not the Lab Report) must be submitted on this site.

Please note that in case of unforeseen events, the lab schedule might be changed on short notice.

Day 1

Our first day will be dedicated to limnology and we will visit various ponds, rivers and streams in the surroundings of the Athabasca University campus. Bring water, change of clothes and footwear, towel, hat, insect repellent, rain gear, sunscreen and footwear that can get wet.
Note: For workshops held at Athabasca University, please arrive by 8:50 AM at the Science Lab (watch for Science Lab signs in the AU Main Building). We will not wait for latecomers. Students can park in any parking space that is not restricted as a loading zone, handicapped parking, etc.

Schedule

Time	Activity
9:00 AM (sharp)	Welcome, introduction, safety briefing, photo release forms, handout of lab manual, student groups to be assigned.
Morning	Athabasca River, Tawatinaw River, Muskeg Creek, various ponds: Aquatic project sample and data collection (data, water sampling, aquatic invertebrate collecting, and aquatic microbe collection).
Noon	Lunch break
Afternoon	Further field sites, collecting and compiling limnological data
5:00 PM (appr.)	End of day

Day 2

On the second day, we will visit two adjacent terrestrial habitats and characterize the invertebrates, vertebrates, and vascular plants in both.

Schedule

Time	Activity
9:00	In Lab: review of Day 1, data entry. Terrestrial habitats: Invertebrate project, vascular plant project (traps, transect and quadrants).
Noon	Flexible lunch break
Afternoon	
	In Lab: identification and weighing of plant samples; identification and weighing of invertebrate samples, review photos from game cameras. Data entry.
5:00 PM	End of Day 2

Day 3

On the third day, we will visit two adjacent terrestrial habitats and characterize the microclimate and soil in both.

Schedule

Time	Activity
Morning	In Lab: preview of day's activities
	Terrestrial habitats: soil sampling and physical factors.
12:00	Lunch break
Afternoon	
	In Lab: soil testing. Evaluation of Coliscan results from Day 1 (Group 3). Data entry.
5:00 PM	End of Day 3

Day 4

In the morning, we will explore a forest habitat. In the afternoon the groups will discuss the preliminary results of their aquatic research projects. We will wrap up with a review of the 4 days.

Schedule

Time	Activity
9:00	Forest measurement exercise. Data entry.
Noon	Lunch break
Afternoon	Discussion of results
	Lab evaluation by students, Guidelines for Lab reports
5:00 PM	End of Day 4

Day 1: LIMNOLOGY

Introduction

On the first day we will visit various water bodies around Athabasca (Athabasca River, Tawatinaw River, Muskeg Creek, AU Observatory Pond, AU campus pond, etc. We may add other locations if required). Be prepared for some walking to our field sites. Four groups of students will be assigned the following projects:

Group 1. Physical factors
Group 2. Water chemistry
Group 3. Bacteriology of water
Group 4. Aquatic organisms

Required reading

Elements of Ecology Chapter 3, Chapter 21 sect 8, 10, 11, Chapter 24 (excluding marine environment)

Additional resources

Mitchell, P., and E. E. Prepas. (Eds.) 1990. Atlas of Alberta lakes. Edmonton, Alberta: University of Alberta Press

Background

Dissolved oxygen (DO)

Most macroscopic aquatic organisms are aerobic and rely on the oxygen dissolved in water to respire. Oxygen diffuses from the atmosphere into surface water at a rate dependent on the solubility of oxygen in water and the difference in oxygen concentration between the surface water and the atmosphere. The solubility of oxygen into water is dependent on temperature (oxygen is more able to diffuse into colder water). Oxygen can also be introduced to a lake through photosynthesizing plants, cyanobacteria and algae. Oxygen is consumed by the respiration of living organisms (fish, plants, algae, micro and macroinvertebrates) and by decomposition (bacteria, fungi) of organic materials in the lake sediments. In stratified lakes, the oxygen that diffuses from the atmosphere may not reach the hypolimnion resulting in anaerobic conditions.

The Environmental Quality Guidelines for Alberta Surface Waters (Alberta Government, 2014) indicates that to sustain aerobic organisms in the epilimnion, DO should never drop below 5 mg/L and should not drop below 6.5 mg/L over more than seven days. The guidelines also indicate that the concentration of DO should be higher (above 9.5 mg/L) when the early stages of life (e.g. fish larvae) are developing. In lotic water bodies oxygen (and CO_2) dissolve easier, compared to lentic water bodies.

pH

pH is a measure of the acidity or alkalinity of water, measured by the concentration of hydrogen ions. pH ranges from 0 to 14 and is the negative logarithm of the hydrogen ion concentration. The Environmental Quality Guidelines for Alberta Surface Waters suggest that the pH of 6.5 to 9 is desirable in freshwater. pH influences the solubility of nutrients such as carbon, nitrogen, and phosphorus, as well as the biological availability (the amount that organisms can use them). pH also influences solubility of toxic heavy metals such as aluminum. Read section 3.7 in the textbook for more information on pH and carbon dioxide–carbonic acid–bicarbonate buffering system.

Benthos and plankton

Zoobenthos and zooplankton are small heterotrophic protists and animals such as insects, crustaceans, molluscs, rotifers and worms that live in water. Zoobenthos are organisms that live in the bottom sediment. Some organisms will spend only part of their lifecycle in the sediment, and others will spend their entire life cycle in the sediment. Zoobenthos can range in size from less than 200 µm (microbenthos) to more than 2 mm (macrobenthos). Zooplankton are free- floating, microscopic (typically) heterotrophic organisms. They feed on phytoplankton, bacterioplankton, other zooplankton, and detritus. They are primary consumers and act as a crucial food source for higher trophic levels including fish. In Alberta lakes, zooplankton includes culicids (mosquito larvae), rotifers, copepods, and cladocerans (water fleas). Zooplankton can range from 0.2 µm to more than 10 cm. Phytoplankton are autotrophic photosynthetic organisms that include eukaryotic protists, eubacteria and archaebacteria. Phytoplankton rely on light and nutrient availability and are important primary producers in aquatic ecosystems. In Alberta lakes, algal blooms are often a concern due to the toxins that some groups produce.

Bacterioplankton

Aquatic bacteria are found floating in the water (bacterioplankton), associated with decomposing material in the lake substrate, or in biofilms coating rocks and sand. They can contribute a considerable amount of

biomass to a lake system. Bacteria have a wide range of nutritional requirements and ecological roles. Through decomposition, heterotrophic bacteria play an important role in recycling nutrients from nonliving organic matter (detritus). They free nutrients for use by primary producers and increase the nutritional value of detritus for scavenging organisms (Smith, 2016). Autotrophic bacteria are able to use sunlight and CO2 to synthesize carbohydrates. Cyanobacteria ("blue-green algae") are primary producers that are often prevalent in seasonal blooms. Cyanobacteria are bacteria that are unrelated to algae but share the ability to photosynthesize (RAMP, "Aquatic Organisms: Microorganisms", 2017). In the plankton environment, bacteria are consumed by protists such as flagellates and ciliates, and in the benthos by metazoan animals (rotifers and cladocerans) and amoeba-like protists. Viruses also are estimated to consume between 10% to 30% of bacterial communities every day (Smith, 2016).

Project 1: Physical Factors

Group 1 will measure and record all available physical (abiotic) factors at the various water bodies.

Materials to take outdoors: Secchi Disk and attached rope (for measuring depth), 3 thermometers, 2 GPS units (check batteries), extension pole, camera (cell phone OK) notebooks, pens, pencils, labeled bucket to hold materials, some plastic bags (rain protection), fluxmeter, clipboard.

Checklist of parameters to record in the field:

- GPS location, general description of location
- Description of water body (lotic/lentic)
- Date and time
- Weather
- Temperature at depth x
- Flow speed (if applicable)
- Notes
- Photos of water bodies, surroundings, measurement processes, etc.
- Smell, colour and turbidity (Secchi disk)

Safety considerations

Avoid taking samples from slippery slopes or stones. Test first if measurement and sampling can be done safely. Always work with at least one partner.

Methods

GPS: Switch on the GPS unit and give ample time for stabilization as the instrument picks up various satellites. Results must be presented as decimals (e.g. N 60.5674$^{0;}$ E 35.5677^{0}).

Fluxmeter for the measurement of flow speed in rivers. The operation of the fluxmeter will be explained in the field. Use the maximum extension (length) of the unit. Select sample locations that are free from obstructions. Take notes of the specifics of the stream (bends, depth at sample location, etc). Take a minimum of 2 readings per water body and note the depth of the sonde for each reading. Readings must be recorded in meters/second.

Thermometer: allow for some lag time for the thermometer to display a stable reading. Whenever feasible, take readings at various depths.

