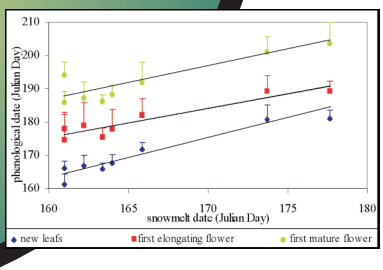
CANTTEX

Canadian Tundra and Taiga Experiment

Ecological Monitoring in the Canadian Arctic

FIELD MANUAL

Part B - Additional Methods and Experimental Manipulations



EMAN NORTH

CANTTEX Field Manual

Part B – Additional Methods and Experimental Manipulations

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1.0 Introduction

This second part of the CANTTEX Field Manual details additional monitoring and experimental manipulations that can be added to a northern ecological monitoring site. Once the monitoring activities described in the first section of this manual have been implemented all CANTTEX participants are encouraged to engage in as much additional monitoring as is feasible with the resources at your disposal. The methods vary in the amount of effort and equipment required, but will all add valuable information on environmental change at your site. When selecting variables to measure, try to keep in mind the repeatability of measurements or observations and the likelihood that these observations will be maintained over a long period of time. See section 2.6 in the first part of the manual for a discussion of sample sizes. As with the monitoring described in the first volume of the manual, you are encouraged to contact CANTTEX (see section 10) if you have any questions or suggestions about the methods described here.

2.0 Biomass and Leaf Area Index

2.1 Introduction

Additional baseline data will be very useful for monitoring future changes in vegetation and environmental parameters. These data will provide context for the observations of biotic variables, and may also be repeated in future years to assess changes in baseline variables. To enhance the value of plant composition data, it is useful to know more specifically the quantities of biomass that are present and whether these quantities are changing over time. This involves point-framing a plot and then gathering and weighing the plants in that plot. Gathering data on plant biomass is painstaking and time consuming, but provides important information.

2.2 Field Methods - Biomass

If biomass collection is going to be undertaken it requires the establishment of new plots that will be used for this purpose alone because the vegetation in the plot is destroyed. The first step is to select plots that are comparable to your long-term monitoring plots and carry out point framing as described in Part A of this manual. Once point framing is complete, harvest all of the vegetation in the plot. This is best accomplished with hand-held garden shears ensuring that the vegetation is cut at the ground surface and that individual plants remain as intact as possible. Carefully put all of the samples in a plastic or paper bag and make sure they are labelled with the plot number.

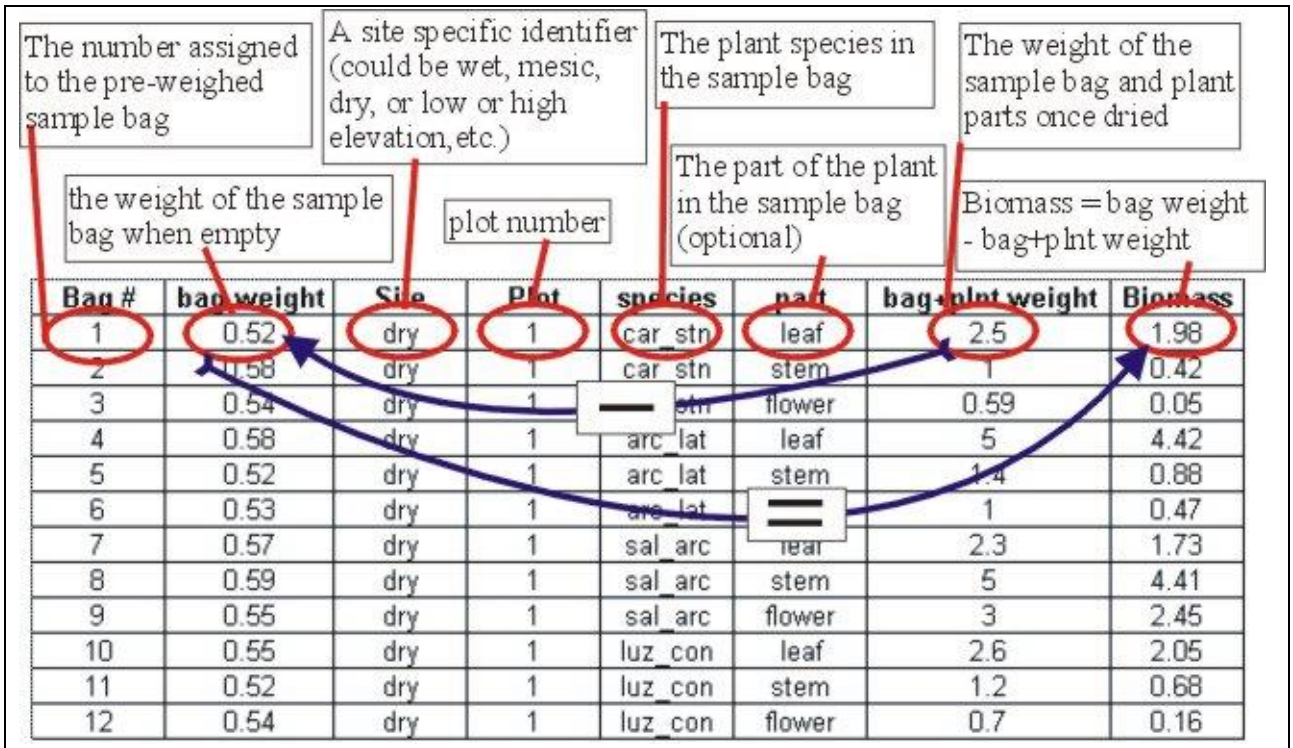
2.3 Laboratory Methods - Biomass

The samples should be sorted as soon as possible after collection to make identification easier. You should have paper bags that have been weighed and numbered. The plants harvested at each plot should be spread out on a table one at a time and sorted by species. If, when point-framing, you have kept track of what part of the plant was hit (leaf / stem / flower) you will want to further separate each species into these parts and weigh them separately. As you sort the plants, remove all soil and litter not attached to live plants. Have a separate pile for litter and weigh that as well. Place the biomass in the pre-weighed bags, air-dry them for at least 24 hours at 80°C. Allow them to cool to room temperature and then weigh them on a scale accurate to at least .01 grams.

2.4 Data Entry - Biomass

When you start sorting you should already have a list of numbered sample bags and their weight when empty (Fig. 1). As you fill each sample bag with the plant material, record the

characteristics of the sample (site, plot, species and plant part) in the table. After the samples have been dried and cooled, weigh them and record the weight in the "bag+plnt weight" column.



Bag #	bag weight	Site	Plot	species	part	bag+plnt weight	Biomass
1	0.52	dry	1	car_stn	leaf	2.5	1.98
2	0.58	dry	1	car_stn	stem	1	0.42
3	0.54	dry	1	[illegible]	flower	0.59	0.05
4	0.58	dry	1	arc_lat	leaf	5	4.42
5	0.52	dry	1	arc_lat	stem	1.4	0.88
6	0.53	dry	1	arc_lat	[illegible]	1	0.47
7	0.57	dry	1	sal_arc	leaf	2.3	1.73
8	0.59	dry	1	sal_arc	stem	5	4.41
9	0.55	dry	1	sal_arc	flower	3	2.45
10	0.55	dry	1	luz_con	leaf	2.6	2.05
11	0.52	dry	1	luz_con	stem	1.2	0.68
12	0.54	dry	1	luz_con	flower	0.7	0.16

Figure 1. Biomass data entry.

The biomass is calculated by subtracting the "bag weight" from the "bag+plnt weight" giving you the weight of the dried plant matter alone. This can be done automatically in your spreadsheet by writing a formula (e.g. =B2-G2) and copying it to the rest of the cells in the column. You should be very careful when using formulas in spreadsheets. Check to be sure that the cells you have designated are the correct ones and in the correct order. Try calculating a few of the values on a calculator to make sure the spreadsheet formula is giving the result you want. Once you have calculated the biomass values you should convert the formulae to numeric values. Consult the help files in your spreadsheet to find out how this is done. If you do not do this, you may end up with the wrong values if you do any manipulations in the spreadsheet that change the location of the data that are part of your formula (e.g. sorting, subtotals, etc.).

Once this is done, the data need to be reformatted so that they can be analyzed. To do this you must set up a table (Fig. 2) that is in the same format as the point-framing data (see part A of the manual section 4.3). All the columns (plant species and plant parts) that are included in the point framing data table should be included in the biomass table and vice-versa.

Plant species (e.g. *Carx stans* = car_stn)

Plant part (optional) (L = leaf, S = stem, F = flower)

Biomass, as calculated in the previous figure

Year	Site	Plot	car_stn_L	car_stn_S	car_stn_F	arc_lat_L	arc_lat_S	arc_lat_F
2003	dry	1	1.98	0.42	0.05	4.42	0.88	0.47
2003	dry	1	2.08	0.62	0.35	4.57	1.13	0.82
2003	dry	1	1.88	0.22	-0.25	4.27	0.63	0.12
2003	dry	1	2.03	0.52	0.2	4.495	1.005	0.645
2003	dry	1	2.1	0.64	0.37	4.59	1.15	0.84
2003	dry	1	2.28	0.62	0.15	4.67	1.03	0.52
2003	dry	1	2.08	0.62	0.35	4.57	1.13	0.82

Figure 2. Data format for biomass. It should correspond to the point framing data table.

2.5 Leaf Area Index

Leaf area index (LAI) is a measure of the leaf area over a standard ground surface area, usually 1 m^2. LAI is obviously related to *albedo*, but also provides a measure of the leaf area available for photosynthesis and evapotranspiration. LAI can be measured very laboriously by obtaining a sample of leaf areas of the major species, and using correlations with cover measured with the point frame. However, there are also sensors that can be used to obtain LAI directly, much the same as for *albedo*. Researchers interested in measuring LAI at their site should also contact Dr. Greg Henry for further information and advice. The names of some manufacturers of portable LAI meters are listed below:

ADC BioScientific Leaf Area Meters:
http://www.adc.co.uk/

LiCor Leaf Area Meters:
http://env.licor.com/Products/AreaMeters/area.htm

CID Inc. Leaf Area Meters:
http://www.cid-inc.com/products/

3.0 Annual Environmental Monitoring

3.1 Climate

All CANTTEX sites are strongly encouraged to collect accurate and detailed meteorological data. This is best accomplished with the installation of an automatic weather station (autostation). This consists of a datalogger recording hourly or daily means of temperature, relative humidity, wind speed and direction, and global radiation. The design and installation instructions are thoroughly described in the ITEX Manual (Molau and Mølgaard, 1996) and anyone interested in setting up an autostation should contact Dr. Greg Henry for more assistance (see Contact Information).

3.2 Snowdepth and Date of Snowmelt

Though the focus on climate change tends to be on temperature, changes in precipitation regimes will likely have as strong effects on ecosystems. These changes may include increases or

decreases in winter snowpack and changes in the timing of snowmelt. The amount of snow and the date of snowmelt are important indicators of climatic conditions and also have a large effect on the growth of plants during the summer. The date of snowmelt is essentially the start of the growing season. If your site is consistently occupied during the snowmelt period it would be very useful to monitor snowdepth and melt phenology.

In particular, knowing the date of snowmelt for each of your plots is very important information to correlate to changes in vegetation phenology and composition. To get an idea of the pattern of snowmelt it is important to know how long it takes the snow to melt and when this occurs. For this reason, there are three dates to record for general snowmelt at your site:

1. *10% melted*. This marks the initiation of the snowmelt period as temperatures are regularly above 0° C and approximately 10% of the snow has melted.
2. *50% melted*. Roughly half the winter snow cover has melted.
3. *90% melted*. Only late-lying snowbanks remain with the rest likely to melt within one week.

If there are no observers at the site during the snowmelt period there is another way to accurately estimate snowmelt date. If you have an autostation or dataloggers recording plot level data, ensure that one of the temperature sensors is close to the ground surface. In the temperature record there will be a sharp increase in temperature (likely from negative values to positive) when the sensor is released from the snowpack. Make sure to record this date somewhere, such as the snowmelt date column of plant phenology sheets.

More detailed information on the snowpack is also valuable as this is recognised as one of the most important variables that is likely to be influenced by global change. A snow course is a permanent transect of snow depth and density measurements. Standard snow survey methods are found in Goodison *et al.* (1981) and are summarized here, but there are also many other detailed references on these methods. The simplest snow survey will simply be a series of snow depth measurements that can be taken with a rod that has graduated measurements on it. To maintain a proper snow course you will want to obtain a snow sampler. This consists of an aluminium tube about 3.5 cm in diameter and 75 cm long with a sharpened edge on one side to cut through ice layers and graduated markings on the side to determine snow depth. The equipment also includes a spring scale and cradle for measuring the weight of the tube and snow sample.

A snow sampler can be purchased from:

Leupold and Stevens, Inc.
P.O. Box 688
Beaverton, Oregon
97075
United States
(503) 646-9171

Select a location for your transect that is comparable to the location of your vegetation monitoring plots and ensure that the location of specific samples is not near ground irregularities such as boulders. Slightly sloping terrain is preferred over flat ground but avoid setting up a snow course on a steep slope. A snow course doesn't have to be a straight line but this will make sampling somewhat easier. The transect should be 100 – 200 m long and have at least 10 samples taken along its length. It is important that the samples be taken in approximately the same place each time to ensure comparability over time. You should install permanent markers (stakes driven into the ground that will be visible when the snow is at its maximum) at both ends of the transect and record specific instructions about where samples are taken along the transect. A simple map (Fig. 3) would be extremely helpful to ensure that future monitoring is consistent.

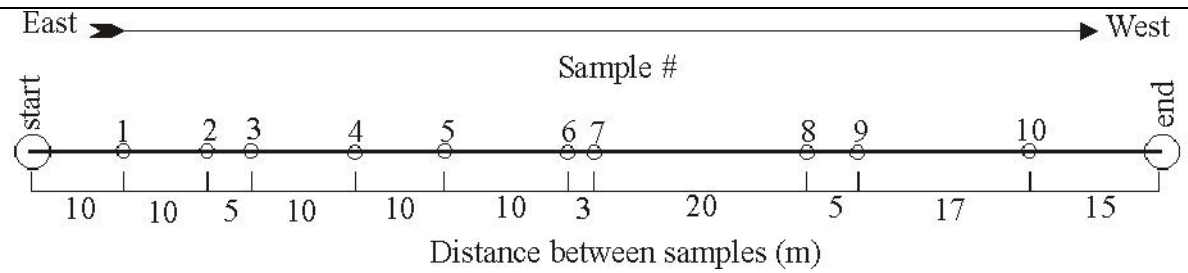


Figure 3. Sample snow course map. The start and end points of the transect should be marked with a permanent post and the geographic coordinates recorded somewhere to ensure that the transect can be found if the post is not visible.

Lower the sampler vertically in to the snowpack in an undisturbed area, twisting it if necessary to cut through any ice layers. It is helpful if the sampler is driven into the soil as the soil plug will prevent the snow from sliding out of the tube when it is raised. Pull out any soil from the bottom of the tube and place it in the cradle. It's important to balance the tube in the cradle so it doesn't tip and lose the snow core. Record the weight in your field data form (example in Appendix A). The spring scale should be calibrated to read in water equivalent depth units (the depth of water that would result if the snow melted). If not, you must know the inside radius of the tube to calculate the volume of snow based on the depth. Then, empty the snow from the tube and weigh the tube alone. The difference between these two values is the water equivalent of the snowpack. Also, make general observations about the snow conditions (e.g. are there ice layers in the snow? Is there a crust on top? What is the approximate elevation of the snowline if you are in an alpine area?, etc.). A sample data entry form is provided in Appendix A.

3.3 Active Layer

The active layer is the seasonally thawed surface layer above the permafrost. This is the soil layer of tundra ecosystems. The depth of the active layer and the timing of its development over the growing season are important variables for climate change detection and have direct consequences for arctic flora and fauna.

The use of a narrow diameter steel probe to measure the distance to the top of the permafrost is the most inexpensive and commonly used method. However, in areas where the soil is coarse or the active layer is greater than 1.5 m thick, the use of other methods such as monitoring sub-surface temperature profiles or the use of frost tubes should be considered. Described below is the method using a probe. For more information on other methods contact Dr. Antoni Lewkowicz (Dept of Geography, University of Ottawa, alewkowi@uottawa.ca).

