VEGETATION SAMPLING:
STANDING CROP AND PRIMARY PRODUCTION

Robert L. Jefferies, University of Toronto
Gilles Gauthier, Université Laval
Esther Lévesque, Université du Québec à Trois-Rivières

PURPOSE

Primary production is at the base of all terrestrial food webs and is a key parameter determining the length of food chains, and the abundance of herbivores and carnivores at higher trophic levels. It is therefore an important parameter to measure at all our primary field sites. In terrestrial ecosystem, primary production is measured by sampling the vegetation. We are interested into two aspects of the vegetation. First, **standing crop**, which can be defined as the amount of live above biomass present at a given time (usually at the peak of the growing season). Second, **annual net primary production**, which is the amount of vegetation biomass that has been produced over the course of a growing season. In annual plants, standing crop and annual net primary production are often very similar but in perennial, the two can differ considerably, as the standing crop can represent the amount of live plant biomass that has accumulated over several years. In all ecosystems, production can occur both above and belowground. However, because belowground is exceedingly difficult and time consuming to measure, we will be concerned only by aboveground production (usually, green biomass).

Measuring primary production at the landscape level is very difficult due to the complexity of vegetation communities. At most sites, it will be desirable to stratify the landscape into different habitats. Habitat can be broadly defined as discrete patches recognizable by herbivores and usually characterized by different plant communities and physical characteristics (e.g. amount of moisture). Although it would be desirable to sample as many habitats as possible, resource and time limitation will force us to sample only a few (2 or 3) habitats at each site. In many cases, habitats may be reduced only to wetlands and mesic tundra, perhaps the simplest categorization. Even within these broad categories, we may be force to choose only one habitat subtype in areas where habitat heterogeneity is large. We should aim to choose habitats that are most important at each study site (in terms of either area covered or significance for the herbivores). Stratification by habitats has the advantage of facilitating comparison of primary production and standing crop among sites, while allowing an upscaling at the landscape level, providing we have an estimate of the relative area covered by each habitat. A disadvantage is that we cannot sample all habitats, hence some biases will occur.

This protocol details methods to use in order to sample standing crop and primary production in various types of habitat/plant communities. Another protocol will provide instructions on how to sample habitat availability at each site.

TIME PERIOD

Standing crop and primary production are usually measured at peak plant growth. Depending of the latitude, this may occur between mid July and early or even mid August. For some habitats, an additional sampling at the beginning of the plant growth may be required to accurately assess annual primary production. The habitat may consist of a number of vegetation types that may necessitate biomass sampling of the different types. Sampling and processing biomass samples are both labour-intensive and time-consuming.

METHODOLOGICAL CONSIDERATIONS

Minimal area

The empirical sizes of sampling plots for scoring cover and frequency of plant species are as follows for different types of plant communities:

Moss layer	0.01-0.1m^2
Herb layer	0.5-2 m^2
Low shrubs	4 m^2
Tall shrubs	16 m^2
Trees	100 m^2

Estimation of annual net primary production

Net primary production (NPP) during the interval from $t = 1$ to $t = 2$ is defined as follows:

$$NPP = \Delta B + \Delta H + \Delta E + \Delta D$$

where ΔB is the change in biomass, ΔH is the amount of biomass consumed by herbivores, ΔE is the amount of biomass lost from other causes, ΔD is the amount of biomass lost to death and detachment. The units are in grams of dried biomass m^{-2} yr^{-1}. In practice, each of the methods outlined in Appendix 1 is applied to either above- or below-ground NPP separately because the methods used for above- and below-ground NPP place a different emphasis on the components indicated in the above equation. Extensive variability in each of the estimates is to be expected, creating uncertainty. Taking into account the various constraints, measurement of below-ground changes in biomass is impractical as a general requirement for the different studies. A number of studies indicate that the two Methods outlined below provide an estimate close to values obtained from the use of more complex methods (see Appendix 1) where the perennial growth habit predominates at a site.