Secchi Disk: Lower the Secchi Disk until you can no longer / barely see its pattern. Record the length of the attached rope in cm. This is a measurement of water turbidity (in cm).

Camera: Document your measurements and the field sites.

Sample notebook record:
Amur River, western China, Jilin Province, 31-05-2000, Time 10:52, N 60.5876^0 E 35.5678^0
Lotic, major river, app. 300 m wide (estimation), heavy rainfall, air temperature 19.8^0 C.
Temperature at surface: 9.1^0 C
Temperature in 10 cm depth: 9.0^0 C
Temperature in 20 cm depth: 8.9^0 C
Temperature in 30 cm depth: 9.0^0 C
Flow speed at surface: 6.5 m/sec
Flow speed at 10 cm: 0.5 m/sec
Flow speed at 20 cm: 0.0 m/sec
Measurement was taken at the south bank of the river (straight flow, no river bend)
Record was taken by Francois Guirre, student at Beijing University.
Instruments: digital Hansen 3.45 thermometer, Fisher Fluxmeter 45.1, GPS: Garmin 550
Photos of the research sites are deposited at http://landing.athabascau.ca

Collection of data

Group 1 will complete and submit copies of Table 1 separately for each water body. In addition, the field sites must be documented with photographs.

Table 1. Physical factors (aquatic)

- Use one sheet per location

Location*	
Date:	
Time:	
Weather, Air temperature	
Notes	

		Site 1	Site 2
	GPS location		
	Secchi disk		
Surface	Temp (°C)		
	Flow speed		
Depth 1	Temp (°C)		
	Flow speed		
Depth 2	Temp (°C)		
	Flow speed		
Depth 3	Temp (°C)		
	Flow speed		

*Athabasca River, Tawatinaw River, Muskeg Creek (Middle Bridge), Observatory Pond, ARC Pond

Project 2: Water Chemistry

Group 2 will measure and record some water chemistry data in the field. Others will be taken in the lab. Note: the dissolved oxygen content and the dissolved CO_2 content are very sensitive to environmental conditions (temperature, water movement, etc.) and are therefore taken in the field (and subsequently in the lab as a control).

Materials to take outdoors: pH test strips (different types), multi parameter test strips for pH, alkalinity, hardness, etc., dissolved oxygen test kit (1 unit), (colourimetric, CHEMMETS), dissolved CO_2 test kit (1 unit), phosphorus test kit (aquatic, 1 unit), potassium test kit (aquatic, 1 unit; NOTE: the sodium hydroxide in the kit must be made fresh before the lab), notebooks and pencils, labeled bucket to hold materials, 20 disposable plastic pipettes, 5 plastic beakers (appr. 250 ml), 5 empty petri dishes, 15 water sample bottles (clean), 3 bottles of distilled water, 3 marker pens, gloves (10 pairs), small plastic bags for waste, camera (cell phone OK).

Materials to be used in the lab (depending on time available and weather conditions, we may take some of these measurements in the field): pH test strips, multi parameter test strips (pH, alkalinity, hardness, etc.), dissolved oxygen test kit, (colourimetric, CHEMMETS), dissolved CO_2 test kit (1 unit), phosphorus test kit (aquatic, 1 unit), pH-meter (4 units), buffer solutions for calibration, potassium test kit (aquatic, 1 unit), 20 disposable plastic pipettes, 3 bottles of distilled water, 5 plastic beakers (appr. 250 ml), camera (cell phone OK), clipboard.

Parameters to record in the field:

- dissolved oxygen
- dissolved CO_2
- pH (various test strips)
- multi parameter test strips (pH, alkalinity, hardness, etc.)

Three water samples per location will be taken back to the lab for further measurements.

Parameters to record in the lab (or in the field, depending on available time and weather conditions):

- dissolved oxygen (repeat measurement for comparison)
- dissolved CO_2 (repeat measurement for comparison)
- potassium content
- phosphorus content
- pH using a pH-meter

- pH using various test strips (compare with pH-meter results)

Safety considerations

Field: Avoid taking samples from slippery slopes or stones. Test first if measurement and sampling can be done safely. Use gloves. Always work with at least one partner. Lab: Read MSDS and individual safety precautions for the tests.

Methods

Water Sampling

Use a collection bottle to take a water sample from the surface of the water body. Whenever feasible, use the extension pole. Use care to not create any turbulence, especially in lentic, sediment rich waters.

Water Testing in the field

Dissolved Oxygen O_2 Test Kit (Indigo Carmine Colourimetric Method)

Contents: Sample cup, 25 ml, plastic, ampoules of indigo carmine, Oxygen Comparator with several blue coloured vials, 1-12 ppm.

1. Fill the sample cup to the 25 ml mark with your water sample.
2. Place an ampoule (with yellow liquid) tip down in the sample cup. Snap the tip by pressing the ampoule against the side of the cup. The ampoule will fill, leaving a small bubble (air-space) to facilitate mixing.
3. Mix the contents of the ampoule by inverting it several times, allowing the bubble to travel from end to end each time. Then wipe all liquid from the exterior of the ampoule. Wait 2 minutes for colour development.
4. Hold the comparator in a horizontal position while standing directly beneath a bright light source (try to avoid reflective interference). Place the ampoule between the colour standards moving it from left to right along the comparator until the best colour match is found. If the colours do not match, a concentration estimate can be made (range).
5. Dispose of excess water sample in a sink. Dispose of broken tip of ampule (if not already emptied into a sink) and ampule in a “sharps” container. Return the chemicals and equipment to the container.
6. Repeat the measurement of dissolved oxygen with a water sample taken to the lab.

Carbon Dioxide (CO_2) Test Kit (LaMotte)

Contents: 20 mL glass test tube with cap, Phenolphthalein indicator 1%, Reagent B, Titrator (syringe), 0-50 divisions

1. Fill test tube to 20 ml mark with sample water.
2. NOTE: For best results test on freshly obtained sample, avoid splashing or prolonged contact with air.
3. Add 2 drops of phenolphthalein indicator. If solution remains colourless, proceed to step
4. If the solution turns red, no free Carbon Dioxide is present.
5. Fill titrator with Reagent B. Insert titrator into center hole of test tube cap.
6. While gently swirling the test tube, add Reagent B one drop at a time, until a faint pink colour is produced and persists for 30 seconds. Read test result where plunger tip meets titrator scale. Record as ppm carbon dioxide.
7. Rinse test tube and titrator with distilled water. Return the chemicals and equipment to the container.
8. Repeat the measurement of dissolved carbon dioxide with a water sample taken to the lab.

Water Testing in the lab (or field, if feasible)

Phosphate CHEMets® Kit

Contents: 25 mL sample cup, activator, ampoules, low and high range comparators.

1. Fill the sample cup to the 25 mL mark with the sample to be tested.
2. Add 2 drops of A-8500 Activator Solution. Cap the sample cup and shake it to mix the contents well.
3. Place the CHEMets® ampoule, tip first, into the sample cup. Snap the tip. The ampoule will fill leaving a bubble for mixing.
4. To mix the ampoule, invert it several times, allowing the bubble to travel from end to end.
5. Dry the ampoule and wait 2 minutes for colour development.
6. Obtain a test result using the appropriate comparator.
7. Low Range Comparator: Place the ampoule, flat end first, into the comparator. Hold the comparator up toward a source of light and view from the bottom. Rotate the comparator until the best colour match is found.
8. High Range Comparator: Place the ampoule between the colour standards until the best colour match is found.

Potassium (K) Test Kit (Lamotte)

Contents: Sodium Hydroxide 6% (must be replaced before the lab),

Tetraphenyl-Boron powder, Spoon, Potassium double tube. Brush.

1. Fill sample water into the round tube up to lower line.
2. Add distilled water to the upper line.
3. Add two drops sodium hydroxide (corrosive, use gloves). Cap and mix.
4. Add one measure of Tetraphenylboron powder, using the 50 mg spoon.
5. Cap and shake vigorously for 30 seconds. If potassium is present, a white precipitate will form.
6. Wait for 5 minutes.
7. Shake the tube again, remove cap and insert the square tube with the collar. The square tube will slide down through the collar and fill with liquid.
8. Looking from above, slide the square tube down into the solution until the black dot is no longer visible.
9. Read the scale as ppm K (parts per million potassium).
10. Clean test tubes thoroughly after the experiment.

pH

The pH value is measured with an electrode connected to the instrument. Ensure that the basal part of the electrode is completely immersed in the liquid to be measured. Before and after each measurement, the electrode must be rinsed with distilled water. If you are not using the electrodes for more than a few minutes, ensure that they are immersed in liquid.

Before you take any measurements with the pH-meter, the instrument must be calibrated (standardized) with various buffer solutions. A calibration with various buffers should be done at least once every day.

When you take a measurement, wait for the “Stable” indicator before taking a reading.