Active layer measurements should be made in the vicinity of each of your plots and at an associated 100 m^2 grid. The grid should use the systematic stratified unaligned design with 100 points. The layout of the grid is provided with the associated sample forms in Appendix A. Where possible make a set of measurements at the peak of the growing season and again at the end of the growing season. The probe should be made of metal and be less than 1 cm in diameter with 1 cm increments marked on the side. Push the rod vertically down into the soil until it reaches the ice surface. With some experience you will be able to identify the difference between the permafrost surface and a rock buried in the soil. If you encounter rocky soil, move to the southwest in 1 m increments until you are able to make a reliable measurement. Remove the rod carefully to avoid disturbing the soil excessively.

3.4 Ice Phenology

As with snowmelt, the timing of the freeze-up and break-up of river, lake and sea-ice are important indicators of climate conditions and have a strong influence on local ecosystems. Monitoring the ice phenology of local water-bodies will provide valuable long-term datasets for assessing the impact of climate change.

Lake Ice

An appropriate lake to monitor freezing and thawing should be at least 0.5 km^2 in area and at least 5 m deep at its deepest point. Choose a lake that you will see on a regular basis while at your study site. Like snowmelt, the freeze-up and break-up of lake ice takes place over a period of time and there are a few dates that you can record:

Freeze-up:

1. *Ice formation on shore* – permanent ice has formed along the shoreline
2. *Ice cover on bays* – bays completely covered with ice
3. *Completely frozen* – the entire surface of lake covered with ice

Thaw:

1. *Open water on shore* – roughly half of the shoreline showing open water
2. *Open water off shore* – several openings are visible in the lake ice
3. *Ice in movement* – the ice is broken-up and floating in blocks in the water
4. *No ice visible* – all of the lake ice has melted

Sea Ice

As for monitoring lake ice, choose an area of sea ice that you will see on a regular basis and over a number of years. Please record the name of the water body (i.e. Lancaster Sound, Hudson Bay, Mackenzie Bay etc.) and the type of water body (i.e. open ocean, inlet etc.) The dates you record for the sea ice are the same as those for lake ice.

3.5 Albedo

The reflectivity of the surface (albedo) for solar radiation is a major feedback between terrestrial ecosystems and the atmosphere. Albedo is the ratio of reflected to incoming solar radiation. A dark, dense plant canopy, such as closed boreal forest, absorbs significantly more solar radiation than open, low plant canopies, such as high arctic tundra. Hence, forests provide much more

sensible heat to the atmosphere than tundra ecosystems. Increasing the density and cover of plants will affect the energy balance of the surface by decreasing the albedo.

Measurement of albedo is relatively simple, but requires a radiation sensor, such as a pyranometer, or albedometer. These instruments are usually included in an autostation and are connected to a datalogger to record the measurements. However, hand held measurements can be made over individual plots. Periodic measurements (e.g. every 3-5 years) associated with the collection of plant community composition data made over individual plots will provide a record of changes in the albedo of the surface in relation to changes in the structure of the plant community of the site. Researchers interested in measuring albedo as part of their monitoring program should contact Dr. Greg Henry for more advice and recommendations (see section 10 – contact information).

3.6 Animals and Insects

Animals and insects are also highly sensitive to their environment and are good indicators of changes to climatic patterns. There are many species that have annual hibernation or migration patterns that are easily observed simply by recording the date at which an animal or insect is first seen. The forms in Appendix B list many species for which you can simply record their first observed appearance. If you are not sure about the identification of a given species consult a field guide.

4.0 Individual Plant Measurements: Phenology, Growth, and Reproduction

4.1 Introduction

In addition to being able to produce more seeds and flowers, warmer and longer growing seasons will have an effect on plant phenology and growth of the vegetative parts of plants. Again, if some species are better adapted to take advantage of warmer and longer summers, such a competitive advantage might result in changing the relative abundance of species in a given area over time. Plant phenology has been shown to be sensitive to changes in summer warmth and growing season length. The more simple PlantWatch methodology for monitoring spring phenology is described in Part A of this manual. Detailed monitoring of plant phenology is labour intensive and requires regular visits to monitored plots throughout the growing season. Remember to record the date of snowmelt for each plot if you are monitoring your site in the spring. The establishment of a phenological observation program should only be considered if it can be maintained over several years. Other growth and reproductive measures are made once, at peak season, and many of these can be carried out in a matter of minutes for each tagged plant.

4.2 Field Methods

When selecting the plant species that you will monitor, it is important that you use the dominant species at your study site and try to monitor a species belonging to each of the major growth forms (forb, graminoid, dwarf shrub, etc.). It is not necessary to monitor each of the species described below. If there are species that are abundant at your site but not included in this manual you may develop your own set of reproducible measurements or observations. You are encouraged to contact CANTTEX with questions or suggestions (see Section 10 - Contact Information).

The vegetative growth measurements should be made at the same time as the reproductive growth measurements, but these must be made on individual plants. There are some exceptions to this – for example male willow catkins fall off earlier in the season once they senesce and should be measured before this occurs. These are single point in time measurements made once each year at approximately the same date every year. It is best if you make the measurements on the same plants within your plots from year to year (see **4.3 *Selecting and Tagging Plants*** below). The more vegetative growth measurements you can make the better, but concentrate on the dominant species in your study area. You should monitor at least **20** individuals of each species being studied dividing these among all your plots. Phenological observations will likely be made on the same species for which you are making vegetative and reproductive growth measurements. Select the dominant species at your study site and try to pick plants from as many different growth-forms / functional groups as possible.

4.3 Selecting and Tagging Individual Plants

Many of the monitoring methods described in this manual require the selection and monitoring of individual plants. The best way to select a plant depends on the growth form of the species. For some species, identifying an entire individual plant is relatively easy and for others it is quite difficult. Plants should be selected at random to avoid any bias in the sample. Ideally this can be done by using a random number table (Appendix D) to choose a random number of steps, walking the designated number of steps, and selecting the plant that is closest to a pre-determined point on your foot or distance and direction from your foot on the last step. A convenient way to select plants within a plot is to divide the plot in four and choose a plant at random in each quarter to monitor. Any other method of selecting the plants at random is acceptable.

For cushion plants or mat-forming plants (e.g. *Dryas* spp., *Silene* spp.) or those with a tufted growth form (such as some grasses and sedges) an entire clone should be used as the monitoring unit. For mat-forming plants in which individual clones are impossible to discern, an area of the plant should be defined for monitoring and marked with wooden stakes and string. This area will normally be 100 cm^2, but might be the entire plot. For woody plants such as *Salix* and *Cassiope* individual branches should be used as the monitoring unit. As twigs grow and branch, tags should be moved up the stem every year or two to ensure there is no confusion about which stem is being monitoring. Issues specific to each species are described in the methods for monitoring that species.

If you are maintaining a plant phenology monitoring schedule or making reproductive or vegetative growth measurements on individual plants you will need to mark each of the plants with a tag to identify them. There are several types of plant tags that can be purchased from forestry supply retailers, but the most common is a metal tag with a hole through it and a piece of soft wire that can be wound around a branch or stem. For cushion plants, use a nail or wire to fix the tag to the ground adjacent to the plant. The tag must have a unique identifying number within the plot and it helps to identify the site and whether the plot is a control or treatment in the number or code. You should try and minimise the disturbance caused by the placement of the tag (e.g. not shading the plant or twisting the wire too tightly around a branch or stem). It is helpful to record the general location of each plant within the plot in your datasheets (i.e. in the northwest corner of the plot). It is often surprisingly difficult to locate plants in densely vegetated plots and

this will shorten the search. Birds and rodents are known for removing tags so make sure they are attached securely without damaging the plant.

Though all monitoring activities associated with these plants should be non-destructive, occasionally tags will go missing or the plant that you have chosen to monitor will die or be eaten by herbivores. When this occurs, select a new plant at random and install a new tag. Then, mark *"re-tagged"* on the datasheet and the date the plant was retagged. This will be essential when analysing the data to separate changes due to changing climate from changes resulting from monitoring a different plant. To make it less confusing, new recruits should be assigned a new unique number so that someone looking at the data does not confuse it with the previously monitored plant.

4.4 Individual Plant Descriptions

What follows is a list of the plant species that are currently being monitored at CANTTEX sites. Each description includes one or several common names, the scientific name of the species, the growth form of that species and a description. If there is any uncertainty about the identification of a plant a field guide or flora should be consulted. The Flora of the Canadian Arctic is available on the internet at:

http://www.mun.ca/biology/delta/arcticf/

To use the flora you must first download and install the associated software from:

http://biodiversity.uno.edu/delta/win32/intk32.exe

Alpine Bistort / Viviparous Knotweed
Bistorta viviparum (L.) S.F. Gray (formerly: *Polygonum viviparum* L.)
Growth form: Forb

The alpine bistort has long narrow leaves growing up from the base of an erect stem that has small white or pink flowers on top. Below the flowers are small round bulbils that look like little seeds growing along the stem. It flowers early.

Tagging

For the alpine bistort an individual plant is the observational unit. This plant is a delicate forb with a solitary stem so the tag must be affixed to the ground adjacent to the stem.

Quantitative Observations

Q1. *Length of the flowering stalk*: At full flower, measure the height of the stem from the ground to the top of the flower stalk (in mm).

Q2. *Length and Width of largest leaf*: The length is measured from the base to the tip of the leaf not including the leaf stalk (petiole). The width of the largest leaf should be measured at it's widest point (in mm).

Q3. *Number of leaves per individual:* For each tagged and monitored plant or for five random plants in each of your plots, count the number of live green leaves. Be sure to record evidence of herbivory on any of the plants.

Q4.*Number of bulbils per shoot*: count the number of bulbils on the tagged flower shoot.
Q5.*Number of flowers per shoot*: count the number of flowers on top of the tagged shoot.

Phenology

P1. *First Flower*: the date that you see the first flower open on the top of the stem;
P2. *Bulbil Shed*: the date that the bulbils that form below the flowers drop off when touched.

Arctic White Heather
Cassiope tetragona L. D. Don.
Growth form: evergreen dwarf shrub

Arctic white heather is an ericaceous shrub with scale-like evergreen leaves. The flowers are white and bell-shaped dangling from red pedicels that extend out from between the leaves at the tips of the branches or stems.

Tagging

When monitoring *Cassiope tetragona* a single branch is identified and tagged. The tag must be attached to the chosen branch with wire or string, but be sure not to tie it too tight and make sure the tag is not shading the monitored branch. In the spring, a very small drop of liquid paper is used to mark the previous year's leaves (apical meristem) before they open. Be sure not to cover the tip of the branch as this could prevent it from opening and potentially kill the branch. Just place a small drop on each of the previous year's leaves.

Quantitative Observations

Q1.Length of annual growth increment: At the end of the season measure the distance from the tip of the previous year's leaf to the tip of the current year's stem growth (in mm) (Fig. 4). Be careful not to handle the branch any more than is necessary to obtain the measurement.
Q2.Number of Flowers: The total number of mature flowers on the tagged branch.
Q3.Number of Capsules: The total number of immature fruit on the tagged branch.
Q4.Number of Mature Fruit: The number of mature fruit on the tagged branch.

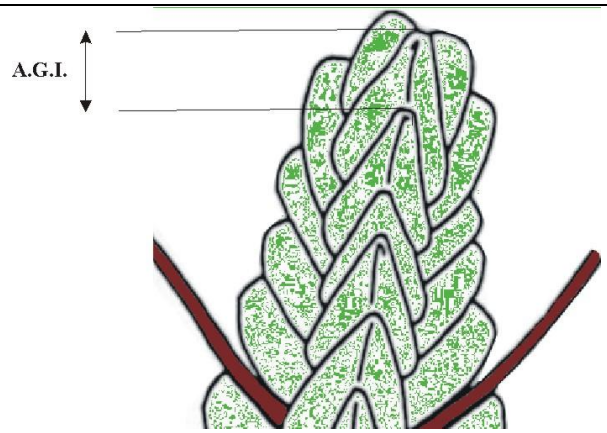


Figure 4. Annual growth increment (A.G.I.) for Arctic White Heather.

Phenology

P1. *Flower Bud Break*: as soon as any of the red pedicel (flower stem) is visible as it emerges from between the green leaves.

P2. *Full Flower*: the date that the white bell-shaped flowers are fully formed and have opened.

P3. *Immature Fruit*: after the petals fall off what remains should be a small hard fruit on a curved pedicel dangling like the flowers.

P4. *Mature Fruit*: as the fruit matures the pedicel straightens and holds the fruit up in the air.

Arctic Avens (White Dryas) / Mountain Avens
Dryas integrifolia Vahl. / *Dryas octopetala*
Growth form: evergreen dwarf shrub

Arctic and mountain avens are members of the rose family and grow in low mats along the ground. Arctic avens has small needle like leaves and mountain avens has slightly larger leaves that are more deeply lobed than the mountain avens. The solitary flowers are usually white but occasionally yellow. After the petals fall off the seeds develop with their long feather-like tails in a tight swirl.

Tagging

When possible the unit of measure for Dryas is an entire clone. In these cases a tag should be affixed to the ground adjacent to the plant. Where large mats of Dryas are found, select an area 10 cm X 10 cm to monitor and mark it with skewers and string. Try to minimize the disturbance caused by labelling the plant. WARNING: Avoid male-sterile clones in gynodioecious populations of *D. octopetala*; these have flowers with the androecium reduced to a ring of 1-2 mm high, brownish staminodes.

Quantitative Observations

Q1. *Total number of flowers per clone*: It is valuable to record this number at each of the stages – the number of buds, the number of mature flowers, the number of swirls and the number of aborted flowers (ones that did not produce a swirl). If the clone is not easily identifiable, you can do this on an area basis but try to keep it constant from year to year.

Q2. *Length of longest new leaves:* At the time when the petals are being shed, use a plastic ruler to measure the length of the five longest green leaves. The petiole is not included.

Q3. *Size of the clone:* If the *Dryas* clone can be clearly delimited, measure the diameter of the clone at it's widest point and then again at a 90 degree angle to that measurement. Multiplying these values together will give a rough estimate of the area of the plant;

Q4. *Height of flowers:* Measure the height from the base of the flower stalk to the base of the flower (in mm). Note whether the flowers are fertilized or not. One way to do this in the field notes is to record all the heights of fertilized flowers first, followed by all the un-fertilized plants in parentheses.

Q5. *Circumference:* If the Dryas clump is readily discernable, use a piece of string to trace the circumference of the clump then measure the length of the string.

Phenology

P1. *New Leaves*: Dryas is an evergreen plant so leaves last for several years. Look carefully among the existing leaves for the first appearance of new small green leaves and record the date.

P2. *Flower Bud Break*: Record the first date any of the white petal is visible as it emerges from the flower bud.

P3. *Full Flower*: Record the date that the first flower is completely open.

P4. *Swirl*: After the petals fall off, the flower will produce a swirl or twist of feather-like seeds. Record the date when the filaments are clearly twisting.

P5. *Dispersal*: Record the date when the swirl has expanded and the seeds are beginning to be released.

Arctic Poppy / Keele's Poppy
Papaver radicatum (*lapponicum*) / *Papaver keelei*
Growth form: forb

Arctic poppies are very distinctive plants. Generally, many flower stems grow out of a bunch of basal leaves that are pale green, deeply lobed and covered with hairs. The flowers are large, usually yellow, but sometimes white with a barrel shaped seed-capsule in the centre. The end of the capsule has radiating lines of black hairs on it. *Papaver* continues to produce new flower shoots throughout the growing season.

Tagging

The unit of observation for poppies is an individual plant. Each year new flower shoots grow out of the base of the plant, so the best way to mark them is to affix a tag to the ground adjacent to the plant.

Quantitative Observations

Q1. Flower heights: At the peak of the growing season measure the heights of the tallest mature flower shoot from the ground to the base of the flower. *Note if height measurements are for flower buds, opened flowers, or fruits.*

Q2. Number of flowers: Count the number of shoots in each clone.

Phenology

P1. *First Flower Bud Visible*: Record the date when the first flower bud is visible as it begins to be lifted upwards on the curved stem.

P2. *First Mature Flower*: Record the date when the first fully open flower is visible on an upright stem.