Method 1: The first method is based on **the difference between the minimum and maximum estimates of live above-ground biomass that gives an estimate of NPP**. The key assumptions are that some live material is carried over from previous intervals and that there is a single peak in live biomass.

Method 2: In some communities, especially graminoid communities composed only of one or two species, it may be possible to have **a single measure of live above-ground biomass as an estimate of NPP**, based on one harvest at peak standing crop in late July or early August, depending on the location.

The estimate of NPP is based on the difference in the amount of live biomass between the two harvests at the start of the growing season and in late July or early August in the first case and the value of above-ground standing crop in the second case.

If herbivory (ΔH) is important at a given site, **the methods may severely underestimate NPP**. Therefore, in order to avoid this bias, we recommend the use of herbivore exclosures whenever possible or at least where herbivory is thought to be non-negligible. We provide recommendation on how to set up exclosures at the end. A good general reference on this topic for grasslands is Scurlock *et al.* 2002.

PROCEDURE

In this section, we describe different procedures to sample vascular plants depending of the dominant type of vegetation. We also provide methods to sample mosses, as sampling this type of vegetation poses special difficulties.

1 - Sampling short-turf graminoid vegetation (no moss layer or only a thinly developed layer that can be ignored)

Examples of graminoid communities that fall into this category include *Puccinellia-Carex subspathacea* swards in intertidal marshes, *Festuca rubra-Calamagrostis deschampsiodes* swards, and *Dupontia fisheri-Poa arctica-Alopecuris alpinus* grasslands.

Field sampling

Decisions that need to be made

a) Size of individual samples (10 × 10 cm to 20 × 20 cm depending on plant density and time available)
b) Number of samples needed per site (6 to 12)
c) Sampling will either be carried out at peak biomass or at peak biomass and at the start of the growing season.
d) Make time available to process the samples (i.e. do not underestimate the time needed)

Equipment needed

- Notebook, 2 magic markers,
- Plastic bags
- Metre stick or ruler, measuring tape
- Wooden plant label sticks or equivalent to mark sampling quadrats
- Bread knife, trowel
- GPS (to position all sampling sites)
- Frame pack with shelves to place samples upon
- Cool dark places to store samples temporally (up to 24 hours)

The size of sampling frame for estimating the above-ground biomass should be between 10 x 10 cm and 20 x 20 cm. Cut the turves so that they are somewhat larger than the required size and then trim them to size in the laboratory with a bread knife. The turves can be 1.5 cm to 2.5 cm deep. Place each turf in a Ziploc Sandwich Bag and mark the outside of the bag with the appropriate codes using a magic marker. The number of replicates required depends on the homogeneity and density of the vegetation, but a minimum of 6 from each site is usually needed. The sites from which the samples are taken should be at least 30 m apart (avoiding pseudoreplication). As a guideline, we increase the sample size until the standard error of the mean of the aboveground biomass (dried) runs between 12% and 15 % or less. Avoid collecting samples from sites where there is evidence of disturbance (i.e. vegetative cover should be uniform), unless samples are needed from disturbed sites. Avoid squashing turves en route to the field laboratory. It is best to place the samples on a rack in a frame pack for transportation. If samples have to be placed one on top of the other, make sure that the vegetation surfaces are facing each other and no more than two samples are stacked together. On arrival at the field laboratory place the samples in a cool dark place (under a building, in a cooler, a refrigerator but *not* in a freezer). The samples should be processed within 24 hrs, otherwise there is a re-distribution of reserves leading to misleading results. Do place vegetation in a freezer until a later date as the sample will turn to mush upon thawing.