Sample notebook record for water chemistry:

14 August 2011, 10:00, Nile River landing
pH field measurement, 10:04 (simple test strip): pH 7
pH field measurement, 10:14 (multi parameter test strip): pH 7.5
pH lab measurement, 14:51 (simple test strip): pH 7
pH lab measurement, 14:55 (multi parameter test strip): pH 7.5
pH lab measurement, pH-meter, 15:32 pH 6.9
dissolved oxygen field measurement, 10:05 (colourimetric): 7 ppm
dissolved oxygen lab measurement, 14:05 (colourimetric): 6 ppm
dissolved CO_2 field measurement, 10:10 (titration): 7 ppm
dissolved CO_2 lab measurement, 14:05 (titration): 6 ppm
potassium (K) lab measurement, 15:10 (titration): 7 ppm
phosphorus (PO_4) lab measurement, 14:05 (colourimetric): 6 ppm

Note: lab measurements were taken with water samples from the field (not freshwater samples)

Measurements taken by Claire Musavene, student at Cairo University

Collection of data

Group 2 will complete and submit copies of Table 2 separately for each water body.

Table 2. Water Chemistry

- Use one sheet per location

Location*	
Date:	
Time:	
Weather, Air temperature	
Notes	

	Field Recordings	Lab Recordings
Time		
Dissolved O_2		
Dissolved CO_2		
pH method 1		
pH method 2		
pH method 3		
pH method 4		
Hardness		
Alkalinity		
phosphate		
potassium		

*Athabasca River, Tawatinaw River, Muskeg Creek (Middle Bridge), ARC Pond

Describe below any differences for pH measurements using different methods (various test strips and pH-meter).

Describe below any differences for dissolved O_2 and CO_2 measurements between lab and field.

Project 3: Bacteriology

Group 3 will take two water samples per location. After completion, Group 3 will assist Group 4 in the field.

Activities in the field:

- Collect two water samples per location. Keep on ice.

Lab activities:

- use the water samples to run the Coliscan test (1 or 2 tests per location)

Materials to take outdoors: 10 Whirl-Pak sample collection containers, 3 marker pens, cooler box with ice, camera (cell phone OK).

Materials to be used in the lab: Coliscan test kit. Disposable 1 mL plastic pipets (1 bag), 37 ^{0}C incubator.

Safety considerations

Field: Avoid taking samples from slippery slopes or stones. Test first if measurement and sampling can be done safely. Always work with at least one partner.
Lab: After analyzing the results and taking high quality photos, the petri dishes must be disposed of in a red autoclave bag for subsequent sterilization.

Methods

Water Sampling

Use a collection bottle to take a water sample from the surface of the water body. When feasible, use an extension pole. Use care to not create any turbulence, especially in lentic, sediment rich waters. After filling your sample bottles, keep them on ice, for later processing in the lab.

Coliscan® Easygel®water testing for *E. coli* and total coliforms.

Coliform bacteria are lactose-fermenting, gas-producing, Gram-negative rods in the family Enterobacteriaceae. They are often found in soil and water. Fecal coliforms are found in human and animal digestive tracts and when found in water bodies can indicate fecal contamination. *E. coli* is a dominant

component of the intestinal microbiome and is often used as an indicator of fecal contamination from waste treatment, natural animal waste, or agricultural activities.
Use each of your samples to prepare a Coliscan Easygel plate. The solidified plates will be incubated at 37˚C.

Coliscan® Easygel® Instructions (slightly modified from the provided instructions)

The Coliscan® Easygel® medium is a patented formulation for water testing. It contains a sugar linked to a dye which, when acted on by the enzyme β-galactosidase (produced by coliforms including *E. coli*), turns the colony a pink colour. Similarly, there is a second sugar linked to a different dye which produces a blue-green colour when acted on by the enzyme β-glucuronidase. Because *E. coli* produces both β-galactosidase and β- glucuronidase, *E.coli* colonies grow with a purple colour (pink + blue). The combination of these two dyes makes possible the unique ability to use one test to differentiate and quantify coliforms and *E. coli*. (Because *E. coli* is a member of the coliform group, add the number of purple colonies to the number of pink colonies when counting total coliforms.)

Instructions

1. Collect your water samples (2 replicates per location) in a sterile container and store on ice. Transport the water back to the lab.
2. Label the petri dishes with the appropriate sample information. A permanent marker or wax pencil will work.
3. Sterilely transfer 1 mL water from the sample containers into the bottles of Coliscan® Easygel®. Swirl the bottles to distribute the inoculum and then pour the medium/inoculum mixtures into the correctly labeled petri dishes. Place the lids back on to the petri dishes. Gently swirl the poured dish until the entire dish is covered with liquid (but be careful not to splash over the side or on the lid).
4. The dishes may be placed right-side-up directly into a level incubator or warm level spot in the room while still
   liquid. Solidification will occur in approximately 45 minutes.
5. Incubate at 35^0 C for 24 hours, or at room temperature (20^0 C) for 48 hours.
6. Inspect the dishes.
   a. Count all the purple colonies on the Coliscan dish (disregard any light blue, blue-green or white colonies), and report the results in terms of *E. coli* or Fecal Coliform per mL of water. Fecal coliforms are typically reported per 100 mL, therefore you multiply the number of counts on your plate with 100 in order to obtain Fecal coliforms/100 mL. (therefore 2 purple colonies equals 200 fecal coliforms/100 mL).

b. Count all the pink **plus** the purple colonies on the Coliscan dish (disregard any light blue, blue-green or white colonies) and report the results in terms of coliforms per mL of water. Multiply by 100 to obtain total coliforms/100 mL. Record and describe any colonies that are neither pink nor purple separately.

Sample notebook record for Coliscan:

1 July 2019, 10:05, Murray River (Physical factors and more details to be obtained from Group 1)
Water was collected in 2 sample bottles and transported back to the lab. The Coliscan test was carried out at 13:03 with collected water samples from the field (not freshwater samples).
Reading of results, Replicate 1: 4 July 2019, 06:20
Pink colonies: 30
Purple colonies: 17
Unidentified colonies: 3 small gray colonies and 2 white colonies, < 1mm
Data recorded by Matilda Beguirre, student at the University of New Delhi

Note: All records above must be accompanied by good quality photos of the plates confirming your stated results.

Collection of data

Group 3 will complete and submit a copy of Table 3. Results will become available on Day 2 or Day 3. In addition, all results must be documented photographically and the photo must be of a quality that allows verification of the count.

Table 3. Bacteriology

Date: Notes:	Replicate 1: # Purple colonies / # Pink colonies Additional colonies (if any)	Replicate 1: Fecal coliforms/100mL Total coliforms/100mL	Replicate 2: # Purple colonies / # Pink colonies Additional colonies (if any)	Replicate 2: Fecal coliforms/100mL Total coliforms/100mL
Muskeg Creek, Middle Bridge Time:				
Athabasca River, Landing Time:				
Tawatinaw River Time:				
AU ARC Pond Time:				
AU Observatory Pond Time:				
Location 6: Time:				
Location 7: Time:				
Location 8: Time:				
Location Peace River: Time: 10:06	Example: 4/7 Two unidentified gray colonies were also observed	400/1100	Example: 2/8 Three large, white unidentified colonies were also observed	200/1000

Project 4: Aquatic organisms

Group 4 will collect invertebrates, aquatic plants and protists from water and sediment. Vertebrates will be recorded but not collected.

Materials to take outdoors: 10 clean water collection bottles (wide mouth, large size), 1 small bucket for sediments, 3 aquarium nets, 3 strainers of various sizes, five forceps (large and small sizes), disposable plastic pipets (1 bag), 2 trowels to collect sediments, 2 large plastic funnels, 10 white plastic trays with various sizes, ziploc bags (1 box, medium size, 1 box large size), 2 clipboards, 3 marker pens, camera (cell phone OK), bags for carrying heavy water samples (ideally back packs), kick nets, 10 glass jars (500mL or larger).

Materials to be used in the lab: 5 dissection microscopes, 1 compound microscope, 5 plastic strainers of various sizes, 10 white plastic trays with various sizes, (same as from field activities), set of sieves with assorted mesh sizes, 30 empty petri plates, 1 regular balance, disposable plastic pipets (1 bag).

Activities in the field

Check out each sample site by taking preliminary observations on the richness of flora and fauna. Evaluate collected samples in the trays.
Field results must be amply documented with photos.

At the main site (Muskeg Creek):

Collect both water (plankton) and sediment samples for lab analysis, as well as individual invertebrates or plants.