P3. *Flower Senescence*: The date that the first flower looses its petals and the seed capsule turns from green to brown.

P4. *Mature Capsules:* The date that the first capsule opens, looking like the spokes of a bicycle wheel.

Arctic Willow / Snowbed Willow / Herbaceous Willow / Net-Veined Willow
Salix arctica Pall. / *Salix polaris* Wahlenb. / *Salix herbacea* L. / *Salix reticulata* L.
Growth form: deciduous dwarf shrub

There are 42 species of willow in the Northwest Territories. They are often difficult to identify because they can hybridize (cross breeding between two separate species). The dwarf shrub arctic willow has a woody stem that grows along the ground bearing many oval to almost round deciduous leaves. In the spring both leaf and flower buds begin as soft furry catkins. Willows have separate male and female plants. When in full flower the male plants have yellow pollen on the stamens that protrude from the catkins and the female plants have forked red stigmas.

Tagging
The unit of observation for willows is an individual branch. The tag should be attached to the chosen branch with wire or string, but no so tight as to damage the plant. It is not always possible to determine whether a branch of Salix being tagged is vegetative or reproductive, or whether it is male or female. To ensure adequate replication for some of the observations it might be valuable to tag more *Salix* branches than you do for other species. For each tagged plant record whether it is male, female, or vegetative (no flowers on the branch).

Quantitative Observations

Q1.Annual growth increment: At the end of the branch of *Salix spp.* the annual growth increments can be identified by the last bud scar at the tip of the stem. It is the last in a series of lips or ridges where each year's new growth has emerged out of the previous year's section of stem (see Fig. 5 below). At peak season, measure the length of the current year's growth from this lip to the tip of the branch (to the nearest mm).

Q2.Length of longest leaf: Measure the length of the longest leaf on each tagged branch from the base of the petiole (stem that attaches the leaf to the branch) to the tip of the leaf (in mm).

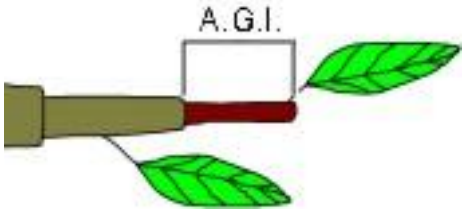


Figure 5. Sketch of the annual growth increment to be measured for *Salix spp.*

Q1.*Number of Catkins:* Record the total number of flowering catkins per monitored branch.

Q2.*Total Mature Catkins*: Record the total number of mature catkins per branch. Count the number of catkins produced on all the green branches coming off last year's stem (usually there will only be 0-2; if you count more, double-check that you are looking at the right stem sections).

Q3.*S. arctica/reticulata*: *Catkin Length*: length of mature catkins from axil of subtending leaf to the top of the catkin (in mm). Make sure to identify male and female catkins. Measure all the female catkins on the tagged branch from where the catkin joins the branch to the

tip of terminal pistil. This should be done at the end of July or early August (record the exact date).

Q4. *S. herbacea / polaris: Number of Capsules*: Record the number of capsules per catkin.

Q5. *Length of the Largest Leaf*: Select the largest leaf on the tagged branch and measure the length from the base of the petiole to the tip of the leaf.

Phenology

P2. *Catkin Development*: Record the date that the soft hairy catkins are released from the protective brown sheaths.

P3. *Leaf Bud Break*: The leaves on this species of willow will burst out of buds that have overwintered. The buds appear as brown pointed structures and the leaves will look a fuzzy grey-green as they push out. Record the date that you can see a leaf protrude from any of the bud scales on the tagged branch. Often the catkins arise from this same bud after the leaves have expanded slightly. Check to make sure there is green, otherwise, it may be a catkin emerging.

P4. *Full Flower*: Record the date that the flowers of the male plants have yellow pollen on the stamens and the red pistils are visible on the female catkins.

P5. *Mature Fruit*: Record the date than the ovaries of the female plants swell producing capsules.

P6. *Dispersal*: Record the date that the capsules of female plants open releasing the woolly seeds.

P7. *Leaf senescence*: Record the date that the leaves show the first signs of changing colour at the end of the growing season.

P8. *Last Green Leaf Senescing*: Record the date when the last green leaf on the tagged branch begins to change colour.

Cottongrass
Eriophorum vaginatum L. / *Eriophorum angustifolium* Honck.
Growth form: graminoid

Cottongrass species are sedges that tend to grow in wet meadows and along the edges of ponds. The live leaves of *Eriophorum* often have a deep purple tip or strip if the tip has senesced. After flowering, these plants produce seeds in bunches like tufts of cotton or wool. *Eriophorum vaginatum* is one of several species of cotton grass that form tussocks in the low arctic tundra. Tussocks appear as large raised "clumps" of vegetation ranging in size from 30cm to 150cm. When in bloom, cottongrass has white fluffy heads that wave in the breeze. Cotton grass is not really a grass but a sedge.

Tagging

The unit of observation for Cottongrass is an individual tussock. Tagging is best accomplished by affixing a tag to the ground adjacent to the plant.

Quantitative Observations

Q1. Diameter of tussock: Measure the diameter of the clone at it's widest point to the tips of the leaves and then again at a 90 degree angle to that measurement. Multiplying these values together will give a rough estimate of the area of the plant;

Q2. *Number of flowering stalks:* Count the number of flowering stalks in each monitored tussock or in five randomly chosen tussocks in each of your plots;

Q3. *Leaf Length:* Measure the length of the longest leaf on each tagged plant.

Q4. *Live and Senesced Leaf length:* Measure the length of the ten longest leaves from the tip of the sheath or from the ground (be consistent) to the apex of the leaf (in mm) and to the end of the live part of the leaf. Measure ten leaves on each tussock that appear to be long relative to the rest of the leaves. Don't worry too much about getting the ten longest. It is sufficient to choose leaves that appear to be long. Take this measurement at the end of July or early August remembering to record the date.

Q5. *Number of flower stalks*: *Eriophorum* tussocks usually produce several inflorescence stalks. However, some tussocks may not produce any. Count the number of inflorescence stalks as soon as all have displayed their anthers.

Phenology:

P1. *First New Leaf Visible*: The cotton grass tussocks often have a number of different species growing on them, including other sedges. Cotton grass leaves are difficult to distinguish from *Carex* leaves. The easiest way to tell them apart is that *Carex* leaves come out of the ground in a sheath, whereas *Eriophorum* emerges as single leaves. New *Eriophorum* leaves from this year will be entirely green. A leaf that is green on bottom and brown on top is a leaf that has overwintered. Record the date when you see an entirely green leaf emerging from the tussock.

P2. *Flower Bud Break:* Inflorescence buds will emerge from the tussock as a brown torpedo shaped structure wrapped in a sheath. It appears dry and can look like something left over from the previous year. The sheath will open to reveal a soft, silvery bud that looks somewhat like a pussy willow. Buds often emerge before all the snow is gone. Record the date that you notice the buds with the sheaths still covering them.

P3. *Mature Flowers*: Once the grey inflorescence head appears it will gradually dry and become fluffy. Within a day or two whitish yellow stamens will extend out. The stamens will mature until the anthers are yellow with pollen. Record the date when the inflorescence head turns bright yellow. After the yellow anthers have been displayed they will turn brown and wither somewhat. If you notice a bud after it has turned brown you have missed the exposed anther stage. If this happens write down the date you first notice the bud and the stage it is at.

P4. *Seed Dispersal*: As the seeds develop, the flower head becomes a tuft of wool-like seeds. When the seeds are mature the fluff will pull away and be carried off on the wind. It will be obvious when this happens as you will see empty areas on the inflorescence head. Record the date when there are noticeable empty areas on the inflorescence head where seeds have blown away.

Diapensia
Diapensia lapponica L. (including ssp. *lapponica* and ssp. *obovata*)
Growth form: cushion plant

Diapensia forms convex cushions with tightly packed pointed leaves and has large white flowers close to the cushion on peduncles which elongate with maturity.

Tagging
Individual plants (independent of other plants) should be selected and marked with a tagged nail beside the cushion.

Quantitative Observations
Q1: *Size of Cushion*: Measure the width of the cushion at its widest and narrowest point (in cm).
Q2: *Number of Flowers*: The total number of mature flowers produced per tagged cushion.
Q3: *Number of Capsules*: The total number of capsules produced per tagged cushion.

Phenology
P1: First Open Flower: Record the date when the 1st flower on the cushion is completely open (i.e. when stamens and stigma can be observed, this is the stage pollinators visit)
P2: First Capsule Cracks: Record the date when the capsule first cracks open at the top.

Lingonberry (Mountain Cranberry)
Vaccinium vitis-idaea L.
Growth form: evergreen dwarf shrub

Lingonberry is a low shrub with small hanging bell-shaped flowers that range from white to pale pink in colour.

Tagging
The unit of observation for Lingonberry is an individual plant. Tagging is best accomplished by affixing a tag to the ground adjacent to the plant.

Quantitative Observations

Q1.*Annual growth increment:* Cranberry shoots will produce new growth that has light green leaves with reddish tips. New growth can be distinguished from old by the remains of the bud scales on the shoot (where the growth started from last year's bud). The new growth is measured from the remnants of the bud scale to the terminal bud (not the tip of the leaves).This can most clearly be seen for V. *vitis-idaea* by looking at the underside of the stem – look for small bud scars and a change to lighter green in stem colour. Current year stems of V. uliginosum will be green, not brown or grey.

Q2.Number of berries on a branch or plant (record which).

Q3.Number of Flowers: *Vaccinium* will produce anywhere from one to six flowers per shoot. They will open at slightly different times. Some may have dropped their petals before the last flower opens so it is best to keep an ongoing record of number of flowers as they open. Record Q1 as soon as all of the flowers have opened. Another benefit of keeping a running total of flowers is that they may get grazed by herbivores. In that case we still want to know how many there were before they were eaten!

Q4.Number of Fruit: The number of fruit on the tagged shoot can be recorded after all flowers have dropped and fruit begins to turn red. The number of fruit can be compared to the number of flowers to give us a measure of reproductive success (how well the plant did in producing seeds compared to the amount of investment made in producing flowers).

Phenology

P1. *Flower Buds Visible*: *Vaccinium*, like many arctic flowering plants, produces flower buds the year prior to the current season. Therefore, as soon as the snow melts you may notice flower buds right away. They will look red or pinkish and will be evident at the end of the shoot. It may be hard to distinguish between a flower bud and a leaf bud. Be patient, it will become evident. Look for buds within a 15 to 20cm radius of the tag. When one shoot on the marked tussock shows a bud that is noticeably pink record the date. You will need to tag the *Vaccinium* shoot on the marked tussock once you have recorded the date of the first visible flower buds. A piece of flagging tape that has been torn in half along its length works well. Just tie it around the base of the shoot - but not too tight! Monitor only this tagged shoot for the rest of the summer even if it doesn't seem representative of the rest of the plants on the tussock (you will want to note this on the datasheet, however).

P2. *First Flower Open*: *Vaccinium* flowers typically open soon after *Ledum* flowers open. They are delicate bell-shaped flowers that hang down from the shoot. It can be difficult to see when the buds are actually open. You may have to carefully tilt the buds up towards you to determine if there is any open space between the petals at the tip of the bell. Record the date when you can see space between petals of any flower on the tagged plant.

P3. *First Flower Shed*: The petals of the *Vaccinium* flower are fused so the entire flower is shed at once. Flowers, however, will not be shed all at once but will be dropped over a number of days. Record the date when the first entire flower drops off the marked shoot.

P4. *Last Flower Shed*: Record when the last flower drops off the marked shoot.

P5. *First Fruit Visible*: Fruit first became visible as small 2mm berries about 7 – 14 days after the flowers open. They are green but will turn red as the season progresses. Record the date as soon as there is a noticeable swelling where the flower was dropped. It is important to wait for the swelling, as some flowers will not be fertilized and will not form fruit.

Moss Campion
Silene acaulis L.
Growth form: Cushion plant

Moss campion forms small round mats with tightly packed pointed leaves and small purple (occasionally white) flowers that sit tight to the plant.

Tagging

Individual plants of moss campion are easily identified and these are the units of observation. The tag should be affixed to the ground adjacent to the plant.

Quantitative Observations

Q1. Size of cushion: Measure the width of the cushion at its widest and narrowest points (in cm).

Q2. Number of flowers: Count the total number of flowers produced on each tagged plant

Q3. Number of capsules: Count the total number of capsules produced on each tagged plant.

Phenology

P1. *First Open Flower*: Record the date when the first small pink flower is completely open.

P2. *First Capsule Cracks*: After the capsules form they will crack open at the top to release the seeds inside. Record this date.

Moss Heather
Harrimanella hypnoides(L.) Coville (syn: *Cassiope hypnoides* (L.) D.Don)
Growth form: evergreen dwarf shrub

Moss heather is an ericaceous shrub with scale-like evergreen leaves. The flowers are white and bell-shaped dangling from red pedicels that extend out from between the leaves at the tips of the branches or stems.

Tagging
Tagging this species is the same as for arctic white heather. For this and other clonal species, you should NOT pull the branch out from the clone when tagging. Small lengths of coloured string/wool also work. You may also choose to delineate an area (10 cm X 10 cm) using string and wooden stakes or nails in the ground marking the corners of the area.

Quantitative Observations
Q1: *Number of Flowers*: The total number of mature flowers on the tagged branch.
Q2: *Number of Capsules*: The total number of immature fruit on the tagged branch.
Q3: *Number of Mature Fruit*: The number of mature fruit on the tagged branch.

Phenology (differences in wording in italics)
P1: Flower Bud Break: as soon as any of the red pedicel (flower stem) and small pinkish bud are visible as it emerges from between the green leaves at the tips of the branches.
P2: Full Flower: the date that the white bell-shaped flowers are fully formed and have opened (i.e. even the smallest opening is visited by pollinators).
P3: Immature Fruit: after the corolla falls off, what remains should be a small hard fruit on a curved pedicel dangling like the flowers. (i.e the corolla starts turning yellow from the base, touch the corolla with a pencil as it tends to persist at this stage)
P4: Mature Fruit: as the fruit matures the pedicel straightens and holds the fruit up in the air.

Mountain Sorrel
Oxyria digyna (L.) Hill
Growth form: forb

Mountain sorrel has stout rounded leaves that grow on long stems originating from the base. The seeds are red with a fringe or wing around them and are attached along the stem.

Tagging
Individual Sorrel plants are ideally the unit of observation. However, in some cases it is easier to stake out a clump if individual plants are too difficult to identify. The stems are fragile so you shouldn't wrap string or wire around them, but affix the tag to the ground adjacent to the plant.

Quantitative Observations

Q1. Number of inflorescences per clone: Simply count the number of flowering stalks per clone (0, 1, 2, etc.);

Q2. Length of inflorescence stalk: At full flower, measure the height of the tallest flowering stalk from ground to the base of the raceme (in mm);

Q3. Width of largest leaf: Measure the width of the largest leaf at its widest point (in mm).

Q4. Number of mature fruits per plant: Count all of the mature fruits from each tagged clone and harvest in paper bags.

Q5. Mean fruit weight: Weigh all the fruits from each clone, dried at room temperature, as a single batch, and calculate the mean fruit weight (to an accuracy of 0.1 mg).

Phenology

P1. *First Flower Open*: Record the date when the red stigmata are visible inside open flowers.

P2. *First Seed Dispersal*: The date at which seeds drop off easily when touched.

Purple Saxifrage
Saxifraga oppositifolia L.
Growth form: cushion plant or mat-forming forb

Purple saxifrage is one of the most common plants in arctic and alpine tundra. It grows low to the ground often in a tight rosette with pink to purple flowers having five petals. It flowers extremely early, often within ten days of snowmelt. During fruiting the pedicel (flower stem) extends upward with the two fruits, which eventually split open, releasing the seeds.

Tagging

How you tag purple saxifrage will depend on the growth habit of the plant at your site. In cases where it makes a tight rosette such that an individual plant is easily recognizable this is the unit of measure. In some cases mats of saxifrage will be present in which case you will have to pick a 10 cm X 10 cm patch to observe. Use wooden skewers to mark the four corners of the patch being observed and attach string between the skewers to delineate the area. In both cases the ID tag is affixed to the ground adjacent to the plant or patch.