Processing of plant material

Decisions that need to be made

a) We suggest sorting the aboveground biomass into **functional groups** (e.g. grasses, sedges, etc). It may be possible to divide a given functional group further (e.g. grasses, *Eriophorum* and *Carex*) depending on time, expertise, and the objective. An alternative, simpler division could be between **monocotyledons** and **dicotyledons.** Dead biomass is discarded.

b) You should record those species that are flowering and the number of inflorescences of the different species per sample of turf. If the density of inflorescences is so low, it may be necessary to record the number for each species in a larger plot (50 cm × 50 cm) or simply record species that are flowering in the field.

Equipment needed

- Bread knife
- Ruler
- Filter paper or good quality paper towels (c. 15 cm diameter)
- Aquarium net frame (c.7.5 cm × 5.0 cm)
- Cheese cloth
- Needle and thread
- Small coin envelopes or larger envelopes and pencils
- Tray (c. 30 cm × 18 cm × 4 cm), soil waste bucket
- Drying oven with a convection heater if possible
- Fine-pointed dissection scissors and a pair of forceps
- Paper towels or a box of tissues

Above-ground plant tissue contains soil particles and dust, particularly if samples are collected close to the ground. Hence, all plant samples should be washed before the processing for dry weight estimates starts. In the case of very short turf characteristic of *Puccinellia phryganodes-Carex subspathacea* type swards, cut the above-ground biomass from a turf of known dimensions. Record the mean height of the turf of each replicate. A pair of fine surgical (dissection) scissors is very suitable for clipping above-ground vegetation (in addition, a ruler, bread knife, a waste bucket and paper towels are needed, the latter to clean the knife between sampling and also the researcher's hands!). Be careful not to cut into the underlying sediment or the thin moss later that may be present. The clippings can be placed in a tray of water about 4 cm deep. The soil will fall to the bottom of the tray. Make sure that you sample the edges of each turf and that clippings are not left on the scissors or on the sides of the tray. If the graminoid vegetation is taller (<15 cm), proceed as above, but cut the shoots at their base and do not cut the shoots into smaller sections as this will make the identification more difficult. There may be a low density of dicotyledenous species in turves. These plants also should be cut at the base of the shoot. If there is a shallow moss layer, the base of the shoots maybe embedded in this layer.

Gently swirl the water in the tray to allow soil particles to sink. With a pair of fine forceps remove obvious debris (insect cases, flies) from the surface of the water. Separate the live plant material in the tray into functional plant groups (mosses, graminoids, herbs, annuals, woody vegetation) or to lump into monocots, dicots and mosses. The separation into functional groups or equivalent can be done using forceps and the floating plant material is removed from the tray and placed on filter paper or good quality paper towels. After removal of dead material, an aquarium fish net can be used to scoop up the cut live plant pieces from the water in the tray. (Cheese cloth should be stretched tight across the metal frame [a rectangle] and sown to the frame using a needle and cotton thread). Plant material collects on the cloth and can be removed with a pair of forceps and placed in the appropriate plant category on a filter paper or

tissue. When complete, each sub-sample can be added to an envelope of a suitable size (coin envelopes are very useful for small samples) which is labeled with the date of collection, the site and, or treatment, the size of the turf, the type of plant sample and the name of the investigator.

All samples are oven-dried for 7 days up to 50°C depending on the type of plant material. Samples should not be heated at higher temperatures as this leads to a breakdown of plant tissue. If an oven is not available, samples should be air-dried for 7 days with the envelope flap open. Do not fold the filter paper tightly. Store samples in a cool dry location, otherwise mould may develop on the plant tissue. If there is no cool dry location, ship the samples to a dry site at the earliest opportunity. Upon return to a fully-equipped laboratory, dry *all* samples for a further 5 days before weighing. Be careful to check the envelopes for plant tissue that may have separated from the filter paper. Seal the envelope with Scotch tape either after the drying process or after the weighing (see below), so that pieces of tissue cannot fall through the flap slits and store (see above) until you weigh or analyze the tissues. Do not simply weigh the envelope and its contents and subtract the mean weight of an envelope, as there are very big differences between the weights of individual envelopes (when using such procedure, envelopes need to be weighed individually). If you plan no further analyses of the tissues, the samples can be discarded once the weight data have been entered and the records are complete. Remember once removed from the oven, envelopes, filter paper and plant tissues gain moisture rapidly from the air, hence do not leave samples outside the oven a long time before weighing.