- collect water sediment and water samples for immediate preliminary observation in the field (use white trays for examination in the field); take sediment in a bucket back to the lab for further observations
- collect plants and invertebrates in larger bottles or large ziploc bags
- take photographs in the field and record all aquatic animals and plants that are not taken to the lab

Lab activities

- use identification keys and microscopes to identify the collected organisms as accurately as possible
- use the stacked sieves for filtering your sediment samples and record your organisms qualitatively (see below)

Safety considerations

Field: Avoid taking samples from slippery slopes or stones. Test first if measurement and sampling can be done safely. Always work with at least one partner.

Methods

Sediment samples are separated in the lab through a set of sieves with assorted mesh sizes.

1. Pass the entire sample (do not sub-sample) through a nested column of sieves ranging in opening sizes of 4 mm to 425 µm. The larger opening sizes must be towards the top.

2. Collect the organisms caught in the sieve into white trays for sorting. Use Petri plates to sort the organisms into subgroups.

3. Enter the results for Zoobenthos in Table 4.

4. Enter the results for Plankton in Table 5. These are the freely floating organisms in your water samples (not zoobenthos).

5. Enter any notes about aquatic plants in Table 6.

Note: Compound and dissection microscopes will be necessary for most identifications. Consult your lab instructors if you need help with the microscopes.

Identify the collected organisms as precisely as possible using books and online resources provided in the lab manual.

Table 4. Zoobenthos

Location, Date and Time:		
Phylum	**Class/Order**	**Count**
Porifera	Sponges	
Cnidaria	Hydrozoa	
Plathyhelminthes	Flatworms	
Nematoda	Roundworms	
Annelida	Olligochaeta	
Annelida	Hirudinea (leeches)	
Mollusca	Gastropoda (snails & limpets)	
Mollusca	Bivalvia (clams)	
Arthropoda	Hydrachnidia (aquatic mites)	
Arthropoda	Cladocera (water fleas)	
Arthropoda	Ostracoda (seed shrimp)	
Arthropoda	Copepoda (copepods)	
Arthropoda	Amphipoda (scuds)	
Arthropoda	Isopoda (sow bugs)	
Arthropoda	Decapoda (crayfish)	
Arthropoda	Ephemeroptera (mayflies)	
Arthropoda	Odonata (dragonflies and damselflies)	
Arthropoda	Plecoptera (stoneflies)	
Arthropoda	Hemiptera (true bugs)	
Arthropoda	Megaloptera (fishflies, alderflies)	
Arthropoda	Lepidoptera (aquatic moths)	
Arthropoda	Trichoptera (caddisflies)	
Arthropoda	Coleoptera (beetle adult)	
Arthropoda	Coleoptera (beetle larva)	
Arthropoda	Diptera (flies and mosquitoes)	
	Other animal groups	
	Additional observations and observations from other sites	

Table 5. Plankton organisms

Group	Total count	Concentration (Organisms / mL): for highly frequent groups
Culicids		
Copepods		
Cladocerans		
Rotifers		
Ostracods		
Others		
Additional Notes:		

Table 6. Aquatic plants

Mosses	Notes on aquatic plants:
Monocots	
Dicots	
Additional Notes:	

Day 2: TERRESTRIAL PRODUCERS AND CONSUMERS

Introduction

Over 91% of the total area of Canada is terrestrial and roughly 9% of the world's forests are found in Canada. In these exercises we will be surveying the terrestrial plant community and invertebrate community in two adjacent habitats.

Project 5: Terrestrial Producers

Group 5 will use transect sampling to explore a transition zone from a natural forest environment to a human-created meadow.

Materials to take outdoors: Small metal quadrats, large plastic quadrats, grass sheers, pruning sheers, white plastic or enamel pans, measuring tape, 100 m and 30 m chain,
data sheets (multiple copies from lab manual), scrap paper (several sheets), plastic bags for specimen collection, rulers to measure tree diameters, 30 marker pens, several pens and pencils.

Materials to be used in the lab: 5 balances, dissection microscopes, trays for plants, identification keys.

Methods

A 100 m transect (with a surveyor "chain") will be set up between the forest and meadow. The large quadrats are positioned to the right of the "chain" in 10 m intervals and all 0.1 m^2 quadrats are positioned equally within the 1 m^2 quadrats.

Figure 1. Small rectangle within large quadrat.

Run the chain from the meadow into the forest. Start in the meadow and position the chain so that approximately 50 m of the chain is in the meadow and 50 m is in the forest.

At the 0 m mark both a 0.1 m^2 and a 1 m^2 quadrat will be placed to the right of the meter marking on the chain. The 0.1 m^2 quadrat is nested within the 1 m^2 quadrat.

In each 0.1 m^2 quadrat at ground level clip off live material of all herbaceous plants and record numbers of each within the quadrat, separately for monocots and dicots. (As a rule of thumb, monocots have parallel veins on their leaves, while the veins of dicot leaves are reticulate, with a net-like pattern). Save each aboveground plant class (monocot and dicot) in separate plastic bags for later weighing. Record the quadrat number and plant class on a slip of paper and put the paper in the plastic bag with the appropriate plants.

In each 1 m^2 quadrat, record the diameter and height of woody shrubs greater than 1.0 m tall.

Repeat these steps for the remaining 10 intervals on the chain.

In the lab

- Use a pan balance to weigh all individuals of each group per quadrat.

Data Recording

Table 7. Understory plants

(for herbaceous plants in a 0.1 m^2 quadrat)
** you will need 1 copy per quadrat or 11 in total

Location on transect (m)	Plant class (M = monocot, D= dicot)	Number of individuals in quadrat	Wet Mass (g)	Wet Mass per m^2 (multiply by 10)

Table 8. Woody plants

Shrubs greater than 1 m high (1 m^2 quadrat)

Location (m)	Plant species	Basal diameter estimate (cm)	Height estimate (m)	Volume (cm^3)	Biomass (g)

Note. Biomass of woody plants over 1 m tall will be estimated by measuring the basal radius (r = 1/2 diameter), estimating the height (h in centimeters) of the plant, using the formula for the volume a cone $V = 1/3\ \pi r^2 h$, and using 1 cm^3 = 1 g for the biomass of the tall woody plants.

Table 9. Quadrats in an Ecotone between Two Terrestrial Habitats (meadow and forest).

Plant class	Transect station (m from start)	0		10		20		30		40		50	
	Plant Species/ Group	Absolute Density (number/m^2)	Mass (g/m^2)	Absolute Density (number/m^2)	Mass (g/m^2)	Absolute Density (number/m^2)	Mass (g/m^2)	Absolute Density (number/m^2)	Mass (g/m^2)	Absolute Density (number/m^2)	Mass (g/m^2)	Absolute Density (number/m^2)	Mass (g/m^2)
Monocots													
Herbaceous dicots													
Woody dicots													
Miscellaneous													
	Totals												

Plant class	Transect station (m from start)	60		70		80		90		100	
	Plant Species/ Group	Absolute Density (number/m^2)	Mass (g/m^2)	Absolute Density (number/m^2)	Mass (g/m^2)	Absolute Density (number/m^2)	Mass (g/m^2)	Absolute Density (number/m^2)	Mass (g/m^2)	Absolute Density (number/m^2)	Mass (g/m^2)
Monocots											
Herbaceous dicots											
Woody dicots											
Miscellaneous											
	Totals										

Project 6. Terrestrial animals

Invertebrates are a very diverse group of animals. We will compare invertebrate and vertebrate communities in adjacent forest and meadow habitats using traps, game camera recordings and nets.

Materials used in the field: Mechanical aspirators, clear plastic vials for holding insects, beating nets, butterfly nets, beating trays, insect killing jars, ethyl acetate or chloroform for killing jars, collection bottles and sieves for pit-fall traps and ramp traps, funnel with narrow opening to fit into collection bottles, light trap, battery, buckets for carrying, 8 pails for pit traps, 4 pails for ramp traps

Trap killing mixture: equal amounts of 50% propylene glycol and 75% ethanol, added by a few drops of detergent.

Materials used in the lab: Dissecting microscopes, Dissecting kits, Petri dishes, Pan balances, Insects of Alberta display box Appendices for invertebrate identification, Library resources for invertebrate identification

Methods

Activities in the field

- Collect the contents of pit-fall traps (Figure 2 and Figure 3). Pit-fall traps are plastic containers filled with a propylene glycol mixture. The container is placed in a small hole such that the top of the container is flush with the ground. Small ground-dwelling animals will fall into the trap where they are preserved by propylene glycol. The pit-fall traps were set up two weeks before the lab. Carefully remove the plywood cover. Place a funnel with sieve over the propylene glycol waste container. Lift out the trap and pour the contents through the sieve. Place the insects into a collection jar. Label.
- Collect the contents of ramp traps. Ramp traps are similar to pit-fall traps except that the top of the trap is elevated, and small animals enter the trap through clear plastic ramps on either side of the trap. Ramp traps are most effective in rocky and hard ground where it is not possible to dig a whole.
- The instructors will demonstrate the use of beating nets, butterfly nets, beating trays and aspirators. Use each technique of collecting insects for the same amount of time in the forest and the meadow habitats. For example, if you use the butterfly net for 20 minutes in the meadow then use the butterfly net for a second 20-minute period in the forest. Count the number of each order of insects collected using these methods. If live identification is possible then release the

insects. A killing jar may be used to kill insects that must be identified in the lab.