Quantitative Observations

Q1. Size of plant: Measure the diameter of the plant at its widest point and at 90 degrees to that measurement (in cm).

Q2. Vegetative growth: Once *Saxifraga oppositifolia* has gone to fruit the pedicels will extend upwards holding the fruit well above the plant cushion. Using a small plastic ruler or other measuring device pick 5 shoots per tagged plant and measure their length from the point where they leave the branch to the base of the flower or fruit or to the tip of the stem if there is no fruit (in mm).

Q3. Number of buds: Count the total number of flower buds at the beginning of season for each tagged plant.

Q4. Total number of flowers per individual: At the Full Flower stage, count the number of flowers for each tagged plant.

Q5. *Number of mature fruits:* Count the number of flowers that produced mature fruits (with seeds). The presence of seeds in a capsule is easily detected by squeezing the capsule gently between two fingers.

Phenology

P1. *Flower Bud Break*: Record the date as soon as any of the purple petal is visible as it emerges from the green bud.

P2. *Full Flower*: Record the date that the flowers are completely open and bright orange pollen is visible on the stamens inside the flower.

P3. *Mature Fruit*: After the petals fall off the ovaries will swell producing two relatively large fruits. Record this date.

Snow Buttercup
Ranunculus nivalis L.
Growth form: forb

The basal leaves of snow buttercups are divided into between 3 and 5 lobes each of which has a toothed or slightly lobed border. There are usually 1 to 3 stem leaves and a single yellow flower on top.

Tagging

Individual buttercup plants are the unit of observation. The stems are fragile so you shouldn't wrap string or wire around them, but affix the tag to the ground adjacent to the plant.

Quantitative Observations

Q1. *Height of flowering shoot:* At full flower, measure the height of the flowering shoot from ground to base of flower (in mm).

Q2. *Width of largest basal leaf:* Measure the width of the largest basal leaf (in mm).

Q3. *Number of Nutlets per Flower*: At the date when the nutlets are released count them and harvest them in paper seed bags.

Q4. *Average Seed Weight*: Dry the seeds at room temperature and weigh them to an accuracy of 0.1 mg. Divide the weight by the number of seed to arrive at an average seed weight.

Phenology

P1. *First Flower Open*: Record the date when the first flower is open and bowl shaped.

P2. *First Seed Dispersal*: Record the date when the small nutlets are released.

Arctic Lupine
Lupinus arcticus Wats.
Growth form: forb

Arctic Lupines have irregular purple flowers that grow out of a clump of palmate leaves at the base. Lupines are in the pea family so in the fall seeds develop in pods along the stem.

Tagging
The unit of observation for Arctic Lupines is an individual plant. Tagging is best accomplished by affixing a tag to the ground adjacent to the plant.

Quantitative Observations

Q1. Clump Cross-Section Length: The cross dimensions of a clump, measuring the longest diameter in the clump and the diameter at 90 degrees to this.
Q2. Petiole length: The length of the longest petiole;
Q3. Leaflet length: The length of the longest leaflet on the longest petiole;
Q4. The number of flower heads in the clump

Dwarf Birch
Betula glandulosa Michx. / *Betula nana*
Growth form: deciduous dwarf shrub

Dwarf birch, as the name implies, is a small bush that, in this region, typically grows to 50 or 60 cm in height. It may grow quite tall (1 metre) in areas that accumulate snow (in the lee of eskers, rocky outcrops or in protected valleys) as the snow protects the plant from wind damage in the winter. The "flowers" of birch are borne in catkins (simply a collection of flowers) but do not look at all like flowers; they are oblong structures approximately 1 to 1.5 cm in length. Catkins are familiar as "pussy willows" on other trees. On birch, male and female catkins will appear on the same branch. Male catkins produce the pollen and female catkins produce the ovules that will give rise to seeds.

Tagging
The unit of observation for birch is an individual branch. The tag should be attached to the chosen branch with wire or string, but no so tight as to damage the plant.

Quantitative Observations

Q1. Annual Growth Increment: The birch branch will grow in length from all the tips that come off the main branch. Some tips will grow more than others. It will appear reddish in comparison to the old growth, usually extending from a small bulge in the branch. When measuring the growth increment (the amount of new growth) on the tagged branch, measure what appear to be the three largest increments. Measure from where growth started (the bulge in the branch) to the tip of the terminal bud on the branch (a leaf may extend beyond this). Take measurements at the end of July or in early August.
Q2. Length of longest leaf: Choose 3 of the longest leaves on the tagged branch. Measure their length from the base of the petiole (the stem of the leaf, which attaches to the branch) to the tip of the leaf. Place the ruler on the inside of the leaf so that it can rest in the notch created by the petiole and the branch.
Q3. Shoot Number: Number of short and long shoots on a main branch. Short shoots can be identified by having only a very short woody branch length, that is rough from numerous, closely spaced bud scars. There will be 2 or more leaves growing from the tip of short shoots. Long shoots have clear woody stem elongation, and have leaves spaced **individually** along the stem. Look for bud scars at the base of the current year growth

(generally green in appearance) to determine the base of the long shoot. Short shoots are common on the lower portion of the branch, and long shoots common at the tip.

Q4. Number of Male Catkins: It will be especially important to keep a running total of male catkins. Number will increase as more are produced but will decrease as they are eaten by songbirds and other critters. Also, once the pollen is dispersed they become dry and blow away. Make sure that you count the number of male catkins at snowmelt then every few days after that. Keep notes of your tallies and the date they were taken. Record the highest number of catkins you counted and record the date you counted it.

Q5. Number of Female Catkins: Count the number of female catkins once stigmas are visible and then every few days after. This will ensure that you will catch any that are late to emerge. Record the largest number of female catkins you counted once you are sure that no more are being produced. Record the date of the largest number in the notes.

Q6. Leaf Length: Leaf size reflects the environmental conditions of the current year and provides a good measure of how well the plant is doing. Measurements are typically done with callipers (although a steel ruler will do). Measure the 3 largest leaves on the tagged branch at the end of July or early August. Measure from the tip of the green leaf to the end of the petiole (where the leaf attaches to the branch).

Phenology

P1. *First Leaf Bud Burst*: Birch leaf buds are formed the previous year and are covered with brown scales. Watch for the bud scales to open slightly and the green leaves to protrude. Record the date when the first bit of green leaf can be seen protruding from the scales.

P2. *First Catkins Visible*: Male catkins will likely be present at snowmelt as they are produced in the fall of the previous year or in the spring. They will be obvious at the end of branches as brown, oblong structures.

P3. *First Pollen Shed*: The male catkins start off small and tight but loosen and pull apart to release the pollen. When the catkin "opens" the yellow pollen is visible and is shed. You will notice puffs of powdering pollen when the branch is moved as you check it and also as you walk through adjacent bushes. Pollen shed can be recorded when yellow pollen is visible in the "open" catkin.

P4. *First Stigmas Visible*: The female catkins will first appear like a small, green "cone" and will come from within a leaf bud encased in surrounding leaves. The purplish-red-coloured female stigmas appear a few days after the female catkin becomes visible. Record in the notes when the first female catkin becomes visible. They will enlarge and then the stigmas will elongate and make them look slightly fuzzy. When you see the red stigmas sticking out from each "scale" of the "cone" record the date.

P5. *First Leaf Senescence*: Birch is a deciduous bush meaning that it sheds it leaves every fall and grows new ones in the spring. Before shedding leaves, the plant will conserve nutrients by moving them from the leaves into other parts of the plant for storage. Birch leaves turn a beautiful red in the fall as chlorophyll and plant nutrients are translocated to other organs in preparation for winter. Leaf senescence is recorded when one leaf on the branch begins to turn yellow or rusty, as opposed to turning brown from predacious insects.

P6. *Last Leaf Senescence*: Record the date when the last green leaf turns red or orange.

P7. *First Leaf Shed*: After most of the nutrients in the leaves have been translocated to storage organs in the plant the leaves will dry up and fall off. Record the date when the first leaf falls off the branch.

P8. *All Leaves Shed*: Record the date when the last leaf falls off the branch.

Labrador Tea
Rhotodentron decumbens / Rhotodendron groenlandicum (Formerly *Ledum*)
Growth form: evergreen shrub

Labrador Tea is a low shrub, seldom exceeding 20 cm in height. It has white flowers in small bunches above stem leaves with edges that are curled and undersides covered in reddish-brown wool. It generally has many branches extending from an extensive underground root system.

Tagging

Individual Labrador tea plants are the unit of observation. Affix the tag to the ground adjacent to the plant.

Quantitative Observations

Q1. Annual Growth Measurement: *Ledum* grows from the tip of the stalk and the new growth produced this year will be obviously lighter green and "fresher" looking. There will be a noticeable colour change from old stalk to new stalk (reddish to light brown/green). You will also be able to see bud scales (brown, triangular shaped) around the stalk where growth started. Measure the growth of *Ledum* at the end of July or early August. Measure from the remnants of the bud scale on the stalk to the tip of the stalk. Be sure to note if the parent branch produced a flower head in the current year.

Q2. Number of Flowering Stalks: A *Ledum* branch typically has one, two or three flowering stalks. Or it can have three stalks of which only one or two are flowering. Often, as well as having a stalk that is flowering this year, a branch will have stalks that have flowered in previous years. In this case you will be able to see the remnants of previous year's fruits. The number of flowering stalks can be counted as soon as the first flower opens. Be sure to count only the number of stalks that are flowering (or have swelled flower buds) in the current year.

Q3. Number of Flowers per Stalk: The number of flowers per stalk range anywhere from 1 to 18 flowers (perhaps more). These flowers can open at very staggered intervals or all at once. This can make it difficult to count. Flowers per stalk can be counted as soon as most of the flowers have opened. Be sure to count all the flowers on each stalk and enter them on the data sheet separately so that an average number per stalk can be calculated.

Q4. Number of Fruit per Stalk: As the fruits mature they will become oblong, brown little "nutlets" on the end of the flower stalks. Continue to watch these plants after the flowers drop to observe the development of the fruits. When you no longer see any more fruits developing record the number of fruits per stalk. It is important to wait a while for the fruit to develop after the flowers drop for two reasons. First, fruit will become larger and easier to count. And secondly, not all flowers will produce fruit so you want to know for sure that all the fruit that is going to develop has done so.

Phenology

P1. *Flower Buds Visible*: *Ledum* produce spherical buds at the tips of their branches. These buds can either be leaf buds or flower buds, and are difficult to distinguish when they are small. In general, the flower buds appear before the leaf buds. The flower buds swell into a spherical shape and are slightly whitish in colour, whereas leaf buds elongate and develop a rusty fuzz on the outside of the bud. Check for flower buds as soon as the snow is free as buds are created in the fall and overwinter. Mark the first branch you find with a flower bud on it (tie a piece of flagging tape around the base of the branch), and monitor that branch only for the rest of the season (even if it is not the first branch to proceed through the other phenological events).

P2. *Flower Bud Break*: As the flower buds mature, they will swell further and the scales surrounding the bud will split open to reveal a lumpy white surface. Record this date. These lumps represent individual flower buds, which will further separate and spread out on stalks before opening up into flowers.

P3. *Full Flower*: The date the first flower has opened completely with the stamens visible between the unfolded petals.

P4. *First Flower Shed*: Just as flowers on a single branch opened at various times, flowers will be shed at different times. First flower shed is when the first flower falls off of any of the stalks on the marked branch.

P5. *Last Flower Shed*: The date when the last flower is shed from all of the stalks on the tagged branch.

P6. *First Fruit Visible*: A few days after the flower drops the fruit will begin to become obvious. It will be a round greenish-brown swelling that turns brown as the season progresses. Record this stage as soon as there is an obvious swelling where a flower had been. Be sure to wait for the swelling, as some flowers will not be fertilized and will not form fruit.

Oxytrope / Locoweed
Oxytropis nigrescens
Growth form: forb

Oxytropis nigrescens, a member of the legume family, is small dull green plant with an extensive root system. This plant is small and can be difficult to locate at the beginning of the year, as only a small amount of dead plant material remains above ground over winter.

Tagging

The unit of observation for Oxytrope is an individual plant. Tagging is best accomplished by affixing a tag to the ground adjacent to the plant.

Quantitative Observations

Q1. Diameter of Plant: The diameter of the plant is an average of two measurements that bisect the plant and stretch from leaf tip to opposite leaf tip. If the plant was oblong, measurement should be taken of the length and width. Try to take a measurement of the longest diameter and the shortest to get a good estimate of average diameter.

Q2. *Number of Buds*: The number of buds should be noted as soon as they start appearing (in case an animal decides to dine on them). Keep an ongoing record of the number of buds, and when you are satisfied that no more will appear record the number.

Phenology

P1. *First Green Leaf*: Record the date as soon as you can see faint trace of green in the brown leaves from last year. The faint green will become lush quite quickly, and flower buds will soon appear.

P2. *First Flower Bud Visible*: The flower buds are little fuzzy green ovals with brown fringes on the end and are quite large in comparison to the plant. They will appear at the base of the leaves and can be partially obscured when they first appear, so look carefully. Record the date as soon as you notice the first flower bud. Not all of the plants will flower, but continue checking plants occasionally until buds start to open to ensure you don't miss the appearance of any late buds.

P3. *First Flower Open:* The flower will start to push through the green bud case, first as a yellowish-purple mass, which will elongate into a purple protrusion and finally unfurl into the *Oxytropis* flower. It only takes a few days for the bud to open into a flower, so once things start happening, the plants should be monitored daily. Record the date when the flower is fully open.

P4. *First Petal Dropped:* The petals will wilt and brown quite quickly, but some may cling on tenaciously. Don't touch the plants, but just watch carefully for when the petals have finally fallen off of the bud. Record the date when the first petal of any of the flowers on that plant has fallen off.

P5. *Last Petal Dropped:* Record the date when the last petal of all the flowers on that plant has fallen off.

P6. *First Seeds Dropped:* The base of the flower will develop into a structure that looks like a pea pod. The green sides of the pod will dry out into paper-like pods and will split down the middle and you will see dark seeds inside. Record the date when the pod has split open.

Polar Grass
Arctagrostis Latifolia
Growth form: graminoid

Tagging

The unit of observation for polar grass is an individual tussock. Tagging is best accomplished by affixing a tag to the ground adjacent to the plant.

Quantitative Observations

Q1. *Flower height:* Measure the heights of mature flowers from the ground to the top of the terminal spike. Choose random plants in the plot at peak season (end of July – early August).

Q2.b *Number of Leaves*: Count the number of fully unrolled leaves.

Q3.b *Green and Total Leaf Length*: Measure the length of the longest leaf and then measure the length of the same leaf that is green.

Phenology

P1. *Leaf Bud Break*: first sign of leaf appearance.

P2. *Mature Flower*: inflorescence opens, stamina and stigmata visible or pollen shed from anthers.

P3. *Senesced*: The first sign of seed development or when the bud turns yellow in colour (some buds don't develop into a mature stage and turn yellow after the bud stage).

Prickly saxifrage / Spotted saxifrage
Saxifraga tricuspidata
Growth form: evergreen forb

Prickly Saxifrage is an evergreen species. The leaves will turn red in the fall and then green up again the following spring but they are not shed. The prickles are found on the leaves which have three points on the ends. Another name for this plant is Spotted saxifrage in reference to the orange spots on the white flowers. The plant grows in mats on the ground and can be virtually covered in white blossoms in late June. Old, dry flower stalks from last year will be present extending from the clumps in the spring.

Tagging

Individual prickly saxifrage plants are the unit of observation. The stems are fragile so you shouldn't wrap string or wire around them, but affix the tag to the ground adjacent to the plant.

Quantitative Observations

Q1. *Number of Flower Stalks*: The flower stalk extends from the end of the shoot and carries a number of flowers. Be sure to count only the stalks that arise from the shoots and not those that hold each flower. Count the total number of flowering stalks on the marked plant as soon as flowers start to open.

Q2. *Diameter*: The diameters of the clumps are measured every year at the end of July or beginning of August. Three measurements are taken: along the longest width, the narrowest and then an approximation of the diagonal.