2 - Sampling moderate to tall graminoid vegetation (height 15-70 cm) with or without a moss layer (mosses are considered below)

In a number of wetland habitats in the Arctic and sub-Arctic tall graminoid vegetation is widespread. Species that are abundant in this type of habitat and may form single-species stands include *Carex aquatilis, C. stans, Calamagrostis canadensis, Eriophorum angustifolium, E. spissum and E. scheuchzeri.* (Sampling of the moss layer and mixed graminoid-herb-woody vegetation appears below). Often these graminoids produce large amounts of above-ground biomass.

Field sampling

Decisions that need to be made

a) Size of individual samples (20 × 20 cm to 30 × 30 cm)
b) Number of samples needed per site (6 to 12 depending on the vegetational heterogeneity)
c) Sampling will either be carried out at peak biomass or at peak biomass and at the start of the growing season.

Equipment needed

- Sampling frame, metre rule
- Small garden shears
- Spade
- Polythene corer and piston plunger
- Large plastic bags and ties
- GPS (to position all sampling sites)
- Marking device for bags (e.g. magic marker and a code card that is placed in the bag)

The sampling frame should be 20 cm × 20 cm to 30 cm × 30 cm. Rather than use a fixed frame, we use four yellow potting label sticks to which are attached strings of the appropriate dimensions. This is less clumbersome in the field. Record the mean height of the vegetation with the metre rule. The above-

ground vegetation can be cut with the small shears. Make sure that the shoots are cut at the base which may necessitate pulling the moss layer aside to find the base of the shoot which is often white tissue below the chlorophylous tissue. If possible keep individual shoots intact and do not cut them into smaller units; this will make the sorting into functional groups much easier. Place in a large Ziploc bag or a kitchen bin liner bag. Again the number of replicates should be at least 6 samples collected over an appropriate area per site (i.e. each sample separated from the next by at least 30 metres). The SEM of the dried tissue should not normally exceed 15%.

Processing of plant samples

Decisions that need to be made

a) We suggest sorting the aboveground biomass into **functional groups** (e.g. grasses, sedges, etc). It may be possible to divide a given functional group further (e.g. grasses, *Eriophorum* and *Carex*) depending on time, expertise, and the objective. An alternative, simpler division could be between **monocotyledons** and **dicotyledons.** Dead biomass is discarded.

b) You should record those species that are flowering and the number of inflorescences of the different species per sample of turf. If the density of inflorescences is so low, it may be necessary to record the number for each species in a larger plot (50 cm × 50 cm) or simply record species that are flowering in the field.

Equipment needed

- A large drying oven with a convection unit if possible
- Large scissors, knife, soil buckets for discarded material and as a soil/peat trap if the turves are washed in a sink.
- Large lunch bags or envelopes
- Magic markers, Scotch tape
- Salad spinner, bowl
- Newspapers
- Fine-pointed dissection scissors and a pair of forceps

Place each sample on newspaper, remove standing dead and plant litter from the sample and separate the living biomass into functional groups or monocots and dicots. It is somewhat arbitrary whether a leaf or plant is "standing dead". If more than half of the leaves or stem are yellow or brown it is usually classified as standing dead.

Wash each sub-sample in a washing-up bowl of water for about 30 seconds (no longer). Drain the plant material of excess water. The sub-sample maybe placed in a salad spinner to remove additional water. Afterwards place the above-ground tissue in a fully-labeled brown lunch bag of appropriate size. Information on the bag should include the date of collection, the name of the investigator, the type of sample, site or treatment details and any other codes used by the investigator.