- The instructors have previously set up a light trap in each of the forest and meadow habitats. Light traps are used to catch night-time flying insects that are attracted to UV light (Figure 4). These traps consist of a light source powered by a car battery. The insects are attracted to the light (simulating the moon) and collide with clear Plexiglas panes radiating from the light source. The stunned insects drop into a bucket that contains a preservative (ethanol). The traps will be in place for one night at each site. Collect the content of the traps. The ethanol in the trap must be collected for disposal.

Activities in the lab

- Empty the contents from traps into plastic trays. For each trap type count the number of invertebrates in each order. Use Petri plates (used) to help separate the insects into groups.

Evaluate game camera results for both sites:

Forest:

Meadow:

Game camera photos must be uploaded to the Landing

Figure 2. Uncovered pit-fall trap. Photo courtesy of C. Allen.

Figure 3. Pit-fall trap with plywood cover. Photo courtesy of C. Allen.

Figure 4. Light trap in operation. Battery enclosed in plastic bin. Photo courtesy of C. Allen

Table 10. Invertebrates collected using traps in two terrestrial habitats (meadow and forest).

Animal group	Common names	Forest			Meadow		
		Pit traps	Ramp traps	Light traps	Pit traps	Ramp traps	Light traps
		No.	No.	No.	No.	No.	No.
Annelida, Oligocheata	earth worms						
Mollusca, Gastropoda	snails, slugs						
Arachnida, Acari	mites and ticks						
Arachnida, Araneae	spiders						
Arachnida, Opiliones	harvestmen/ daddylonglegs						
Chilopoda	centipedes						
Collembola	springtails						
Insecta, Orthoptera	grasshoppers, crickets						
Insecta, Hemiptera	bugs: “half wings”						
Insecta, Homoptera	aphids, plant hoppers						
Insecta, Coleoptera	beetles						
Insecta, Trichoptera	caddisflies						
Insecta, Lepidoptera	butterflies and moths						
Insecta, Diptera	flies, mosquitoes						
Insecta, Hymenoptera	ants, wasps, bees						
Insecta, Ephemeroptera	mayflies						
others							
Totals							

Dates of pit trap collections:
Numbers of pit traps per habitat:
Trap days (number of traps x number of days):

Dates of ramp trap collections:
Number of ramp traps per habitat:
Trap days (number of traps x number of days):

Dates of light trap collections:
Numbers of light traps per habitat:
Trap days (number of traps x number of days):

Table 11. Invertebrates collected using hand nets in two terrestrial habitats (meadow and forest).

Time spent beat netting: Time spent butterfly netting: Time spent with beating tray:

Animal Group	Common names	Forest			Meadow		
		Beat net	Butterfly net	Beating tray	Beat net	Butterfly net	Beating tray
		No.	No.	No.	No.	No.	No.
Annelida, Oligocheata	earth worms						
Mollusca, Gastropoda	snails, slugs						
Arachnida, Acari	mites and ticks						
Arachnida, Araneae	spiders						
Arachnida, Opiliones	harvestmen/ daddy longlegs						
Chilopoda	centipedes						
Collembola	springtails						
Insecta, Orthoptera	grasshoppers, crickets						
Insecta, Hemiptera	bugs: “half wings”						
Insecta, Homoptera	aphids, plant hoppers						
Insecta, Coleoptera	beetles						
Insecta, Trichoptera	caddisflies						
Insecta, Lepidoptera	butterflies and moths						
Insecta, Diptera	flies, mosquitoes						
Insecta, Hymenoptera	ants, wasps, bees						
Insecta, Ephemeroptera	mayflies						
other							
Totals							

Day 3: ABIOTIC FACTORS (TERRESTRIAL)

Introduction

In the first part of this exercise, we will compare the microclimates of two adjacent terrestrial habitats. We will also use a soil auger to collect soil samples and take temperatures of the soil at various depths and determine various soil nutrient levels.

We will measure and record climate and temperature data in the field. Soil cores will be taken for later analysis.

Materials to be used outdoors: 2 Stevenson screens, 2 rain gauges, 2 HOBO loggers, sling psychrometer, Garmin GPS, Max/min thermometer, alcohol/mercury thermometer, soil thermometer, soil auger with extensions, spatula, 3 thermocouples on soil probe, bags for soil samples, anemometer, light meter, marker pens, infrared thermometer, camera (cell phone OK).

Materials to be used in the lab: pH-meter (use only the units designated for soil suspensions), pH test strips (different types), 20 beakers (250 mL), 20 beakers (400 mL), 20 small strainers, 10 beakers (plastic or glass, 100 mL), 10 empty petri dishes, distilled water, stock solutions of KCl and $CaCl_2$, 6 spoons, 30 plastic pipettes, 30 snap-cap vials (different sizes).

Methods

Activities in the field

The lab instructors will demonstrate the use of devices.

Instantaneous meteorological measurements in each habitat type:

- Use a GPS device to determine the location and elevation of the habitats. Try a few different units to see if they are consistent.
- Measure the air temperature using an alcohol thermometer.
- Measure the relative humidity using a hygrometer.
- Use an infrared thermometer to measure the temperature of a plant surface in the sun and the shade.
- Measure the light intensity using a light meter.
- Measure the wind velocity at ground level and 1 m above the surface using an anemometer.
- Use a soil auger to drill 1.2 m into the soil substrate. Record the depth of each soil horizon (O, A, B). Take photos.
- Use a pre-prepared stick with thermocouples attached at 0 cm (surface), 20 cm, 40 cm, 60 cm, 80 cm, 100 cm and 120 cm to measure

the soil temperature.

Prolonged meteorological measurements in each habitat type:

- Open the Stevenson's screen
- Measure the current temperature using a maximum/minimum thermometer.
- Record the maximum and minimum temperatures experienced by the thermometer since the last reset.
- Collect the Hobo Logger with thermocouple and bring it to the lab for data harvest.
- Use rain gauge to measure the amount of precipitation.

Soil sampling

- Use a soil auger to view the soil profile for the two sites. Record as described above (notes and photographs). Collect soil from the A (topsoil), and B (subsoil) horizons for pH measurements. Place the samples in labeled plastic bags.
- Determine the soil type using hand texturing:
  - Gather the soil sample
  - Wet the soil and kneed into a ball for the ball test (Figure 5)
  - Form a wire with the soil as demonstrated by the instructors for wire test (Figure 6)
  - Form a ribbon with the soil by squeezing the wire between your thumb and forefinger for the ribbon test (Figure 7)
  - Rub a small sample between your thumb and forefinger to assess texture for feel test (Figure 8)

Step 1: Ball test

Place approximately 25 g (about one to two tablespoons) of soil in your hand.

Moisten the soil and work it to break down all aggregates (this can be time-consuming in a clay soil).

Squeeze the soil and try to make a ball.

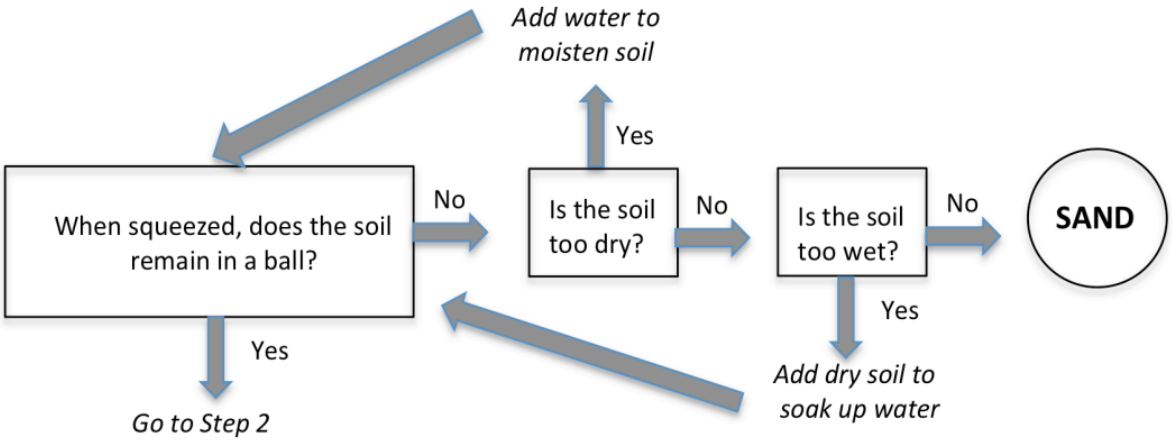


Figure 5. Ball test (Watson and Pennock, 2016)