Phenology

P1. *First New Leaf*: New, green leaves will emerge from the centre of each shoot. Old leaves will be red as they first become snow free. They will slowly turn green as the weather warms but usually the tips remain red. It is after the old leaves green up that new leaves are produced. Watch the end of each shoot in the clump carefully. You will notice a small oblong swelling, light green in colour protrude from the centre. It will slowly enlarge and will open to present three or four separate leaves that are entirely green. Record the date when the old leaves at the end of the shoot open slightly to reveal small leaves on at least one shoot of the tagged plant.

P2. *Flower Buds Visible*: Some shoots will produce flowers, not new leaves. They will emerge from the centre of a shoot as small, whitish balls instead of the oblong green swellings of new leaves. They are contained on the end of a stalk that will elongate to 10 to 20 cm. However, you will notice the buds long before the stalks are that length. Record the date as soon as you can distinguish the buds at the end of the shoot from new leaves.

P3. First Flower open: A single saxifrage plant will produce many flower stalks with several flowers on each stalk. The flower is considered open when you can see space between the petals of a flower. Record the date when you can see space between the petals of any flower on any of the stalks on the marked plant.
P4. *First Petal Shed*: Flower petals will fall individually. Record the date when the first petal falls off of any of the flowers on the marked plant.
P5. *Last Petal Shed*: Record when the last petal falls off the flowers on the plant

Water Sedge
Carex aquatilis (stans)
Growth form: graminoid

Carex aquatilis is a sedge, like *Eriophorum*, but is quite different in its reproductive structures (flowers) and strategy (it flowers quite late in the season). *Carex aquatilis* grows from underground rhyzomes. Shoots or tillers come out of the ground along the rhizome. Each shoot grows for several years, flowers then dies. Other shoots will come up from other parts of the rhyzome.

Tagging
Because flowering shoots of water sedge die within a year a different flowering shoot is monitored every year. You will notice tags on the ground throughout the plot. When a flowering shoot is observed in a 15cm radius (approximately) it is marked and observed for the rest of the season.

Quantitative Observation
Q1. Flower height: At full flower, measure the length of the flowering stem to the base of the terminal spike (in cm).
Q2. Number of green leaves: At full flower, count the number of green leaves on each plant;
Q3. Length of longest leaf: Measure the length of the longest leaf from the base of the stem to the tip of the leaf (in mm).
Q4. Age class of shoot in flower: A specific tiller or shoot will grow for several years, producing leaves each year. It will flower and then die the following season. The number of dead leaves surrounding a flowering shoot can be used to estimate how many years the shoot grew before producing a flower. Count and record the number of dead leaves that surround the flowering spike right after stigmas and anthers have become visible.
Q5. Green and yellow leaf lengths: The amount of green leaf production by a single shoot or tiller can provide insight into the growing conditions of the particular season. Measure the length of the live green portion of the leaf and the length of the senescing yellow part of the leaf. Measurements are made from the ground to the appropriate part of the leaf. Measure all the green leaves surrounding the flowering stalk. Place the ruler in the centre of the tiller where the leaves join the stem and measure to the tip of the green portion of leaf. For leaves that are partially brown, measure and record the length of the green portion only. This will provide a measurement of how much growth occurred this season.
Q6. Length of Flowering Stem at Full Flower: By August the inflorescence will be enlarging and drying out. Measure the length of flowering stem at full flower, which occurs in late July or early August after the stigmas and anthers have emerged. Measure the length of

the flowering stem from the ground to base of terminal spike (base of tallest male spike, not the tip of this spike).

Phenology

P1. *First New Leaf Visible*: Old, brown leaves will be evident near the tag. Growth will continue on these leaves so they will be green on the bottom portion and brown at the tips. A new leaf from this season will be entirely green and come up from the centre of a clump of older leaves. You will have to pull apart the older leaves slightly to watch in the centre. Record the date when you first see a small, entirely green leaf arising from the centre of a *Carex* shoot

P2. *Flower Bud Break:* The flowering stalk will come up from the centre of the Carex shoot. Record the date of the first sign of an inflorescence visible at the base.

P3. *First Stigmas Visible*: The flowering stalk usually consists of three parts: a female inflorescence and two male spikes at the end of the stalk. As the stalk emerges you will see the male structures first and the female afterwards. The female inflorescence is oblong, compact and brown. Soon after emergence, the inflorescence will loosen and white stigmas will extend outward. Record the date when the white stigmas are protruding out from the inflorescence.

P4. *First Exposed Anthers*: Once you see the male spikes, anthers will appear shortly. The spikes will open slightly and the yellow anthers will extend outwards. Record the date when you see yellow anthers protrude from one of the male spikes.

P5. *First Yellowing of Leaves*: *Carex* leaves will start to yellow in early to mid August. Tips of the leaves will yellow first then slowly the entire leaf. Record the date when the tips of leaves on the marked shoot become yellow.

P6. *First Seed Shed*: The female inflorescence will slowly enlarge and dry out as it matures. It will have obvious separate seeds that will come loose and blow away. Seeds are usually shed in mid to late August. Record when you see holes in the inflorescence where seeds have come loose.

Luzula arctica / Luzula confusa
Growth form: graminoid

Luzula species are in the rush family. There can be from 5 to 60 flowers in a dense head per inflorescence, growing out of a basal tuft of leaves that can last for several years.

Tagging

You will likely have to mark out a 10 cm X 10 cm area with wooden skewers and string as your unit of observation for Luzula species.

Quantitative Observations

Q1. Length of the longest leaf: Measure the length of the longest leaf from the base to the tip.

Q2. Height of the inflorescence: Measure the height of the inflorescence from the base of the stalk to the top of the terminal spike.

Q3. Clump diameter: Measure the clump at its widest point and again at 90 degrees to this.

Q4. Flower height: Measure the distance from the ground to the base of the inflorescence.

Q5. *Flowers per plant:* The number of inflorescences in the whole clump regardless of what state they're in.

Phenology

P1. *Flower Bud Break:* Record the date at the first sign of a flower inflorescence in the base of the tagged plant.

P2. *Mature Flower:* Record the first date the stigmata or stamina are visible protruding from the flower head.

P3. *Flowers Senesced:* Senesced flowers are recognized by dried up stamina or stigmata, the single flowers swell slightly.

4.5 Damage to Plant by Herbivory and Disease

Many factors influence the growth and development of plant species. Vegetation might not be responding directly to changes in climate but could be responding to increases or decreases in damage caused by herbivores, insect infestations or disease. It is important to track these other variables in order to assess their significance in any trends observed in vegetation monitoring.

During all baseline measurements and annual monitoring, observations on herbivory and disease should be recorded. The type of damage will depend on the herbivore, especially whether they are insects or mammals. Leaf eating insects, such as species of the woolly bear caterpillar (*Gynophora* spp), can be seen on the leaves of plants, and through simple observation you will be able to identify the damage caused by these herbivores. Grazing animals such as muskox and caribou tend to clip the plants off, leaving blunt ended shoots of graminoids and shrubs. If you are in an area that is known to have moderate levels of herbivory, then you should examine plants in vicinity for signs of damage. The level of damage can be monitored by examining tagged shoots for evidence of herbivory, or counting the number of grazed or damaged shoots in a standard area (e.g. 10 x 10 cm subsections of a 1 m^2 plot). There are generally three grazing related features: 1) faeces/droppings, 2) browsing damage, and 3) trampling (including human trampling). In the data forms used to record observations you may choose to have dedicated columns for recording damage caused by herbivory or disease, or simply a general '*notes*' column in which this information could be included. Where possible, the researcher should consider establishing exclosures to remove the effects of the grazers (see section 5.5).

Plant disease should also be recorded in the plots. There is a large variety of diseases that attack arctic plants, and it would be impossible to list all potential diseases and their symptoms. The researcher should note whether there are lesions or other manifestations of disease on the plant, and attempt to quantify the frequency of these symptoms. This can be done by determining the ratio of plants (parts of plants, such as stems, leaves) showing symptoms of disease to the total population of plants within a standard areas (e.g. 1 m^2). For further advice and recommendations, CANTTEX researchers should consult references on plant pathogens, and Dr. Greg Henry.

4.6 Data Entry

Data on individual plant growth and phenology are entered on a plant by plant basis (Fig. 6). A sample form for field data entry for one species is provided in Appendix A as an example. If you use abbreviations make sure that these are properly explained somewhere accessible to field observers and to those entering and processing the data. Phenological observations should be

recorded as the day number (sometimes erroneously referred to as Julian day – see Appendix C). Make sure that quantitative observations are always recorded in the same units (mm, cm, etc.).

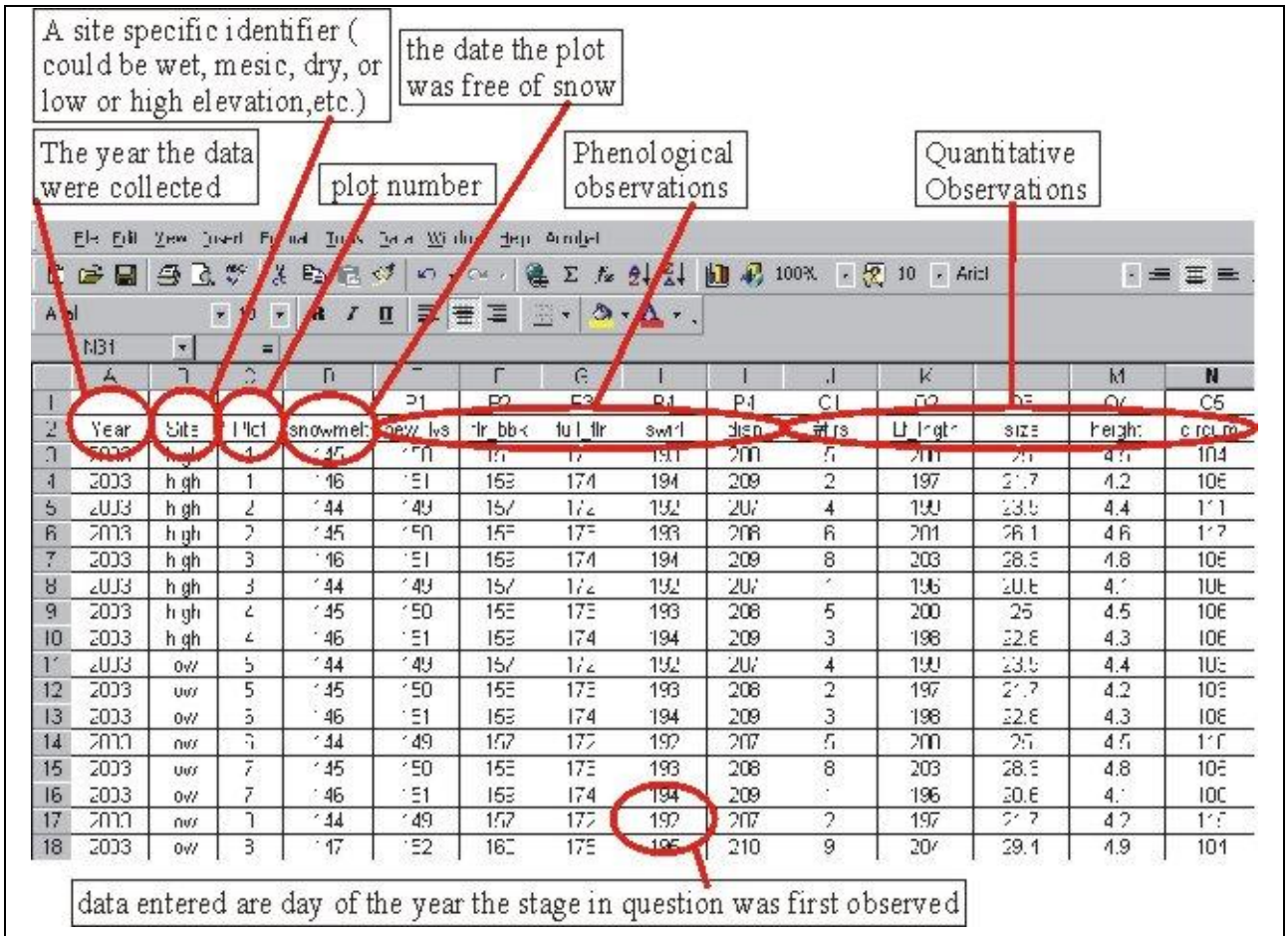


Figure 6. Example of spreadsheet data entry sheet for phenology and growth observations. The example is for *Dryas integrifolia*.

5.0 Experimental Methods

5.1 Introduction

The most powerful test of any theory is to use a controlled experiment. Controlled experiments deliberately modify one or a few variables in order to gauge the effect of the change on a system. Many different types of experiments are possible in arctic ecosystems with the purpose being to study how tundra systems respond to changes in temperature, precipitation regime, and nutrient availability all of which are possible consequences of climate change. The International Tundra Experiment (ITEX) is a network of sites throughout the North American and Eurasian Arctic all conducting similar experiments designed to examine the responses of tundra ecosystems to environmental change. At ITEX sites, small greenhouses are used to warm patches of tundra vegetation, snow is added or removed from plots and fertilizer is added to plots in an attempt to mimic some of the effects expected with climate change. Researchers then carry out the same

monitoring activities described above on the treated plots to measure the effect of the change. There are currently five sites in Canada with ITEX experiments and more experimental sites are always desirable.

The basis of experimental design is the evaluation of the difference between the treated plot (the one that has been warmed or fertilized) and the control plot (which is left in its natural state). The other crucial principle is that of replication to ensure the consistency of results. This means having enough treated plots and enough control plots to be confident in the observed differences. ITEX protocols suggest having six treatment plots and six control plots to ensure adequate replication (see section 2.6 in first volume of the manual for a discussion of sample sizes). Equal numbers of experimental and control plots should be located randomly within an area that has similar vegetation and environmental attributes. Within each plot, individual plants of each target (e.g. dominant) species should be monitored as outlined above in section 3.3.3.

If more than one variable is being manipulated, then the experimental design becomes slightly more complicated. In order to maintain control and replication it is necessary to set up a factorial design, meaning that every combination of manipulated and un-manipulated variables is sufficiently replicated. For example, if you were planning to test warming and fertilization you would need to have the following set of plots:

1. warmed and fertilized;
2. warmed but not fertilized;
3. fertilized but not warmed;
4. neither fertilized nor warmed (control).

For each of these combinations at least five plots would have to be monitored to ensure proper replication. The selection of which plots receive which treatment must be made at random. The specific methods for applying treatments for warming, snow manipulations, fertilization, watering, and herbivore exclusion are described below.

5.2 Warming

Warming experiments have been established in many sites throughout the Arctic (Henry and Molau 1997; Arft et al. 1999; Shaver et al. 2001). The warming is achieved either passively, by placing a small greenhouse over the plot, or actively by using an electrical heating device. The active heating devices include heating cables laid on the ground and infra-red heating lamps placed above the surface. The advantage of the active systems is that the research can control the level of warming, and provide a constant temperature differential between the warmed and control plots. However, in most arctic situations the lack of electrical power limits the researchers to the use of passive heating treatments. The open-top chambers (OTC) used in the International Tundra Experiment have proven to be very useful in warming small plots of tundra ecosystems. Researchers are directed to the ITEX Manual and to a paper by Marion et al. (1997) for descriptions of the OTCs and their warming performance. The open-top design was adopted in ITEX because it would allow for normal gas exchange and would not interfere with precipitation, though there are some secondary effects including differences in relative humidity and herbivore and pollinator exclusion. However, closed greenhouse designs are also used (e.g. Chapin et al. 1995). While it is recommended that new CANTTEX sites adopt the ITEX methods for warming treatments, any greenhouse design is acceptable. The researcher must

ensure that the temperature effect is properly measured using temperature sensors in both warmed and control plots, preferably including both air and soil temperature.

5.3 Snow Manipulations

Climate change will involve changes in precipitation patterns. Snow cover is important as an insulation layer during the winter, and for providing melt water to plants at the beginning of the growing season. Snow melt date is the beginning of the growing season, and changes in the snow depth will affect the length of the growing season. Changes in the snow fall regimes will, therefore, strongly affect tundra ecosystems. The manipulation of snow cover generally involves two approaches: 1) use of snow fences, or 2) manual addition and removal of snow.