All bags can be placed in a drying oven with a convection fan for one week at a temperature not more than 50°C. If an oven is not available, the samples should be air-dried for one week with the bags fully open. All material should be further oven-dried at these temperatures, prior to weighing in a fully-equipped laboratory (see notes above).

The plant samples can be discarded after the data are entered, unless the tissues are needed for chemical analyses, in which case sub-samples of well-mixed plant material are placed in envelopes and sealed with Scotch tape. To prepare the sub-sample, cut up the entire sample into small pieces of stem or

leaves or reproductive organs and thoroughly mix the sample before taking a sub-sample of 10 grams which should be sufficient material for the different chemical analyses.

3 - Sampling of Mixed (Mesic) Tundra Vegetation (woody plants, graminoids, forbs and mosses). Mosses are considered below

This type of plant community poses considerable sampling difficulties on account of the range of plant functional groups present, particularly woody tissue and mosses. It is very difficult to estimate the net primary production of this type of vegetation, without considerable effort. These can be made at peak standing crop but on other occasions during the season when herbivory or browsing occurs.

Field sampling

Decisions needed to be made

a) The size of the sampling frame should be 20 × 50 cm or something similar
b) Number of samples needed per site (8 to 12).
c) Sampling will either be carried out at peak biomass or at peak biomass and at the start of the growing season. In this habitat where perennial are dominant, we strongly recommend sampling at both the beginning of the season and at peak plant biomass.

Equipment needed

- Small pair of garden shears
- Pair of secateurs (pruner), good quality spade
- Sampling frame, metre rule
- Large Ziploc bags or kitchen bin liner bags with plastic ties
- GPS (to position all sampling sites)
- Notebook, pencils, magic markers

We suggest that the dimension of the sampling frame is between 20 cm × 50 cm Because of the vegetational heterogeneity, a sufficiently large number of samples (c. 8-12) is needed to maintain a S.E.M of about 15% of the total mean dry weight. Record the mean height of the vegetation. The above-ground vegetation needs to be cut with care using the secateurs to remove woody tissue, and the small shears to remove graminoid species and forbs. All plants should be cut at the base, so that individuals remain intact as far as possible that will make the sorting into functional groups easier. In the case of woody plants that are layered (strong horizontal clonal growth), the stems may have to be lifted carefully from the moss and the organic peaty layer. Do not rip them from the moss/peaty layer. Place the sample in the kitchen bin liner bag.

Processing of plant material

Decisions that need to be made

a) We suggest sorting the aboveground biomass into **functional groups** (e.g. willows, grasses, rushes, etc). It may be possible to divide a given functional group further depending on time, expertise, and the objective. An alternative, simpler division could be between **monocotyledons** and **dicotyledons.** Dead biomass is discarded.

b) You should record those species that are flowering and the number of inflorescences of the different species per sample of turf. If the density of inflorescences is so low, it may be necessary to record the number for each species in a larger plot (50 cm × 50 cm) or simply record species that are flowering in the field.

Equipment needed

- Newspaper sheets
- Envelopes, lunch bags
- Magic markers, Scotch tape
- Notebook, pencils
- Large bowl, salad spinner (optional), soil waste bucket, soil trap bucket for the sink
- Large scissors, shears
- Oven with a convection unit

Spread each sample out on newspaper sheets. Remove plant litter and standing dead from the material. The above-ground living biomass should be sorted into functional groups. In the case of woody tissue separate leaves and non-woody organs from wood. (Bag them separately). Each plant component should be washed in a bowl for about 30 seconds and the excess moisture shaken-off. The material can be placed in a salad spinner to remove further surface water.

The samples can be bagged and the lunch bags marked as described above and the samples oven-dried for 7 days at temperatures up to 50°C. If an oven is unavailable, the samples can be air-dried for 7 days with the bags fully open. After this drying period, seal all bags with Scotch tape and store the samples in a dry cool place. If this is unavailable, ship the samples to a dry location at the earliest opportunity. Re-dry the samples in a well equipped laboratory at the above temperatures before weighing. If samples are required for chemical analysis, follow the procedures under (4) above.

