CONCEPTUAL DESIGN AND SAMPLING PROCEDURES OF THE BIOLOGICAL PROGRAMME OF NUUKBASIC

NERI Technical Report no. 745 2009

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Peter Aastrup[1]
Josephine Nymand[2]
Katrine Raundrup[2]
Torben L. Lauridsen[1]
Paul Henning Krogh[1]
Niels Martin Schmidt[1]
Lotte Illeris[3]
Helge Ro-Poulsen[3]
[1]National Environmental Research Institute, Aarhus University
[2]Greenland Institute of Natural Resources
[3]University of Copenhagen, Department of biology

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Data sheet

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Institutions:	[1]National Environmental Research Institute, Aarhus University [2]Greenland Institute of Natural Resources, [3]University of Copenhagen, Department of biology – Terrestrial Ecology
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Abstract: This manual describes procedures for biologic climate effect monitoring in Kobbefjord, Nuuk. The monitoring is a part of NuukBasic which is a cross-disciplinary ecological monitoring programme in low Arctic West Greenland. Biological monitoring comprises the NERO line which is a permanent vegetation transect, and monitoring reproductive phenology of *Salix glauca, Loiseleuria procumbens, Eriophorum angustifolium,* and *Silene acaulis*. The progression in vegetation greenness is followed along the vegetation transect and in the plant phenology plots by measurement of Normalized Difference Vegetation Index (NDVI). The flux of CO_2 is measured in natural conditions as well as in manipulations simulating increased temperature, increased cloud cover, shorter growing season, and longer growing season. The effect of increased UV-B radiation on plant stress is studied by measuring chlorophyll fluorescence in three series of plots. Arthropods are sampled by means of yellow pitfall traps. Microarthropods are sampled in soil cores and extracted in an extractor by gradually heating up soil. The rate of decomposition is measured in three habitats. The avifauna is monitored with special emphasis on passerine birds. Only few terrestrial mammals occur in the study area. All observations of mammals will be recorded ad-hoc. Monitoring in lakes include ice cover, water chemistry, physical conditions, species composition of plankton, vegetation, bottom organisms and fish. Physical-chemical parameters, phytoplankton and zooplankton are monitored monthly in the period when the lakes are ice-free.

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Executive summary

Below is given a short overview of all monitoring elements. The following sections give detailed descriptions of each element.

Plants

Table 1 gives an overview of the monitoring of plants.

Table 1. Overview of monitoring elements

Monitoring element	Species	Number of plots	Sampling frequency	Sampling object	Sampling period
NERO-line	Plant communities and species		Every 5th year		
Phenology	*Salix glauca*	4	Weekly	Buds, flowers, and seeds	May - October
	Loiseleuria procumbens			Buds, flowers, and seeds	May - October
	Silene acaulis			Buds, flowers, and seeds	
	Vegetation analysis - pin point analysis - to be done	4 per plot	Every 5th year	Plot - four sections per plot	-
Total count of flowering shoots	*Salix glauca* *Loiseleuria procumbens* *Silene acaulis* *Eriophorum angustifoliu*m		Once per season at peak flowering	Flowers	Depending on phenology
NDVI	Along vegetation transect		Monthly		May - October
	In phenology plots	20	Weekly		May - October
CO_2 - Flux	Control, C Increased temperature, T Shading, S Hessian tents Long growing season, removal of snow, LG Short growing season SG, addition of snow during spring	6 plots per treatment	Weekly	Plot	May - October
UVB	Control Mylar film (0,25 mm) Filter control (Teflon)	5	3	Plots	Weekly, week 29-31
Arthropods	All Taxonomic groups	4 plots - 8 sections	Weekly	Specimens	May - October
Micro-Arthropods	Collembolan species Orbatid and actinedid mites and others	3 plots	3 times per season	Specimens	June - September
Birds	Passerines etc.	13 census points	Weekly		May - October
	Ad hoc (Passerines)	Nests			
Mammals	Ad hoc				
Lakes	Water chemistry, Chlorophyll a, phytoplankton, zooplankton	Two lakes	5 times per season		June - October (ice free period)
	Flora		Every year		
	Fauna		Every 5th year		
	Arctic char, Stickleback				
	Isotopes				

The NERO-line

The NERO line is a permanent vegetation transect which was established in July 2007 in order to monitor future changes in the distribution and composition of vascular plant species in the plant communities. Surveys of the transect will take place with 5 year intervals. It is intended to include mosses and lichens in the monitoring programme in the following years.

The concept relies on the assumption that changes in the distribution of plant communities can be seen by changes of boundary lines between vegetation zones. Therefore each boundary between vegetation zones has been marked by pegs. The species composition of the vegetation zones has been documented by Raunkjær analyses. Immigration of new species is assumed to be documented by the surveys with five year intervals. The concept is also used for the ZERO-line in Zackenberg in high arctic North East Greenland (Fredskild & Mogensen 1996, Bay 2001, 2006).

Movement of zones is documented by the position of pegs, while changes in species composition are recorded by Raunkjær–analyses.