1) Snow fences: Snow fences have been used successfully in a number of arctic sites for altering snow depth and melt dates. The main effect of the snow fence is to increase snow depth behind the fence. However, there is usually an area just in front of the snow fence with shallower snow depths than in the surrounding (ambient) area. This allows the comparison of areas with increased and decreased snow depth with ambient conditions.

   The height and length of the snow fence will depend on the site and objectives of the researcher. In most tundra situations, a 1.5 or 2 m high fence will be enough. Plastic snow fencing material can be purchased in rolls of varying widths, and attached to wooden or metal stakes placed into the ground at regular intervals. The fence must be arranged across the prevailing winter wind direction. Plots can be arranged systematically at distances away from the snow fence in both directions.

2) Snow addition and removal: In areas where snow fences cannot be established, researchers can manually change the snow depth through the addition and removal of snow. While this can be labour intensive, it allows the researcher to control snow melt date to a greater degree than with a snow fence. The major disadvantage is that the snow cover is disturbed, which changes the properties of the snow. It is best to perform the snow manipulations prior to snow melt. Removing snow from plots must be done carefully to avoid damaging plants. You should not try to remove all the snow. Take the snow down to a level just above the plants, leaving about 5 cm over the tallest plants. This will ensure the plants are not damaged. Snow should be added from areas around the plots so they receive approximately twice as much snow as the ambient areas. This is necessarily a subjective measure, since the snow compacts as it is added. However, some control can be gained by measuring the depth and area in the addition plots, and removing a similar depth and area from outside the plot to be added to the experimental plots.

3) Alteration of snow melt rate: The melt rate of the snow pack can be increased by adding material to reduce the albedo (reflectivity) of the snow surface. Typically, a black plastic mesh or screen can be placed over a specific area, and the increased net radiation results in a more rapid melt rate. See Walsh et al. (1997) for an example of this technique.

5.4 Fertilization

Fertilization experiments are meant to determine the response of tundra systems to increased nutrient supply. Tundra ecosystems are known to be strongly limited by nutrient availability. It

is still unknown how the changes in climate will affect the supply of nutrients in tundra systems, but it is hypothesized that warmer temperatures will increase decomposition and mineralization rates, resulting in greater nutrient supply to plants. Fertilization studies have been conducted in a variety of tundra ecosystem types, and have shown that these nutrient limited systems respond very strongly to increased nutrient supply (e.g. Henry et al. 1986; Chapin et al. 1995). These experimental studies are usually plot-based, with a typical plot size of 4 - 9 m^2. Fertilizer is usually added each season at prescribed rates, which depend on the objectives of the research. In general, two rates of fertilization are preferred, with one considerably smaller than the other. The small rate of application would be designed to mimic predicted increases in supply through climate warming effects, while the higher rate would elicit a strong response from the plants. Many fertilization studies involve the addition of standard, composite fertilizer (e.g. equal concentrations of N, P, and K), unless the researcher is interested in the relative limitation of nitrogen, phosphorus or other elements. For the purposes of CANTTEX, a single addition per year of a standard, composite fertilizer to replicate plots would suffice. The researcher should attempt to increase the nutrient supply by 2-10 times the ambient supply rate, if known.

5.5 Watering

Climate change will result in areas receiving more or less precipitation. In areas that are expected to received increased rain in summer (e.g. the eastern Canadian Arctic), watering experiments can be used to mimic the predicted increase in rain.. As with fertilization experiments, watering treatments are applied to plots, and usually at rates that attempt to increase the summer precipitation by 2 - 5 times. The water is usually added manually, using a watering can.

5.6 Herbivore Exclusion

Tundra systems, like other terrestrial ecosystems, are strongly affected by large herbivores (Henry 1998). An indirect measure of the effects of herbivores on the systems is to exclude them from small areas and monitor the changes. Again, these experiments are plot-based, with comparisons made between plots with and without exclosures. Exclosures are designed to remove the effects of all herbivores at the site, and usually involve placing a mesh barrier around the plot. The size of the mesh will depend on the size of the herbivore.

In some cases, herbivore exclosures would be used in conjunction with other treatments, such as warming, where the researcher would need to separate the effect of temperature enhancement effect from the herbivore exclosure effect of passive warming devices such as greenhouses. There are many studies using herbivore exclosures, and researchers interested in establishing these at their site should consult the literature and contact Dr. Greg Henry for further advice and recommendations. Again, care must be taken to ensure that OTCs do not act as enclosures which would result in a confounding effect.

6.0 Nutrients

6.1 Tissue Carbon and Nutrient Concentrations

The nutrient concentration of plant parts depends on many factors, including: phenological and growth stage, soil nutrient, moisture and temperature conditions, and tissue type. New growth stimulated by warmer temperatures will not be sustained unless the supply of nutrients can be

increased to meet the growth demands. While there are some good studies of the effects of experimental warming on tissue nutrient concentrations, little is known of the annual variation in nutrient concentrations in ambient plants. Furthermore, the nutrient concentration of plants determine the value or quality of forage for herbivores and for litter. Changes in tissue nutrient concentrations will directly affect the cycling of nutrients through the changes in the quality of the litter for decomposers - generally measured as the ration of carbon to nitrogen concentrations. An increase in C:N ratio will decrease the nutrient value of the litter to decomposers, and could slow the rate of decomposition. For all of these reasons, some systematic measure of the nutrient concentrations of plant parts at the site is desirable.

Many levels of studies can be designed to examine seasonal and annual changes in nutrient concentrations in tundra and taiga plants. The intensity and detail of the study will depend on the objectives and interests of the researcher, and the protocols presented below are meant to serve as a guide for the basic level of study. These basic results can then be used in cross site comparisons. It would be desirable to have plant nutrient concentrations measured in dominant species of the major functional groups at the site once every 3-5 years. If time and funding only allow for one sampling period, measurements of the carbon and nitrogen concentrations in fresh litter at the end of the growing season is most desirable.

C:N ratio

Field sampling: Measurement of the carbon and nitrogen concentration in fresh litter will provide data on the litter quality of the site, and any changes in litter quality over time. Samples should be collected as close to the end of the growing season as possible, but certainly after senescence has begun (see phenological observations). Senesced leaves and flowers (whole inflorescences in some species) should be collected either from the plant or from the ground if you are sure of the species and that the litter is from the current year. Collect at least duplicate samples of each species per plot, and place them in labelled envelopes (coin envelopes are useful for this).

Lab preparation: The samples should be air dried in the field. If you send the samples directly to a lab for C:N analysis, make sure the samples are labelled well. If you prepare the samples, follow the procedures recommended by the lab personnel where you plan to have them analyzed.

Measurements of carbon and nitrogen concentrations in plant tissue (and soils) are made routinely in laboratories using CHN Analyzers. For most labs, sample weights can be as small as 0.1 g when oven-dry, which amounts to a few leaves of most tundra plants.

6.2 Soil Nutrient Availability

Fertilizer addition in several tundra communities has shown that nitrogen (N) is the primary factor limiting plant growth in arctic terrestrial ecosystems. Despite substantial stores of organic N held within tundra soils, low temperatures and anaerobic soil conditions reduce rates of vegetation decomposition and nitrogen mineralization.

Climate warming in high latitudes, as simulated using OTCs, is expected to increase plant productivity and absorption of CO_2 from the atmosphere. This negative feedback to CO_2 -induced

climate warming may be constrained by the availability of N in tundra ecosystems. Therefore, it is important to include assessments of nutrient availability in the CANTTEX protocol.

Suggested methods for the assessment of the nutrient economy at CANTTEX sites have been broadly categorized into 3 separate headings; soil nutrient availability, nutrient transformations, and plant – soil nitrogen transformations. The majority of the techniques listed below focus on N, however, phosphate is also included for some methods. An additional reference to this material is listed below. It provides more in-depth background and instructions for several soil analytical protocols.

Ion Exchange Resin Membranes

Ion Exchange Membranes (IEMs) are thin plastic strips that are coated with ion exchange resin. They are inserted into the soil for up to 100 hours to give an integrated flux of soil ions to the membrane exchange sites. Ions that may be included in nutrient assessments are ammonium (NH_4^+), nitrate (NO_3^-), and phosphate (PO_4^{-3}), however, other soil ions may also be examined (e.g. potassium, calcium, sodium, magnesium). The greatest benefits of IEMs compared to soil coring/extracting methods are:

i. The IEMs are relatively less destructive than soil coring. They are inserted into a thin slit into the ground and removed. Soil coring involves permanent removal of soil from site.
ii. The IEMs provide an integrated flux of nutrients through the soil over time, whereas soil cores will only provide instantaneous measures of nutrient availability. This is particularly important for mobile ions such as NO_3^- and PO_4^{-3}.

Materials and Methods:

Note: all lab materials (vials, beakers, graduated cylinders, containers, etc.) should be cleaned and acid washed before use.

Membrane Preparation:

- The IEMs may be ordered from Ionics Inc. (http://www.ionics.com/). You will need to order 2 types. The anion exchange membrane (AEM) has a catalogue number of AR202 and the cation exchange membranes (CEM) are listed as CR627.
- Both types of IEMs will likely arrive in large sheets (18” x 40) that need to be cut into smaller strips, depending on the depth of soil you wish to sample. 5 x 5cm squares are recommended. Use a fabric cutter (a circular blade available in most fabric stores) to cut the membranes as accurately as possible.
- The IEMs are stored in de-ionized water (DIW) in clean containers or plastic bags prior to use. Refrigeration will prevent bacterial growth.

Membrane Pre-saturation:

- The IEMs are recyclable. To prepare either new or used IEMs for the field, both the AEMs and CEMs membranes must be saturated with bicarbonate and sodium ions, respectively.
- Prepare the 1.0 mol/L $NaHCO_3$ pre-saturation solution by dissolving 84g of $NaHCO_3$ in 900mL DI water, and dilute to 1L.

- While transferring the IEMs to the pre-saturation solution it is recommended that you use a hole punch to make a hole for attaching a label. If you place the hole in the corner of the AEM and in the middle of the CEM, for example, it will be easier to distinguish between them in the field.
- Shake the AEMs and CEMs separately in the solution for 10 minutes, and then rinse them in DIW. Repeat 2 more times and place in baggies for transport to the field. They may be used for up to 10 days.

Field Installation & Removal:

- Thread a loop of nylon fishing line through the hole in each IEM and attach a label to the other end. Wearing latex gloves and using tweezers will reduce contamination of the IEM.
- Make a slit in the soil using a putty knife and insert the AEM and CEM side by side into the soil. They should be placed at a slight angle from vertical (15 – 30 degrees) to obtain good soil-membrane contact.
- The membranes may be placed at any depth, in any soil horizon. Typically, the main rooting zone just below the surface is of greatest interest.
- Record the date and time of insertion.
- The IEM pair should incubate for no longer than 100 hours in order to measure an integrated flux of ions to their surface area (see calculations below). Beyond this time, the membrane may no longer maintain a near zero concentration of NO_3^-, NH_4^+, or PO_4^{-3} at its surface. If the concentration on the membrane approaches that of the soil, the membrane acts as a dynamic exchanger, and not as an ion sink.
- At the end of the incubation period, record the date and time and remove the IEMs using tweezers. Place both the AEM and CEM together in a suitably sized whirlpack.
- The IEMs should be frozen during storage and transport.

IEM Extraction:

- Gently rinse the IEMs in the whirlpack with DIW to remove all soil particles. Drain well.
- Prepare the 0.5 mol/L HCl extract solution: add 41.5mL HCl to 900ml DIW, and dilute to 1L.
- Note: salt solutions (e.g. KCl, K_2SO_4) will also work. Select one without chloride if soluble organic nitrogen will also be analyzed in the extract solution (see section (described above)
- Add 25mL of the 0.5 mol/L HCl to the whirl pack, seal, and shake on a shaker table for 20 – 30 hours. During this time keep the samples in a cooler with ice packs.
- Place new IEMs (pre-saturated only) in whirl pack and extract the same way to provide blanks for analysis.
- The solution may then be stored in clean polyethylene vials. Filtering should not be necessary if all of the soil particles were removed during rinsing.
- The solution is analyzed for NO_3^-, NH_4^+, and PO_4^- on a colorimetric analyzer (e.g. LACHAT auto analyzer).
- Note: if the solutions are to be stored for more than 1 day prior to analysis, they should be frozen.

Calculations:
N Flux = µmol ion/cm^2/hr

- Convert the concentration of the 25mL sample (ppm) to µmol.
- Divide this by the total surface area of the IEM (50cm^2 front and back), and divide by the total number of hours that the IEM was incubated in the soil.

Soil Extractable Ions – Salt Extractions
The instructions below give methods for the extraction of mineral nitrogen (NO_3^-, NH_4^+) and phosphate (PO_4^{-3}). For other ions refer to Chapter 6 of the LTER Soil Methods reference.

Materials and Methods:
Note: all lab materials (vials, beakers, graduated cylinders, funnels, containers, etc.) should be cleaned and acid washed before use.

Soil Collection and Preparation:
- Soil cores should be collected and either extracted immediately (preferred) or frozen for long term storage/transport. The depth of the core depends on the region of interest in the soil (typically the main rooting zone).
- Sieve field fresh soil through a 4mm wire screen to remove small rocks and twigs, and to homogenize the sample.
- Separate a small section of the sample (appx. 15g) and determine its gravimetric moisture content. (see Chapter 3 of the LTER reference for further information on this topic if required).

Extraction Procedure:
- Prepare extract solution: 0.5M K_2SO_4 (preferred if you are also analyzing the extracts for soluble organic nitrogen – described above). Dissolve 87.13g K_2SO_4 (< 5ppm N, HPLC grade) in 900mL de-ionized water (DIW), and dilute to 1L.
- Add fresh, sieved soil to a whirl pack with extract solution in a ratio of 1:4 (e.g. 15g soil: 60mL extract solution).
- Also add extract solution to empty whirl pack to serve as blanks for analysis.
- Shake for 1 hour on a shaker table. If a shaker table is not available, the sample may be shaken intermittently over a 24 hour period.
- Filter the soil solution through Whatman 42 filter paper into clean polyethylene vials. The filter paper should be folded into the funnel and pre-rinsed with the 0.5M K_2SO_4 extract solution prior to filtering.
- The solution is analyzed for NO_3^-, NH_4^+, and PO_4^{-3} and on a colorimetric analyzer (e.g. LACHAT auto analyzer).
- Note: if the solutions are to be stored for more than 1 day prior to analysis, they should be frozen.

Calculations:

a) Gravimetric basis: ***µg element/g soil = (C x V)/W***

Where:
C = concentration of element (NO_3^-, NH_4^+, or PO_4^{-3}) in extract solution as given by the analyzer in mg/L (note: 1 mg/L = 1 ppm).
V = volume of extract solution. *This includes the solution used, plus the water in the soil sample as determined by the gravimetric moisture content analysis.
W = dry mass of the soil. This is also determined using the gravimetric moisture content of the fresh soil.
Note: to convert this to *mol* element/g soil divide the above result by the molecular weight of the ion (e.g. NO_3^- = 62, NH_4^+ = 18, PO_4^{-3} = 95)

b) Area basis: ***g element/m^2 = Cg x BD x SD***

Where:
Cg = element mass on gravimetric basis as µg/g soil (as calculated above).
BD = bulk density as g dry soil/cm^3 (see chapter 4 of LTER Soil Methods reference if further information is required).
SD = sample depth in m.
Note: This will give you the element concentration per surface area for the depth examined.

Soluble Organic Nitrogen – Persulphate Oxidation
Another pool of nitrogen that is important to examine as a source of nutrients to plants is soluble organic nitrogen (SON). It is more abundant in arctic soils than mineral NO_3^- and NH_4^+. Some arctic plant species are able to absorb and assimilate simple forms of SON (e.g. amino acids) directly into their plant tissues, while other plants rely on mycorrhizal associations to mineralize this nitrogen prior to plant absorption.

The procedures outlined below utilize persulphate ($K_2S_2O_8$) to oxidize SON and NH_4^+ in soil sample extracts to NO_3^-. Therefore, to obtain the total SON in the sample, the NO_3^- -N and NH_4 – N concentrations in undigested samples (described above) are subtracted from the final concentration of NO_3^- -N. This procedure is simpler, more accurate, safer, and produces less toxic waste than he traditional Kjeldahl method.

Materials and Methods:
Note: all lab materials (vials, beakers, graduated cylinders, containers, etc.) should be cleaned and acid washed before use.