4 - Biomass measurements of mosses

Equipment needed

- 12 stainless steel rods per site bent to give two vertical sections and a horizontal section or small twist-ties or marker ink
- Serrated bread knife
- Ziploc bags
- GPS (to position all sampling sites)
- Notebook, pencils
- Magic markers

"Typically, production of monopodial mosses such as *Sphagnum* spp., *Pleurozium schreberi* and *Ptilium crista-castrensis* has been measured using cranked wires (Clymo 1970), netting (Bond-Lamberty *et al*. 2004) or marking of the stem (Rochefort *et al.* 1990, Camill *et al.* 2001, Vitt 2007) to assess annual vertical growth increment of the plant. Vertical growth is multiplied by a species-specific bulk density, obtained by destructively sampling the upper portions of the plants to provide an annual estimate of net production." (Besscoter & Vitt 2007). However, feather mosses also grow laterally, extending the length of the previous year's branches. The use of bulk density accounts for new stem production and branch growth on the stems, but does not include lateral growth of the previous year's branches. This leads to a serious underestimate of total annual production in the feather mosses (c. 25%) (Besscotor & Vitt 2007). Hence, the method used to estimate annual production must be picked with care.

Cranked stainless steel wires (shaped like a car or tractor starting handle in former times) are prepared. One end of the wire that is about 10 cm in length is pushed vertically (or parallel to the moss stems if they are not vertical) into the moss carpet. The horizontal section of the wire is about 1 cm long and the wire is pushed into the carpet so that the horizontal section is level with the top of the shoots or with the capitula. The free end, the length of which must be exactly known projects vertically into the air.

The moss plants grow up around the vertical free end of the wire and at some designated interval of time, the growth is measured from the amount of wire still projecting above the moss surface.

The quantity measured is an increase in length and to estimate NPP this length must be multiplied by the average mass of plants in the unit depth of the moss carpet. The method is useful for annual measurements where the growth in length is about 2 cm. Where the moss plants grow horizontally, the method is of little value. A small plastic white disc (2 cm in diameter) can be fitted over the free end of the wire and the disc serves to define the surface. Coloured PVC can be attached to the top of the free end of the wire allowing easy recognition. Another approach is to attach thread from a fixed post to the moss stem and measure the increase in the length of the stem. A difficulty with this method is that the disturbance to make the measurement of length causes the stems to dry out. Alternative measures are the use of natural markers or markers applied to the stem to record incremental growth (see Rochfort 1990). Again the estimate of production must be based on the bulk density of shoots and the incremental growth (see below). Some plant ecologists simply record the amount of green biomass.

The bulk density of the total vertical growth of moss beyond the horizontal section of the wire can be estimated by destructive sampling. Remove a known area of moss with a serrated knife and sample 30, 3 cm stem sections from randomly chosen individuals to estimate mass per unit length (Rochefort *et al.* 1990), NPP is calculated by multiplying density per unit area, per capita mass per unit length, and the growth increment. After cutting the samples are dried at 50 ^{0}C in an oven for 5 days. The result can then be expressed as g m^{-2} yr^{-1}.

5 - Measurements of vascular plant standing crop in grazed and ungrazed swards

If grazing is intense, the only method to obtain an estimate of NPP is to measure aboveground standing crop inside exclosures. In this case, sampling outside exclosures (i.e. paired samples inside and outside exclosures where grazing occurs) may provide a measure of offtake by herbivores. However, this method is likely to work only if grazing is relatively high as light grazing is unlikely to be detected considering spatial variability in plant biomass.