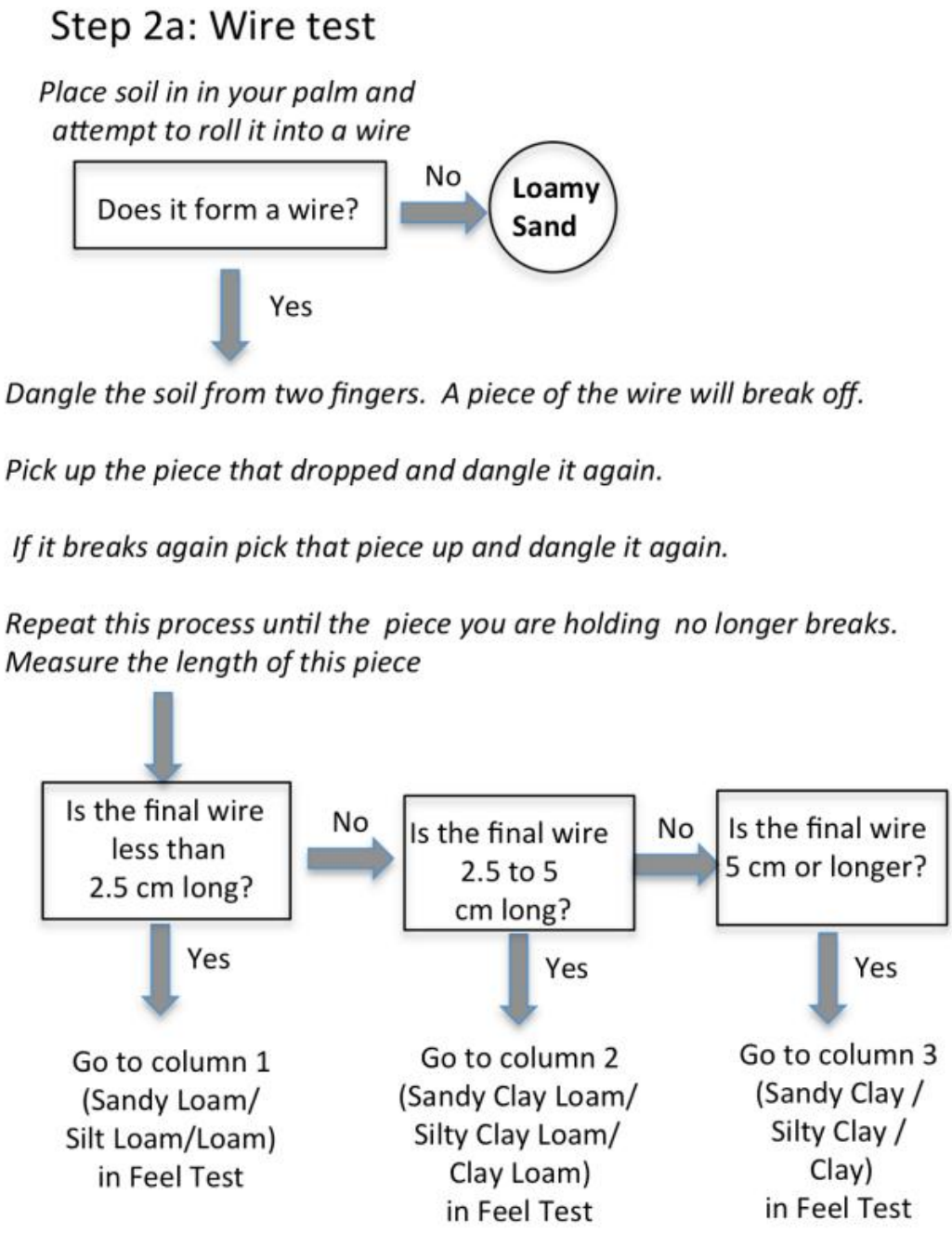


Figure 6. Wire test (Watson and Pennock, 2016)

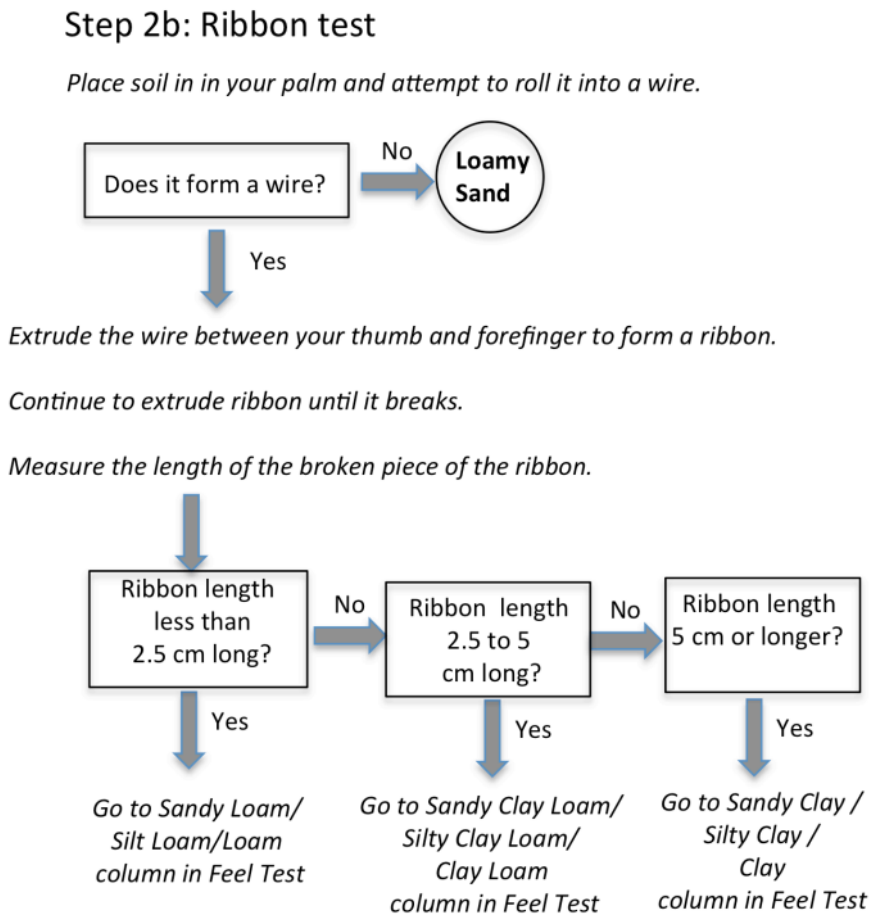


Figure 7. Ribbon test (Watson and Pennock, 2016)

STEP 3: Feel Test

Select column based on Wire or Ribbon test

Wet a small amount of soil and rub between thumb and forefinger

Sand: Feels grainy/gritty; grains are visible for all but very fine grains

Silt: Non-sticky; feels smooth like flour or powder

Clay: Sticky –wet soil adheres to fingers and stretches before breaking when fingers are separated

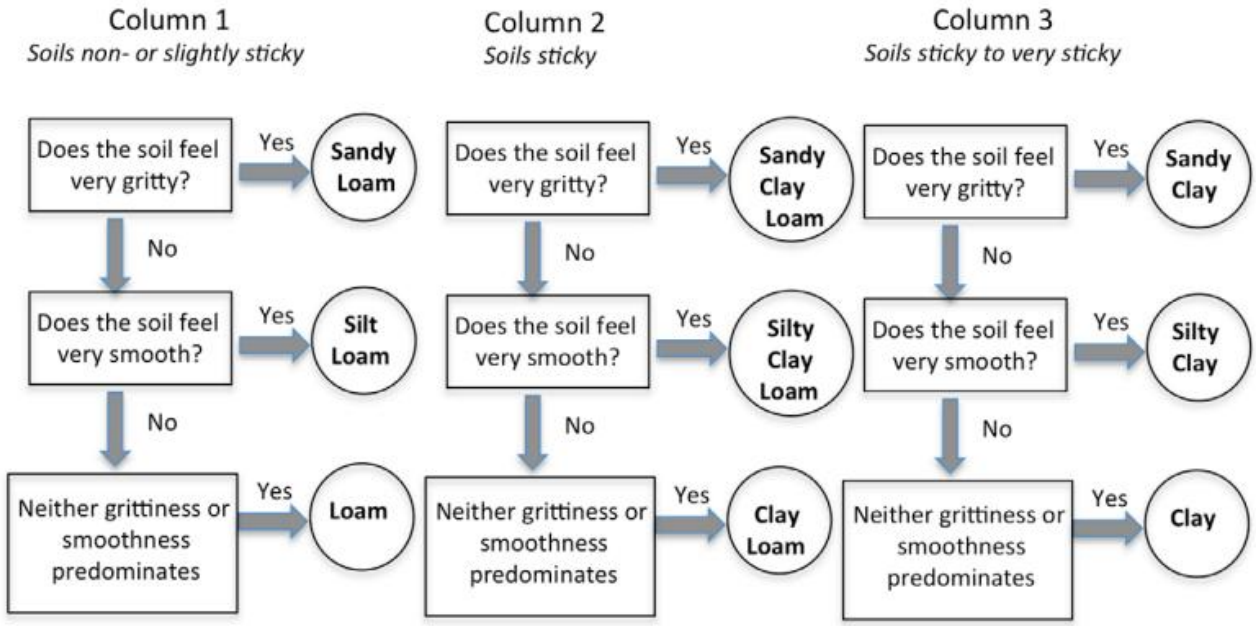


Figure 8. Feel test (Watson and Pennock, 2016)