Sample Preparation:

- This method has been adapted for use with ion exchange membrane or soil extract solutions. The procedures for obtaining these samples are outlined above respectively.
- Some studies have determined that incomplete oxidation occurs with 1M KCl extracts versus 0.5M K_2SO_4. This may be a result of the Cl^- ion that interferes with the

decomposition of urea and/or oxidation of NH_4. For this reason, extracts from soils or ion exchange membranes should be obtained using 0.5M K_2SO_4.

Preparation of Oxidizing Reagent:

- In a 500mL dark bottle dissolve 7.5g NaOH in 50mL de-ionized water (DIW) to make 3.75M NaOH.
- Dissolve 25g of low nitrogen $K_2S_2O_8$ (persulphate) and 15g of H3BO4 (boric acid) in the above solution.
- Dilute the solution to 500mL with DIW.
- This reagent can be stored for 1 week at room temperature in the dark bottle.

Oxidation of the Samples:

- Digestion tubes are standard 40mL borosilicate glass vials with autoclavable Teflon liners and screw caps.
- Prepare inorganic and organic standards of the same known concentration of N (e.g. potassium nitrate, and urea) to evaluate oxidation efficiency. These will be oxidized and analyzed with the prepared samples.
- Mix the sample/standard and oxidizing reagent in the vial in a 1:1 ratio. Do not exceed the maximum fill line on the vials for autoclaving.
- Set aside a separate sample for undigested analysis to obtain the initial NO_3^-—N and NH_4^+—N concentrations.
- Use a pipette with clean pipette tip for each sample/standard. Rinsing with 1% HCl followed by 2 rinses in DIW between samples is adequate.
- Autoclave at 120˚C for 30 minutes.
- Cool and transfer to polyethylene vials.
- The solution is analyzed for NO_3^- (only) on a colorimetric analyzer (e.g. LACHAT auto analyzer).
- Note: if the solutions are to be stored for more than 1 day prior to analysis, they should be frozen.

Calculations:

SON (µg N/L) = (Persulphate N / Digestion Efficiency) – Inorganic N

Where:

- Persulphate N = NO_3^-—N in solution after persulphate digestion (µg NO_3^-—N /L).
- Digestion Efficiency = (NO_3^-—N in organic standard / NO_3^-—N in equivalent nitrate standard). If efficiency drops below 80%, suspect a bad batch of oxidizing reagent and discard the results.
- Inorganic N = NO_3^-—N + NH_4^+—N in solution prior to digestion (µg N/L). This sample was set aside prior to oxidation.

6.3 Soil Nutrient Transformations

Net Nutrient Mineralization – Buried Bag Incubation

The buried bag approach is used to measure the net change in soil mineral nutrients over a period of time. Soil is isolated from plant roots by placing soil cores in polyethylene bags. Polyethylene bags are permeable to gases, but not to water. Therefore, the soil moisture should remain constant throughout the incubation period, which may be a significant source of error if the in situ soil moisture changes substantially over that time.

Materials and Methods:

Note: The incubation period may include an entire growing season, or a series of bags may be buried at the start of the season for sequential removal throughout. The timing and duration of the incubation period must be carefully considered in the design stages of the experiment. A review of other CANTEX studies may be useful in determining the incubation duration for comparison with other sites.

Soil Coring and Incubation:

- Carefully remove 2 adjacent, intact soil cores with a soil corer.
- Place the core to be incubated in a thin polyethylene bag, tie with a twist tie and attach a label.
- Place the soil core back in its original location and cover the top with a thin layer of soil/vegetation.
- The second core is returned to the lab for immediate inorganic N and/or P extraction and analysis as outlined above. If the core is to be stored or transported it should be frozen immediately.
- At the end of the pre-determined incubation period, the bagged core is removed from the ground, sieved, and also extracted and analyzed as outlined above.

Calculations:

- The amount of N and/or P mineralized over the incubation period may be expressed either as a gravimetric measure as: *μg element/g soil*
- Or, if the bulk density of the soil is known, on an area basis as: *g element/m*^{2}
- Subtract the initial mineral N or P concentrations from the final N or P concentrations (incubated core) to obtain the net change over the incubation period.
- *Note: It is not uncommon for growing season incubations to yield very low or negative net mineralization rates. This indicates high potentials for microbial immobilization. Some studies have found that microbial biomass N, a pool that may become available to plants as microbial populations decline at the end of the growing season, is up to 15 times greater than the soil mineral N pool. Microbial populations are also very responsive to changes in soil temperatures, including those simulated by the ITEX warming experiments. Because of this, it is very important to examine the microbial N pool as well, although this requires use of the complicated analytical protocol outlined in the next section.

Microbial Nutrient Immobilization – Chloroform Fumigation

In order to determine the amount of nutrients that are immobilized by soil microbes, soils are fumigated with chloroform to lyse microbial cell walls, thereby releasing the nutrients within.

The total microbial N is calculated by subtracting the nitrogen in un-fumigated soil extracts from that of fumigated soil extracts. This method can be used to obtain a single measure of the microbial Biomass N at any point in time, or it may be incorporated into the buried bag technique (described above) to examine the net change over time.

Materials and Methods:
Note: all lab materials (vials, beakers, graduated cylinders, containers, etc.) should be cleaned and acid washed before use.

Sample Preparation:

- Soil cores are removed from the site and sieved as explained above. Soil Collection and Preparation.
- Be Careful! If you are using this method in conjunction with the Buried Bag Incubations you will need enough soil for all steps of the analysis in BOTH cores (initial core, and the core that is incubated in the buried bag):
    - Gravimetric analysis
    - Available soil nutrients (un-fumigated)
    - Immobilized microbial + soil nutrients (fumigated)
- Complete the gravimetric analysis and extraction of un-fumigated portions of the core as explained above. Extraction Procedure and Calculations.
- Add 20g of the soil to be fumigated into a 50mL GLASS beaker.
- Label the beaker with PENCIL. Ink will run when exposed to chloroform.

Chloroform Fumigation:

- Line the bottom of a GLASS vacuum dessicator with damp paper towels.
- Stack the beakers with soil samples in the dessicator using chicken wire for additional levels if necessary.
- Because of the carcinogenic-volatile properties of chloroform, all fumigation work must be done in a fume hood. Wear a lab coat, goggles, and latex gloves.
- Add a thin layer of boiling chips to one of the glass beakers in the centre of the dessicator and pour ethanol free chloroform ($CHCl_3$) into the beaker so that it is no more than ¾ full.
- Seal the lid on the dessicator and slowly evacuate in the fume hood until the chloroform boils. ***This can be tricky. If the $CHCl_3$ boils too much it will explode and contaminate all of the samples. A trial run with no samples in the dessicator if you have never done this before is highly recommended.
- Continue boiling the $CHCl_3$ for 1 – 2 minutes, then release the vacuum and allow air back into the dessicator.
- Some publications recommend that the above procedure be repeated 2 more times. This forces all of the air out of the soil pores so that it is exposed to the $CHCl_3$ gas as much as possible. Attempt this in the trial run, as it may be difficult to boil the $CHCl_3$ more than twice depending on the strength of the vacuum.
- Do not release the vacuum on the last boil. Close the valve on the dessicator, then turn off the vacuum.
- Leave the samples sealed in the dessicator (in the fume hood) for 5 days. You will need to check the dessicator daily to ensure that it has remained sealed.

- After the incubation is complete, evacuate the dessicator for 3 minutes, 8 times. This will remove the $CHCl_3$ from the soil before you process the samples.
- Extract and analyze the sample as outlined in section above.

Calculations:
a) Microbial N:
= fumigated sample N – un-fumigated sample N
b) Net Microbial N Immobilization:
= Microbial N after incubation – Microbial N before Incubation**

* refers to buried bag incubations as described above
Note: These methods may also be used in conjunction with persulphate digestion to obtain SON immobilized in the microbial biomass.

6.4 Plant - Soil Nitrogen Cycling
Vegetation & Litter Quality – Vegetation C:N Analysis
Under warmer growing conditions, the nutrient use efficiency (NUE) of plants increases with greater productivity. The plant tissues become saturated with carbon (C), relative to nitrogen (N). As litter with high C:N is decomposed, less nitrogen is released and recycled back into the soil for plant and microbial uptake.

This negative feedback to increased productivity has been measured at some of the sites in the CANTEX network. These measurements should continue in order to explain longer term changes in the nutrient economy on these experimental sites

There are two scales that need to be considered while examining C:N ratios of vegetation. At the individual scale, the NUE of a single plant species may change affecting the quality of litter that falls to the ground from year to year. However, analysis of C:N ratios in conjunction with productivity data may indicate how changes in plant community composition over the longer term may be affecting the nutrient economy in these arctic sites.

Experimental Design:
Prior to the collection of samples during the growing season, the species and timing of collection must be carefully considered. Selecting a photometer species will allow for comparisons of changing NUE between several sites, while an analysis of the dominant functional types or species in each site may be more relevant to within site nutrient cycling. It may also be important to consider which species contribute the most litter (e.g. deciduous shrub) versus a species that be more dominant that does not drop its litter each season (e.g. evergreen shrubs).

Experimental warming, as simulated by OTCs, also leads to changes in phenology and, therefore, changes in the seasonal trend of C:N. However, the litter at the end of the growing season may be of particular interest with respect to changes in the nutrient economy. It would be great to address all of these research questions, BUT…these sampling methods are destructive to long term plots,

and we are only human! Create an experimental design that specifically addresses your research questions.

Sample Collection and Preparation:

- Collect vegetation in paper bags. The dry weight must be at least 0.2 g for analysis, so collect enough to yield this amount following drying an grinding.
- Include only the current year's growth. Previous year's litter (brown) is removed. Vegetative samples include the current years stem (fascicle) and leaves. Inflorescences include the stem, cauline leaves or bracts. Collect the flowers representing the dominant phenological stage at that sampling date.
- Dry the samples in the paper bags at a low temperature (~60˚C) for 48 hours, and store in a dry environment.
- Grind the sample with a Wiley Mill and weigh the final product. Ensure there is enough for analysis.
- The sample C and N contents are measured using a dry-combustion CN analyzer (e.g. Carlo-Erba NA 1500 elemental analyzer). The results are usually analyzed as %N, %C, and C:N.

Soil Organic Matter Quality – Soil C:N Analysis

In order to determine if changes in climate manipulations and/or litter quality (described above) have affected soil nutrient stores, soil C:N analyses may also be conducted.

Sample Collection and Preparation:

- While collecting the soil for CN analysis, the depth of the cores should be consistent between treatments and sites for comparison.
- Oven-dry the soil core at 60˚C for 48 hours.
- Sieve the sample through a 2.0mm mesh to remove rocks and twigs, and to homogenize the sample.
- Weigh the sample to ensure there is enough for analysis.
- The sample C and N contents are measured using a dry-combustion CN analyzer (e.g. Carlo-Erba NA 1500 elemental analyzer). The results are usually analyzed as %N, %C, and C:N.

6.5 Soil Decomposition Rates

Increased soil temperatures increase microbial breakdown and thus decomposition, provided that tissue quality (e.g. C:N ratio) is not altered substantially. Two methods are available to quantify decomposition, namely use of litter bags or wooden dowels (tongue depressors). Both methods involve determination of dry mass loss per given time interval. In the case of litter bags, fresh litter samples should be collected at the end of the growing season. These are weighed and placed in mesh bags (mesh size ca. 1mm) and exposed in the field. Make sure that bags are secured at the soil surface (use inert 'nails') and that replicates can be identified. In the case of wooden dowels replicates can be identified via permanent waterproof markers or metal tags. Use a minimum of 5 replicates to incorporate microsite variation.

7.0 Data Entry, Storage and Analysis

Sample data sheets for recording observations are provided in Appendix A but you are encouraged to develop your own to suit the specific purposes of your field site. Field data sheets are best photocopied onto water resistant paper (e.g. Rite in the Rain) to reduce the chances of damage to the sheet and loss of the data. This type of paper also makes data recording in rainy weather easier. Field recording forms should always have room for "Comments" to allow observers to record any factors that might impact the observations such as switching tags to a new plant, an unexpected disturbance at the site, etc.

It is very important to have the data entered comprehensively and consistently at the end of each field season. All too often in the past, data collected through hard work have been lost because they were not entered promptly and properly. The most efficient method is to set up a data entry form or spreadsheet that matches the data recording forms used in the field. This makes data entry easier and facilitates checking the accuracy of data entered with what appears on the form. Spreadsheets software usually allows multiple sheets so consider using the following format for each variable:

Sheet 1 – site description
Sheet 2 – relevant abbreviations and notes on data formatting
Sheet 3 – Data for all years compiled into one comprehensive table
Sheet 4 and subsequent – data for each individual year

In some cases, you will use your spreadsheet to calculate values. For example, you may want to calculate the average number of flowers counted in all plots. When using formulas in spreadsheets be very careful that the cells included in the formula are the ones you intended, that the formula has copied correctly to other cells, and that the function you have specified is the correct one. Once a calculation has been made, it is a good idea to convert the formula to the value of the result of the calculation. This will prevent confusion and errors if the source data are changed or moved.

Data files should be given appropriate names such as DL_phenology_dryint (Daring Lake CANTTEX site data on *Dryas integrifolia* phenology). Make sure that data are backed-up in at least two places (on a hard-drive, server, and on diskettes or CDs) and that in any change of the person or people responsible for those data, they are made aware of the status, format, and location of all collected data. Also, make sure that there is a standard location for storing original data sheets in case values need to be checked or to re-enter data if computer files are lost. It is a good idea to make photocopies of the original sheets and use the photocopies for data entry.

In our efforts to maintain the most up-to-date information on the status of CANTTEX sites, trends in data collected, and to minimize the possibility of data being lost, CANTTEX has set up a data archive to store and, when possible and appropriate, analyze data from the contributing sites. Once data has been entered at the end of each field season, please send the relevant files to Greg Henry (see section 10 Contact Information).

8.0 Frequently Asked Questions

Questions and problems that are communicated to CANTTEX will be addressed and posted on the CANTTEX website as they arise at http://www.emannorth.ca/. They will be included in this section in future editions of this manual.

If I can't find a tagged plant, what should I do?

-If one of your plants or tags goes missing, you will have to select another plant at random. Make sure to give it a new number so that you know that it is a different plant being monitored.

What should I do if I can't define an individual plant?

-For some mat and tussock forming species, it will be impossible to determine an individual. In these cases you should use wooden skewers and string to outline a 10 cm X 10 cm area that will be the unit of observation for that individual.

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10.0 Contact Information

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APPENDIX A: CANTTEX Taiga Monitoring Protocols – Small Scale

Compiled by
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Implemented in Mealy Mountains (Labrador) and Gros Morne NP (Newfoundland), July 2004.

Introduction:
One of the goals of CANTTEX is to monitor changes in the transition from Taiga-Boreal Forest to tundra ecosystems. The focus is to follow changes in the tree-line position and composition across Canada. The monitoring protocols outlined below have been developed to detect changes in biodiversity and structure of the taiga from closed canopy or dense forest to open non-treed area and tundra, as well as dynamics of the major floral elements (trees, shrubs, herbs, lichens/mosses). Key processes mediating change such as duff dynamics and disturbance regimes can also be monitored.

1. Establishment of permanent plots:
Objective: To establish a network of permanent plots that can provide information on decadal-scale changes in treeline community structure.
Methods:

- Establish circular plots across a gradient from closed canopy or the most dense tree stands, through to less dense open canopy tree stands, to tree islands, terminating in open treeless zones. Diameter of each plot will vary depending on tree density: 5 m in closed canopy and treeless zones, to 10-20 m in open canopy and tree islands. Each plot should be centered on a target tree, which should be clearly marked with a durable numbered metal tag, and GPS co-ordinates taken.
- A minimum of 2 (but preferably 3 or more) replicates should be included in each zone. Where appropriate, the plots should be established along parallel transects that cross through each zone of decreasing tree density. Establish additional replication within zones to encompass the landscape variability (e.g., aspect or moisture gradients). The location of each plot should be marked on topographical map and elevation, latitude and longitude should be recorded using a GPS unit.
- [Optional] Measure all soil surface or canopy disturbance (such as tree falls, insect damage or animal grubbing) along the transects in each zone. This can be done using the point-intersect method (record all disturbances that intersect the transect) or within a belt-transect (record all disturbances within 1m on each side of the transect). If the zones are too far distant to be connected along a single transect, establish individual transects within a zone that connect the circular plots. Be certain to record the total transect length and sampling method (point-intersect or belt-transect). Alternatively, these disturbances can be located with GPS for a wider spatial coverage (but be sure to measure the extent of each disturbance and note the total area surveyed).
- Conduct plot-based measurements (i.e., tree, shrub, understory etc. - see each section below) within or near each circular plot, as described below:

2. Tree measurements:

Objectives: (1) To record current stand age and size structure, and monitor future changes in stand recruitment, growth and mortality; and (2) To provide tree-core records that can be used to infer previous growth patterns in relation to climate or other environmental variation.