Equipment needed

- Rolls of chicken wire (1-inch mesh) or the equivalent
- Bundles of laths or posts (1" x 2" piece of wood)
- Wire cutters and pliers
- Heavy-duty staples and staple gun (tie wrap or steel wire is an alternative)
- Measuring tape
- Magic markers, pencils, notebook to record details of construction and positioning.
- Good-quality spade
- Wooden mallet, hammer or small sledge hammer to drive in laths or posts
- Raspberry cane netting and/or string (optional)
- GPS (to position all sampling sites)
- 4 nails (10-inch long) per exclosures with steel wire (in areas where permafrost prevents hammering posts in the ground in spring).

The construction of the exclosures depends upon the length of time the study will be undertaken, the foraging and behavioral habits of the herbivore and logistic and permit concerns. The exclosures should be at least 50 metres apart. Some types of exclosure can be constructed at the base camp, flattened and transported in a helicopter. Alternatively, they can be constructed at the study site. Exclosures should be set as early as possible after snow-melt and are usually taken down at the time when plants are sampled at peak production. Exclosures should not be left standing up overwinter and new exclosures

should be set every year. Vegetation inside exclosures that are left standing up for several years may change due to the permanent removal of grazers. Although this in itself is an interesting question, it is not appropriate to measure annual plant production.

In the case of geese and hares, chicken wire (1-inch mesh) can be used to exclude these herbivores from a site. The height of the wire is about 18" (48 cm), which means that a standard roll of chicken wire (36" wide) can be cut down the middle with wire cutters. The chicken wire is stapled to the *outside* of four corner wooden lath sticks (an alternative is to use tie warp or steel wire). The dimensions of the exclosure are usually 1 × 1 m or less (see below). Leave a space at the bottom of each lath so that they can be sunk into the ground with the wire flush with the ground. It may be necessary to put either strings or raspberry cane netting across the top of the exclosure to keep the geese out at some sites. Both the strings and the netting are tied to the chicken wire. The chicken wire should be anchored to the ground with tent pegs or large stones to stop the geese from entering the exclosure. If the ground is frozen, you may not be able to hammer down the posts. In that case, you can brace each corner with a guy wire anchored to the ground by a 10-inch nail (nails can be hammered in frozen ground). It is important that exclosures be inspected regularly (in some cases, every few days) to ascertain that they are not damaged by caribou or polar bears.

In the case of other herbivores, the construction and placement of the exclosures will be different. For lemmings, although the 1-inch chicken wire may impede their movement to some extent, lemming-proof exclosures will require a smaller mesh size (e.g. ½-inch welded wire). In addition, the wire should be sunk into the ground to a depth of 10 cm using a spade to construct a trench. An exclosure size of 1 m X 1 m or less is satisfactory, but the size will depend on the total number of turves that will be taken from each exclosure during the study. If large herbivores such as caribou are being studied, the exclosure posts need to be sturdy and heavy-duty thickened wire about 1.3 m in height above the ground is required. Again the wire netting should be sunk in the ground to a depth of 25 cm. Short-term exclosures are not very practical in these situations.

REFERENCES

(see also Appendix II and note the publication in which D. Vitt's paper appears)

Benscoter, B.W. & Vitt, D.H. 2007. Evaluating feathermoss growth: a challenge to traditional methods and implications for the boreal carbon budget. *Journal of Ecology*, **95**, 151-158.

Bond-Lamberty, B., Wang, C. & Gower, S.T. Net primary production and net ecosystem production of a boreal black spruce wildfire chronosequence. *Global Change Biology*, **10**, 473-487.

Camill, P., Lynch, J.A., Clark, J.S., Adams, J.B. & Jordan, B. 2001. Changes in biomass, aboveground net primary production, and peat accumulation following permafrost thaw in the boreal peatlands of Manitoba, Canada. *Ecosystems*, **4**, 461-478.

Clymo, R.S. 1970. The growth of *Sphagnum*: methods of measurement. *Journal of Ecology*, **58**, 13-49.

Rochefort, L., Vitt, D.H. & Bayley, S.E. 1990. Growth, production, and decomposition dynamics of *Sphagnum* under natural and experimentally acidified conditions. *Ecology*, **71**, 1986-2000.