Reproductive phenology

It is expected that plant phenology will give an early and distinct response to climate change. This has already convincingly been shown in Zackenberg. In Nuuk we follow four species: *Salix glauca*, *Loiseleuria procumbens*, *Eriophorum angustifolium* (only total flowering) and *Silene acaulis*. These species were chosen because they are widely distributed in the area and they cover a spectrum of different growth forms (deciduous dwarf shrub, evergreen dwarf shrub, graminoids and cushion forming herb), and they are comparable to species monitored in Zackenberg.

For each species four observation plots were established. The specific sites of the plots were chosen in order to cover the ecological amplitude of the species with respect to duration of snow cover, difference in soil moisture at the site and altitude. The size of each plot varies depending on the abundance of individual flowering shoots of the species in question.

Total flowering

Total flowering in the reproductive phenology plots is followed for *Salix glauca*, *Loiseleuria procumbens*, *Eriophorum angustifolium,* and *Silene acaulis*. The number of flowers is counted at peak flowering as the total number of buds, flowers/catkins and senescent flowers/catkins.

Normalized Difference Vegetation Index (NDVI)

The progression in vegetation greenness is followed along the vegetation transect and in the plant phenology plots by measuring NDVI with a scanner. NDVI is used as an index of plant production and vigorousness. The scanner measures the spectral reflectance of the plant canopy.

CO_2 flux plots

The CO_2 flux is important for understanding the balance between CO_2 emission and uptake. This study aims at documenting the present state, but it will also provide data from manipulations simulating increased temperature, increased cloud cover, shorter growing season, and longer growing season.

UV-B exclusion

UV-B radiation will increase as a result of the expected depletion of the ozone layer in the atmosphere. We monitor the effect of increased UV-B radiation on plant stress is monitored indirectly by measuring chlorophyll fluorescence in three series of plots: Controls, plots with a filter excluding UV-B, and filter controls with film without exclusion of UV-B. Measurements of chlorophyll fluorescence are carried out on *Betula nana* and *Vaccinium uliginosum* in a mesic dwarf shrub heath dominated by *Empetrum nigrum* and with *Betula nana* and *Vaccinium uliginosum* as subdominant species.

Arthropods

Arthropods are sampled by means of yellow pitfall traps. The traps are emptied weekly throughout the summer season (early June – late September). At the time being, samples are stored at Greenland Institute of Natural Resources (GINR) awaiting final decision of the future of the programme. Presently there is no financing for determination of the sampled specimens.

Microarthropods and decomposition

Microarthropods are sampled in soil cores from which the organisms are extracted in an extractor by gradually heating up. Microorganisms are determined at NERI, Dpt. of Terrestrial Ecology.

The rate of decomposition of filter paper is measured in the microarthropods plots.

Birds

The avifauna is monitored with special emphasis on small passerine birds representing the highest trophic level. Breeding phenology (first egg dates, hatching, fledging) is monitored throughout the season on an ad hoc basis.

Weekly counts of birds are carried out at census points during the entire season from May untill all the birds have left the area in August-September. Other bird observations are recorded ad-hoc during the entire field season.

Mammals

Only few terrestrial mammals occur in the study area: Arctic fox *Alopex lagopus*, arctic hare *Lepus arcticus*, and caribou *Rangifer tarandus*. The first year's observations, however, indicate that mammals are not observed very often. In 2007 only arctic fox was actually observed, and there were no new signs like footprints or droppings of caribou. In 2008 two caribou were observed at one occasion and fresh foot prints were also observed during the season. All observations of mammals will be recorded ad-hoc. If arctic fox dens are discovered, reproduction will be followed.

Lakes

The two sampling lakes are located in the Kobbefjord catchment area in the bottom of Kobbefjord (Badesø 64º07'48''N, 51º21'23'' W and Qassi Sø, 64º09'11''N, 51º18'18'' W).

Monitoring include ice cover, water chemistry, physical conditions, species composition of plankton, vegetation, bottom organisms and fish. Physical-chemical parameters, phytoplankton and zooplankton are monitored monthly in the period when the lakes are ice-free.

Resume

Nuuk Basic programmet blev startet i 2007 af Danmarks Miljøundersøgelser ved Aarhus Universitet i samarbejde med Grønlands Naturinstitut, Københavns Universitet og Asiaq (Grønlands forundersøgelser). Programmet finansieres af Energistyrelsen og Miljøstyrelsen som en del af Miljøstøtten til Arktis (DANCEA-programmet). Nuuk Basic dokumenterer og undersøger effekter af klimaændringer på marine og terrestriske økosystemer og består at 4 delprogrammer: MarinBasis som varetager, det marine miljø, BioBasis som indsamler biotiske parametre, samt Geobasis og KlimaBasis, som indsamler abiotiske parametre.

Denne manual beskriver de metoder og indsamlingsprocedurer, der anvendes i Biobasis. Manualen vil løbende blive opdateret på www.nuuk-basic.dk . Moniteringen i Biobasis omfatter bl. a. udvalgte planters reproduktive fænologi, vegetationens grønhed (NDVI), CO_2 flux, arthropoder, mikroarthropoder, nedbrydning af organisk materiale, fugle samt sø-økologi.