Methods:

- *All trees*
  - Measure the distance from the centre tree to all trees and tree seedlings in the plot (within the relevant radius around the central tree) and record the compass bearing (from the centre tree). Record the species and basal diameter (BD) or circumference (note which) just above the root collar for each seedling and DBH for larger individuals. Each tree should receive a tag with a unique number for the site. Note the age class of each individual (adult, height>1.4 m; sapling, height 15 cm – 1.4 m; seedling, height<15 cm).
- *Adult trees*
  - In addition to BD, measure diameter at breast height (DBH).
  - Count the number of female cones and record the presence of male cones. If cones are too numerous to count directly, establish a subsample to estimate the number of cones (for example, all cones visible on the western side of the tree - avoid counting only N or S aspects). Note the method used if sub-sampling.
  - Mature trees should be cored with an increment borer at 30 cm above the ground. Core from 10 - 25 trees (where possible) within a tree density zone (i.e., closed forest, open forest, treeline) to encompass the full range of size (DBH). These data can be used to generate size/age relationships. Because different parts of the forest/treeline ecotone are expected to have different size/age relationships, this coring procedure should be repeated at each location.
  - [Optional] Collect cones to measure size and seed weight. Cones should be collected into paper bags at the end of the season, and dried in a laboratory to extract the seeds. Viability trials can also be conducted to determine viable seeds/cone. Levels of insect predation (of seeds) can be determined.
- *Saplings*
  - In addition to BD, measure height from root collar or soil surface (note which).
  - If cones are present, record as with adult trees.
  - [Optional] If possible, determine whether the sapling is connected to an adult tree through layering or is a single stem from a seed. This may require some excavation.
- *Seedlings*
  - In addition to BD, measure height from root collar or soil surface (note which).
  - [Optional] To age seedlings, move OUT of the plot to adjacent area, chose 5 seedlings (where possible) of similar size as those in the plots. Measure BD and height, as done in the plot. Note the position of the moss/substrate surface on the stem, excavate the seedling, and measure height from root collar to the substrate surface. Section the stem at the root collar and count the growth rings (this may require a microscope and should be done in a laboratory with well-prepared

samples). This sampling is most critical in areas close to treeline, where aging young seedlings can provide information on recent recruitment episodes.

- *Singleton trees* [optional]
    - Tag all single trees and individuals in tree islands in areas of dispersed trees (along the monitoring transects). Note aspect, measure height, BD/DBH and elevation. Record GPS locations for each individual.

3. Large Shrubs [High priority]:

Objectives: To monitor changes in shrub cover over time, and to examine if treeline change is dependent on the prior presence of shrubs.

Methods:

- Locate large shrubs (>50 cm height) within the plot and measure distance and bearing from centre tree to the centre of the shrub cluster. Identify the species, and note whether the individual consists of single or multiple stems, and whether connected to other shrubs (i.e., are the shrubs all connected underground forming a "clump" rather than a discreet individual). If individual shrubs are not obvious (e.g., dwarf birch), count the number of single stems in the plot.
- Measure canopy crown size (length X width of canopy) and estimate percent cover within the crown. Measure maximum shrub or "clump" height (from ground surface).
- Note whether the shrub is reproductive with either flowers or fruits.
- Tagging of a minimum of 5 shrubs or single stems (if can't determine individual) is optional, but should be done where possible.

4. Understory vegetation cover:

Objectives: (1) to record changes in understory composition across treeline zones; (2) to monitor changes in understory composition over time; and (3) relate to treeline change.

Methods:

- Locate one $1m^2$ community composition point frame plot within the 5 m plot (this should not be in the plot centre, due to problems with trampling). The plot position can be determined by randomly selecting a distance and bearing from the centre point, or by selecting a single distance and bearing and using this in each 5 m plot. Plots should be established and marked following the standard ITEX point-frame protocol.
- Record all species, including mosses and lichens, following the standard ITEX point-frame protocol. Record all *small-scale* disturbances (<1 m) that produce bare ground and describe.
- Record any additional understory species that occur within the 5 m plot but were not found in the point frame plot.
- Additional 1 m x 1 m plots may be established to provide better representation of the ground cover, as described in the CANTTEX manual.

5. Soil sampling:

Objectives: To document variations in soil characteristics across treeline zones and among sites.

Methods:

- Soils are to be sampled outside the main sampling plot.

- Measure the litter (duff) depth in 3 places (choose 3 points spread evenly around the perimeter of the plot).
- [Optional] Sample soils by using a flat-bladed spade to remove a 15 cm x 15 cm block of organic and surface mineral soil. Using a serrated soil knife, trim the edges of the plug down to a 10 cm x 10 cm block (times whatever depth you have). Allow any unattached litter to fall off. Record the depth of the LFH organic layers (L=litter/moss/lichen; F=fibric material that is partially-decomposed, but fibrous when rolled between the fingers; H=humic material that is well decomposed and smears between the fingers), and the depth of the mineral soil A horizon if present (soil that is obviously coloured by organic material or surface leachates).
- Where possible, a soil pit should be dug to properly describe the soils at the site. Each soil horizon should be described for thickness, colour, and texture.
- [Optional] For more detailed information on soils, put each slice of soil into a separate, labelled bag to return to the lab. The soils can be air or oven dried to obtain bulk density (g dry soil/cm^3) and/or ashed in an oven to determine carbon content (loss-on-ignition). If possible, archive a portion of each sample. Where possible, pH and nutrient concentrations should be determined, and a portion of each sample should be archived

6. Seed bank [optional]:

Objectives: (1) To document variations in seed bank across treeline zones and among sites; and (2) to monitor changes in propagule availability over time, to determine if availability predicts future community composition and stability.

Methods:

- Prior to the initiation of seed bank studies, a voucher seed collection should be collected from the area.
- If possible, split cores from above section and determine seed banks (See "Germinable seed/propagule banks monitoring at ITEX site for background - NOTE may not be relevant for our studies - i.e., extracting to get numbers, rather than germinable banks-see next bullet).
- An alternative method is to remove seeds by washing the soil cores through a series of soil sieves to extract seeds of all sizes. Cores should be dried, large gravel removed and weighed prior to extraction. The seeds can then be separated from the soil particles under a dissecting microscope of magnifier, and identified using the voucher seed collection, and counted to obtain an estimate of each species per volume.
- Cores should be taken at the commencement of the plot and as often as possible, at least every 5 years.

APPENDIX B: Sample Data Sheets

What follows are sample data sheets that you can use for data entry, or use as a model to develop your own forms for use in the field. You might require more columns to differentiate between the different types of sites that you have or between experimental treatments. Blank column headings are there to customize forms such as including other climate variables being monitored or to fill in dominant plant species in the flower count field data sheet. It is recommended that you copy the forms onto water resistant paper (e.g. Rite in the Rain) to minimize the risk of forms being damaged in inclement weather. The data sheets include:

Annual Observations
Snow Course Data Sheet
Example of Phenology and Quantitative Observations Data Sheet
Permafrost Grid Data Sheet
Dates and Day Numbers
Random Number Table

Annual Observations

Year:	
Date arrived at site:	
Members of field crew:	

Snow and Ice

Snowmelt:

date (can be more than one if melts and is re-covered / re-frozen)

Snow 10% melted:	
Snow 50% melted:	
Snow 90% melted:	

Lake Ice:

Name of lake:	
Size of lake (length/width):	
Depth of lake:	

Thaw:

1. Open water on shore:	
2. Open water off shore:	
3. Ice in movement:	
4. No ice visible:	

Freeze-up:

1. Ice formation on shore:	
2. Ice cover on bays:	
3. Completely frozen:	

Sea Ice:

Name of water body	
Type of water body:	

Thaw:

1. Open water on shore	
2. Open water off shore	
3. Ice in movement	
4. No ice visible	

Freeze-up:

1. Ice formation on shore	
2. Ice cover on bays	
3. Completely frozen	

Snow Course

Site: Date:
Names of Observers:

Sample	Snow Depth (cm)	Weight – tube and core	Weight – tube alone	Water Equivalent (cm)	Core Length (cm)
1					
2					
3					
4					
5					
6					
7					
8					
9					
10					

General Observations on the snowpack (i.e. presence of ice layers, hoar frost layers, crust on surface, extent of drifting / clear patches, etc.):

Saxifraga Oppositifolia

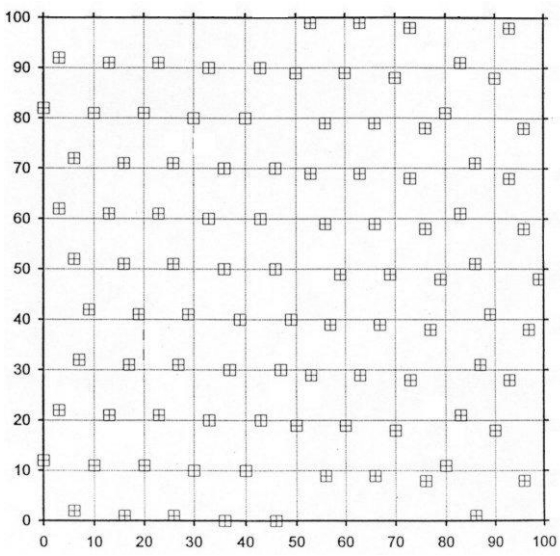
Year:		**Site:**	
Date arrived at site:		**Date left site:**	
Members of field crew:			

Site	Treatment	Plot	Plant	Snowfree Date	Flower bud break	Full Flower	Mature Fruits			# flower buds	# mature fruits	Comments

Permafrost Grid

Site name:	
Geographic coordinates:	
Elevation:	
Date measurements made:	
Measurements made by:	

	1	2	3	4	5	6	7	8	9	10
1										
2										
3										
4										
5										
6										
7										
8										
9										
10										



This grid shows the location in each grid square where the measurements should be made to avoid systematic sampling.

Notes:

Appendix C: Dates and Day Numbers

The Following shows the Day numbers that correspond to the dates in the summer months. The values given are for a normal year; during a leap year add 1 to each day number.

May	day number	**June**	day number	**July**	day number	**August**	day number
1	121	1	152	1	182	1	213
2	122	2	153	2	183	2	214
3	123	3	154	3	184	3	215
4	124	4	155	4	185	4	216
5	125	5	156	5	186	5	217
6	126	6	157	6	187	6	218
7	127	7	158	7	188	7	219
8	128	8	159	8	189	8	220
9	129	9	160	9	190	9	221
10	130	10	161	10	191	10	222
11	131	11	162	11	192	11	223
12	132	12	163	12	193	12	224
13	133	13	164	13	194	13	225
14	134	14	165	14	195	14	226
15	135	15	166	15	196	15	227
16	136	16	167	16	197	16	228
17	137	17	168	17	198	17	229
18	138	18	169	18	199	18	230
19	139	19	170	19	200	19	231
20	140	20	171	20	201	20	232
21	141	21	172	21	202	21	233
22	142	22	173	22	203	22	234
23	143	23	174	23	204	23	235
24	144	24	175	24	205	24	236
25	145	25	176	25	206	25	237
26	146	26	177	26	207	26	238
27	147	27	178	27	208	27	239
28	148	28	179	28	209	28	240
29	149	29	180	29	210	29	241
30	150	30	181	30	211	30	242
31	151			31	212	31	243

Appendix D: Random Number Table

The random number table below can be used when selecting plot locations or plants to be monitored.

10	27	53	23	54	04	26	47
28	41	50	88	83	39	94	89
34	21	42	02	80	05	84	46
61	81	77	23	53	44	42	28
61	15	18	54	90	07	52	59
91	76	21	64	75	39	56	29
00	97	79	06	53	01	30	48
36	46	18	94	78	08	67	25
88	98	99	50	91	43	46	02
04	37	59	21	69	92	55	91
63	62	06	41	16	29	79	30
78	47	23	90	54	12	14	23
87	68	62	43	66	59	50	36
47	60	92	77	95	48	61	12
56	88	87	41	44	50	81	33
02	57	45	67	68	77	06	75
31	54	14	17	67	46	14	01
28	50	16	36	67	24	59	96
63	29	62	50	86	86	92	48
45	65	58	51	74	44	44	12
39	65	36	70	76	30	49	61
73	71	98	04	79	79	08	94
72	20	56	11	77	97	09	89
75	17	20	76	48	26	53	87
37	48	26	29	89	06	45	47
68	08	60	72	01	02	97	17
14	23	02	67	17	10	18	99
49	08	98	44	67	19	41	72
78	37	96	43	65	09	56	16
37	12	06	68	76	44	91	58
14	29	34	04	47	87	59	25
58	43	09	36	10	05	12	09
10	43	28	70	00	90	43	14
44	38	67	54	07	17	38	81
90	69	88	51	20	11	78	95
41	47	59	62	31	68	84	45
91	94	10	19	56	79	14	40
80	06	14	66	34	22	24	31
67	72	54	48	19	80	75	24
59	40	77	27	09	53	85	38
05	90	24	95	20	37	71	71
44	43	35	98	31	77	59	66
61	81	80	82	98	55	08	57
42	88	31	05	24	27	10	12
77	94	07	39	01	49	96	91
78	76	30	16	50	23	88	09
87	76	19	81	90	18	57	72
91	43	59	47	46	01	17	73
84	91	05	73	47	41	91	16
87	41	77	83	12	73	36	59

APPENDIX E: Glossary

Albedo: The ratio of reflected solar radiation to total incoming solar radiation. It is a measure of the amount of solar radiation absorbed by the surface.

Androecium: The stamens of one flower collectively.

Biodiversity: The diversity of living organisms in an area including: habitat diversity, genetic diversity, and species diversity.

Biota: The animal and plant life of a region.

Cushion Plant: Refers to plants which have a rounded shape with low, packed branches

Clone: A set of organisms produced from one parent by vegetative reproduction.

Datum: A model of the earth used for producing maps. This information is found at the bottom of a map. An example datum is WGS-84.

Dioecious: Plant species having separate male and female flowers, sometimes on separate plants.

Dwarf Shrub: A low stature plant with woody stems.

Ecozone: An area with relatively homogeneous physical, geological and climatic processes that determine the physical and biological characteristics of the region. Arctic ecozones include: the arctic cordillera, northern Arctic, southern Arctic, taiga shield, taiga plains and taiga cordillera.

Endemic: Plants or animals only found in a particular region.

Ericaceous: A plant belonging to the Ericaceae family which includes Blueberry and Heather.

Forb: A non-woody plant other than a grass, sedge, rush, etc. (herb)

Graminoid: A plant belonging to the grass, sedge or rush family.

Gynodioecious: having bisexual flowers and female flowers, on separate plants.

Inflorescence: The flowering part of a plant.

Julian days: Days of the year with January 1 as Julian day 1.

Lithology: The type of rock that underlies the study area. This can most often be found on maps of the geology of the region.

Permafrost: Soil which is frozen for at least two years in a row.

Petiole: The stem or foot-stalk of a leaf.

Phenology: Phenology refers to the timing of the stages in the life cycle of an organism or phenomenon. Plant phenology, therefore, refers to the dates on which certain phases of a plant's life cycle are completed.

Quadrat: A square area used as a sampling plot or location.

Replication: Ensuring that a sufficient number of individuals or number of plots are being measured or monitored to be confident in the consistency of the results.

Stamen: The male part of a flowers reproductive structure.

Stevenson Screen: Standard housing for manual meteorological instruments. It consists of a ventilated, white box elevated 1 m from the ground surface.

Transect: A straight line along which observations are made at intervals.