Scurlock, J.M.O., Johnson, K. & Olsen, R.J. 2004. Estimating net primary productivity from grassland biomass dymanics measurements. *Global Change Biology*, **8**, 736-753.

Vitt, D.H. 2007. Estimating ground layer net primary production in tundra, peatlands and forests. *Principles and Standards for Measuring Net Primary Productivity in Long-term Ecological Studies* (ed T. Fahey), in press. Oxford Press, Oxford.

Floras

Porsild, A.E. & Cody, W.J. (1980) Vascular Plants of Continental Northwest Territories, Canada. National Museum of Natural Sciences, National Museums of Canada, Ottawa.

Hultén, E. (1968) Flora of Alaska and Neighboring Territories. Stanford University Press

APPENDIX 1 – Details of methods for measuring NPP

Method 1
NPP is estimated by peak above-ground live biomass **as given above**

Method 2
NPP is estimated as peak above-ground standing crop (live plus dead biomass). The key assumption here is that some live biomass produced during the interval of interest may senesce before sampling.

Method 3
This method is based on the difference between the minimum and maximum estimates of live above-ground biomass that gives an estimate of NPP **as given above**. The key assumptions are that some live material is carried over from previous intervals and that there is a single peak in live biomass.

Method 4
All of the positive increments in live increments in live above ground biomass are summed to estimate NPP. It assumes that live biomass is carried over from previous seasons and that there may be multiple peaks of biomass during the study interval that reflects the phenologies of individual species.

Method 5
The sum of positive increments in live biomass, standing dead biomass, and litter is used to estimate NPP. An increment in live biomass and an increment in total dead biomass, are both counted. This method makes the same assumptions as Method 4 with the additional assumption that live material may be transferred to the dead biomass or to the litter during the interval of interest.

Method 6
This method sums the changes in live and dead biomass (positive or negative) and adds to that the amount of material that decomposed during the interval.. The additional assumption is that some litter produced during the interval of study may decompose.

If herbivory (ΔH) or exudation (ΔE) are important (see Section 9), all of the six methods may underestimate NPP. The method that is chosen will depend on the types of vegetation that need to be surveyed, other priorities and help and time available for estimating NPP. Depending on objectives, different methods may be used by different investigators. A good general reference on this topic for grasslands is Scurlock *et al.* 2002.

Appendix II: Vegetation Types

This brief account is based on the following reference:

> Bliss, L.C. & Matveyeva, N.V. (1992). Circumpolar Arctic Vegetation. In: **Arctic Ecosystems in a Changing Climate.** (eds F.S. Chapin, III, R.L. Jefferies, J.F. Reynolds, G.R. Shaver, J. Svoboda). Academic Press, New York.

The general operational classification is as follows:

Forest-Tundra

Low Arctic Vegetation

a) Shrub Tundras
b) Sedge-Dwarf Shrub Tundra
c) Steppe Tundras
d) Tussock-Dwarf Shrub Tundras
e) Mires
f) Coastal Salt Marshes

High Arctic

a) Mires
b) Polar Semi-Deserts and Polar Deserts

Most of these major types are sub-divided and the descriptions appear in the Chapter.

For those working on **Herschel Island,** at **Alert** or at **Eureka,** the following two references are of particular value. There are accounts of plant communities and standing crop values. A full list of contents appears on line if you type in the title of the first publication.

Svoboda, J. & Freedman B. (eds) (1994). **Ecology of a Polar Oasis, Alexandra Fiord, Ellesmere Island, Canada.**
(It can be obtained on line from Captus University Publications, ISBN 978-1-895712-26-1. The price is CAN $39.00).

Bliss, L.C. (ed.) (1977). **Truelove Lowland, Devon Island, Canada: A High Arctic Ecosystem.** University of Alberta Press, Edmonton.