Activities in the lab

Prepare 3 soil suspensions by mixing equal amounts (e.g. 60 g soil in 60 mL solution) of soil samples with

- distilled water
- $CaCl_2$ stock solution
- KCl stock solution

Immerse for at least 30 minutes. This will work best for the B horizon. You may have to strain the soil suspension repeatedly to obtain a soil suspension that can be used for the pH measurement. Measure the pH both with a pH-meter and with various pH test strips.

Using the pH-meter: Use only the units assigned for soil suspensions. The pH value is measured with an electrode connected to the instrument. Ensure that the basal part of the electrode is completely immersed in the liquid to be measured. Before and after each measurement, the electrode must be rinsed with distilled water. If you are not using the electrodes for more than a few minutes, ensure that they are immersed in liquid.

Before you take any measurements with the pH-meter, the instrument must be calibrated (standardized) with various buffer solutions. A calibration with various buffers should be done at least once every day.

When you take a measurement, wait for the “Stable” indicator before taking a reading. Record your results for both sites.

Table 12. Abiotic factors Forest/Meadow

Date and Time:		
	Forest	Meadow
Location	Coordinates:	Coordinates:
	Elevation:	Elevation:
Air temperature	Current:	Current:
	Max:	Max:
	Min:	Min:
	1m:	1m:
	Soil interface:	Soil interface:
Soil temperature	0cm:	0cm:
	20cm:	20cm:
	40cm:	40cm:
	60cm:	60cm:
	80cm:	80cm:
	100cm:	100cm:
	120cm:	120cm:
Temperature Plant surface	Sun:	Sun:
	Shade:	Shade:
Relative humidity		
Precipitation in mm (absolute) / mm per month		
Light intensity 1 m above surface		
Wind velocity	1m:	1m:
	Ground:	Ground:
Notes on Hobo logger results (e.g. maximum/minimum for both temperature and humidity): In addition to submitting this table, the Hobo logger graphs must be uploaded to the AU Landing.		

Table 13. Soil pH

Date and Time:		
	Forest	Meadow
pH in distilled water (pH-meter)		
pH in KCl (pH-meter)		
pH in $CaCl_2$ (pH-meter)		
pH test strip reading (indicate type)		
pH test strip reading (indicate type)		
pH test strip reading (indicate type)		
Additional notes: (e.g. soil horizon used, comments on variability for different measurement methods)		

Day 4: FOREST MEASUREMENTS

Introduction

The boreal forest covers close to 60% of Canada's land area. The forest stretches from the east to west coast and is mostly above the 50th parallel. The boreal forest benefits Canadians economically, including a widely distributed forestry sector that is crucial to many rural communities across the country. The trees and understory plants filter pollutants from the air and water. The boreal forest creates essential habitat for native plant and animal species, sustaining biological diversity within our country. The boreal forest provides opportunities for recreation, cultural, and spiritual activities.
Disturbances play a large role in the shape of Canada's forests. Natural disturbances have been influencing forest structure since the last glaciation and include fire, insects, disease, drought, and windstorms. These disturbances can renew entire forest landscapes and alter forest composition, structure, and habitat diversity. Humans also disturb the forest ecosystem through harvesting, energy sector activities, road construction, conversion to agriculture, and so on.
Forests are a vital component of the carbon cycle, storing and releasing carbon through growth, decay, disturbance, and renewal.
Forest professionals must be able to measure the condition and state of forest resources to make decisions about forest health and management.

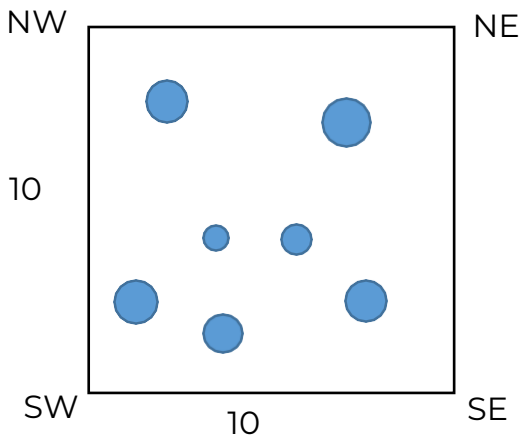
In this exercise we will learn some of the basic techniques of forest measurement.

Materials to take outdoors: 20-meter tape, clinometer, increment borer, DBH tape, compass, flags, flagging tape, core holder, hypsometer, folding meter, Range finder ("Bushnell", for measuring distances), wedge prism to measure basal area.

Materials to be used in the lab: dissection scope

Square plot

In a forested area on the AU campus, we will lay down a 10 m by 10 m plot. The sides of the square plot will run north to south and east to west. Use a compass and measuring tape to define the boundaries of the plot and mark them clearly.



Total area = 100 m^2

Use flags to mark the periphery of the plot.

Using flagging tape to mark trees near the border of the plot

For each tree > 5 cm diameter at breast height (DBH) within the plot

- Identify the species.
- Measure the DBH using the DBH tape. Wrap the DBH tape around the tree at 1.4 m from the base of the tree. Measure the diameter in centimeters. If no DBH tape is available, then measure the circumference at 1.4 m and divide by π.
- Determine the height using a 20 m measuring tape (or a "range finder" and a hypsometer. An instructor will demonstrate how to use the hypsometer.
- For one tree of each species within the plot: Determine the age using an increment borer. An instructor will demonstrate how to use an increment borer.

Table 14. Basal area and volume of each tree using a square plot.

Tree #	Tree species	DBH (m)	Basal Area (m^2)	Height (m)	Volume (m^3)

Basal area (BA) per tree is calculated as $\pi \times (DBH/2)^2$. Volume measurements are taken from the single tree volume tables (Appendix) Data Analysis

Table 15. Tree density using a square plot.

Tree Species	Number of trees per 100 m^2	Absolute density per hectare (move two decimal places)	Relative Density (%)
Totals		Total density =	100%

- For each tree species calculate the absolute density per hectare. A hectare is defined as 100 m by 100 m or 10000 m^2.
- Relative density of a species is calculated as (absolute density of the species/total density of all species) x 100.

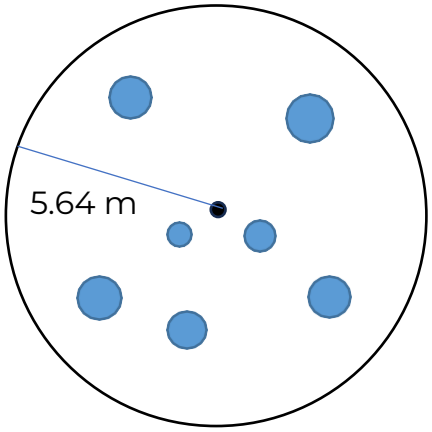
Table 16. Relative dominance using a square plot.

Tree Species	Basal Area in 100 m^2	Basal Area per hectare (move two decimal places)	Relative Dominance (%)
Totals			100%

- For each tree species calculate the total basal area per hectare (m^2/hectare). A hectare is defined as 100 m by 100 m or 10000 m^2.
- Relative dominance of a species is calculated as (basal area of the species/total basal area of all species) x 100.

Fixed radius plot

In a forested area on the AU campus, we will lay down a 5.64 m radius plot (100 m^2). Use a tape measure to define the boundaries of the plot and mark trees in bounds with flagging tape.



Total area = 100 m^2

Using flagging tape to mark trees near the border of the plot

For each tree > 5 cm diameter at breast height (DBH) within the plot

- Identify the species.
- Measure the DBH using the DBH tape. Wrap the DBH tape around the tree at 1.4 m from the base of the tree. Measure the diameter in centimeters. If no DBH tape is available, then measure the circumference at 1.4 m and divide by π.
- Determine the height using a 20 m measuring tape (or a "range finder" and a hypsometer. An instructor will demonstrate how to use the hypsometer.
- For one tree of each species within the plot: Determine the age using an increment borer. An instructor will demonstrate how to use an increment borer.

Table 17. Basal area and volume of each tree using a circular plot.

Tree #	Tree species	DBH (m)	Basal Area (m^2)	Height (m)	Volume (m^3)

Basal area (BA) per tree is calculated as $\pi \times (DBH/2)^2$. Volume measurements are taken from the single tree volume tables (Appendix)

Table 18. Tree density using a circular plot.

Tree Species	Number of trees per 100 m^2	Absolute density per hectare (move two decimal places)	Relative Density (%)
Totals		Total density =	100%

- For each tree species calculate the absolute density per hectare. A hectare is defined as 100 m by 100 m or 10000 m^2.
- Relative density of a species is calculated as (absolute density of the species/total density of all species) x 100.

Table 19. Relative dominance using a circular plot.

Tree Species	Basal Area in 100 m^2	Basal Area per hectare (move two decimal places)	Relative Dominance (%)
Totals			100%

- For each tree species calculate the total basal area per hectare (m^2/hectare). A hectare is defined as 100 m by 100 m or 10000 m^2.
- Relative dominance of a species is calculated as (basal area of the species/total basal area of all species) x 100.

Variable radius plot

Using a Wedge Prism to Estimate Basal Area
Wedge prisms provide a fast and efficient method for estimating the basal area of a forest stand. Whether a tree is included or excluded from the plot depends on its diameter at breast height (DBH) and its distance from the plot center.

Instructions:

1. Positioning the Prism:
   Stand at the plot center and hold the prism at arm's length. Keep the long edge of the prism horizontal, holding the thicker edge with your right hand.
2. Viewing Trees:
   Point the top of the prism at the trunk of a tree at breast height. Look through the prism with one eye. Due to light refraction, the image seen through the prism will appear offset from the actual tree.
3. Determining Inclusion:
   a. If the offset image is perfectly aligned or overlaps with the actual tree trunk (as seen above the prism), the tree is included in the plot.
   b. If the images do not touch, the tree is excluded.
   c. For borderline trees, measure both the DBH and the distance from the plot center to the center of the tree. Calculate the plot radius factor
      $PRF = 50 / \sqrt{BAF}$
      So a tree is "in" if it's within a radius of
      $Distance = DBH \times PRF$

Table 20. Basal area and density of each tree species using a variable radius plot

Tree species	# trees in plot	BAF	Basal Area (m^2/ha)	Relative dominance (%)	Tree density (trees/ha)	Relative density (%)

BAF = basal area factor
Basal area = # trees * BAF
Relative dominance of a species is calculated as
(basal area of the species/total basal area of all species) x 100.
Approximate tree density = # trees included in plot
Relative density of a species is calculated as
(absolute density of the species/total density of all species) x 100

Table 21. Basal area and volume of three representative trees in variable radius plot

Tree #	Tree species	DBH (m)	Basal Area (m^2)	Height (m)	Volume (m^3)

Basal area (BA) per tree is calculated as $\pi \times (DBH/2)^2$. Volume measurements are taken from the single tree volume tables (Appendix)

Table 22. Tree volume per species in variable radius plot

Tree species	Average volume per tree (m^3 /tree)	Tree density (trees/ha)	Estimated volume (m^3/ha)

BIOL 345 Assessment for Field Ecology Workshop

The laboratory component for BIOL 345 is worth 20% of your total grade, and a minimum of 50% on the lab component is required to pass the course.

Your lab grade will be broken into the following components:

Submission of group results on the AU Landing	Participation mark	Group discussion	Lab report, submitted 3 weeks after lab
50%	10%	15%	25%

Group Research Projects

Students will form groups of 2-10 people (depending on class size) to work on specific topics on Day 1 and Day 2. Each group should assign specific tasks to each group member: one person will take the measurements using specific instruments, another person will record notes or take photographs. Importantly, the task of uploading group results will require a degree of responsibility as it affects the whole group (50% of your lab grade will be derived from your uploaded results).

Result submission: To obtain full marks, submit all completed tables with your results, as well as good quality photographs as well as other results (Hobo logger, Game camera photos) to the AU Landing. Results will be marked per group.

Group discussions

On Day 4, each group (from Day 1) will share their data tables, with suggestions on how to analyze their results for the lab report. Your participation in this discussion will be marked and constitute 15% of your lab mark.

Suggestions for analysis of data: Groups 1-3, compare one (or more) representative lentic site with one (or more) representative lotic site.
Group 4: with an emphasis on Site 3 (Muskeg Creek), evaluate its biodiversity (for different biota) in comparison with similar sites that you find in the literature.

How to write a lab report for BIOL 345

Laboratory reports are very similar to the format that scientists use to communicate research findings. Reading some scientific papers in ecology journals will help you to write your own lab reports.

The format for lab reports will be as follows: Title, Abstract, Introduction, Materials and Methods, Results, Discussion, and References. This is a common structure for the presentation of primary research.

Scientific writing avoids direct quotes for citations. Paraphrase instead.
Example:
Incorrect: … Smith and Cornish (2011) reported "a higher amount of iron in all lakes close to the mining activities" which suggests…
Correct: … all lakes adjacent to the mining areas were found to have an increased iron content (Smith and Cornish 2011), therefore …

Use past tense and passive style consistently. E.g. "…flow speed was measured…".

- Title (3). The title tells the reader in one sentence what the lab report is about. The first letter is capitalized, and all other words are lower case (unless a proper name or generic name). The title should not exceed 120 characters.
- Abstract (15). The abstract provides a brief summary of the research including the research hypothesis (or research question), methods (briefly), results, and (briefly) main conclusions. All main results (data) must be included in the abstract. Do not cite references in the abstract. The purpose of the abstract is for a reader to quickly learn the research contents and determine if the paper is relevant to their interests. Abstracts should not be longer than 200 words.
- Introduction (15). The introduction should provide a general background to the research that familiarizes readers with relevant information on your topic and is supported by citations. In the final paragraph the introduction should briefly state the rationale and objectives of the study, as well as the research hypothesis.
- Materials and Methods (7) The methods inform the reader how you did your research. There should be enough detail that someone could repeat the study. A clear description of the experimental design and sampling is required. Describe the study site and the location. The methods section is written in passive style and in the past tense. Do not include results in the methods section.
- Results (15). Describe your group results in the past tense. Do not

interpret your data in the results section. To complement the text you should, where appropriate, include numbered tables and figures (e.g., Table 1 or Figure 1) that clearly and accurately represent the results. Tables are used to display true data. Graphs are used to highlight relationships between data. Tables and figures (photos or graphs) should stand alone, meaning that the reader should be able to look at the table without reading the paper, and gain a general understanding of the results. That means that the title or figure legend must be informative and complete, and axes should have titles and units as well. Each figure and table should be mentioned in the text. The purpose of the text is to point out trends in the data. Do not simply repeat the data from tables and figures in the text but mention the trends that you observe (without interpretation).

Discussion (20). In the discussion you will interpret the data in relation to your original objective or hypothesis. A discussion is a comparison of your results with those in the literature, therefore use ample citations in this section. Move from the specific to the general (i.e. start with the interpretation of your own results, before you start generalizing). Relate your interpretations to current knowledge on the topic and if possible future research direction. Do not merely reiterate the results. The discussion should compare results to those of other researchers, identify inadequacies of methods or analysis (if applicable), suggest explanations of unexpected results, and include some brief conclusions at the end. Avoid general statements with little relevance for your study.

References (15). In the reference section list the references specifically cited in the paper. All citations must be in the reference section, and all references must be cited. For this lab report we will follow CSE style in the Name-Year format: https://libguides.athabascau.ca/citationguide/othercitationstyles. Your lab report needs at least five primary references (articles in scholarly journals).

Language, structure, grammar, style (10). Reading some scientific papers will give you a good idea about scientific style.

Note: SI units are mandatory for all lab reports and science writing in general. Imperial units, such as miles, pounds, inches, degrees Fahrenheit etc. will incur mark deductions.

Resources

MASTERING BIOLOGY on your BIOL 345 course page has some helpful hints on graphs and data.
The links below are helpful for deciding what type of graph or chart you should select:

https://nces.ed.gov/nceskids/help/user_guide/graph/howto.asp
https://www.skillsyouneed.com/num/graphs-charts.html
https://www.doc.govt.nz/documents/science-and-technical/docts32entire.pdf

You can create graphs with Excel. If you haven't worked with Excel a lot, try the following resources: https://www.excel-easy.com/

References

Pennock, D.J., Watson, K., & Sanborn, P. (Eds.). (2016). Field Handbook for the Soils of Western Canada. Canadian Society of Soil Science.