1 Introduction

The programme is run by the National Environmental Research Institute, Aarhus University, in cooperation with the Greenland Institute of Natural Resources, University of Copenhagen, and Asiaq (Greenland Survey). BioBasis is funded by the Danish Energy Agency and the Danish Environmental Protection Agency as part of the environmental support programme DANCEA – Danish Cooperation for Environment in the Arctic. The present manual describes methods and sampling procedures. The manual will be updated regularly. The latest version can always be found here: www.nuuk-basic.dk.

Nuuk Basic is a climate change effects monitoring programme close to Nuuk in west Greenland. The programme studies the effects of climate variability and change on marine and terrestrial ecosystems. In terms of scientific concept, Nuuk Basic copies the investigations carried out in Zackenberg Basic, at Zackenberg Research Station in Northeast Greenland (www.zackenberg.dk).

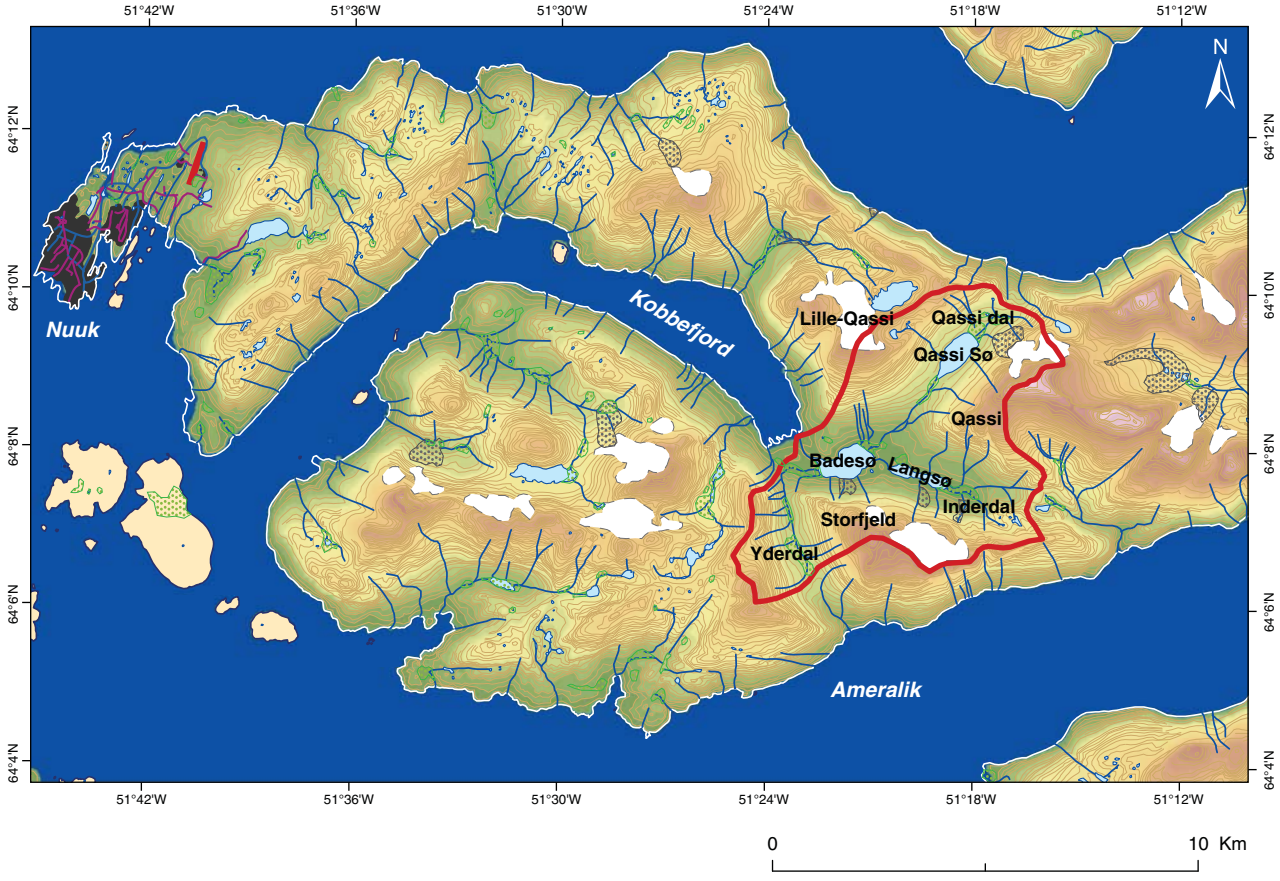


Figure 1. The study area.

The study area is situated app. 20 km east of Nuuk as seen on figure 1 and 2. The local climate is low arctic with a mean annual temperature of -1.4 °C and a mean annual precipitation of 752 mm (1961-90). The drainage basin is situated in an alpine landscape with mountains rising up to 1400 meter above sea level and with glacier coverage of approximately 2 km^2. Geologically, the area is homogenous with Precambrium gneisses as basement throughout the drainage basin.

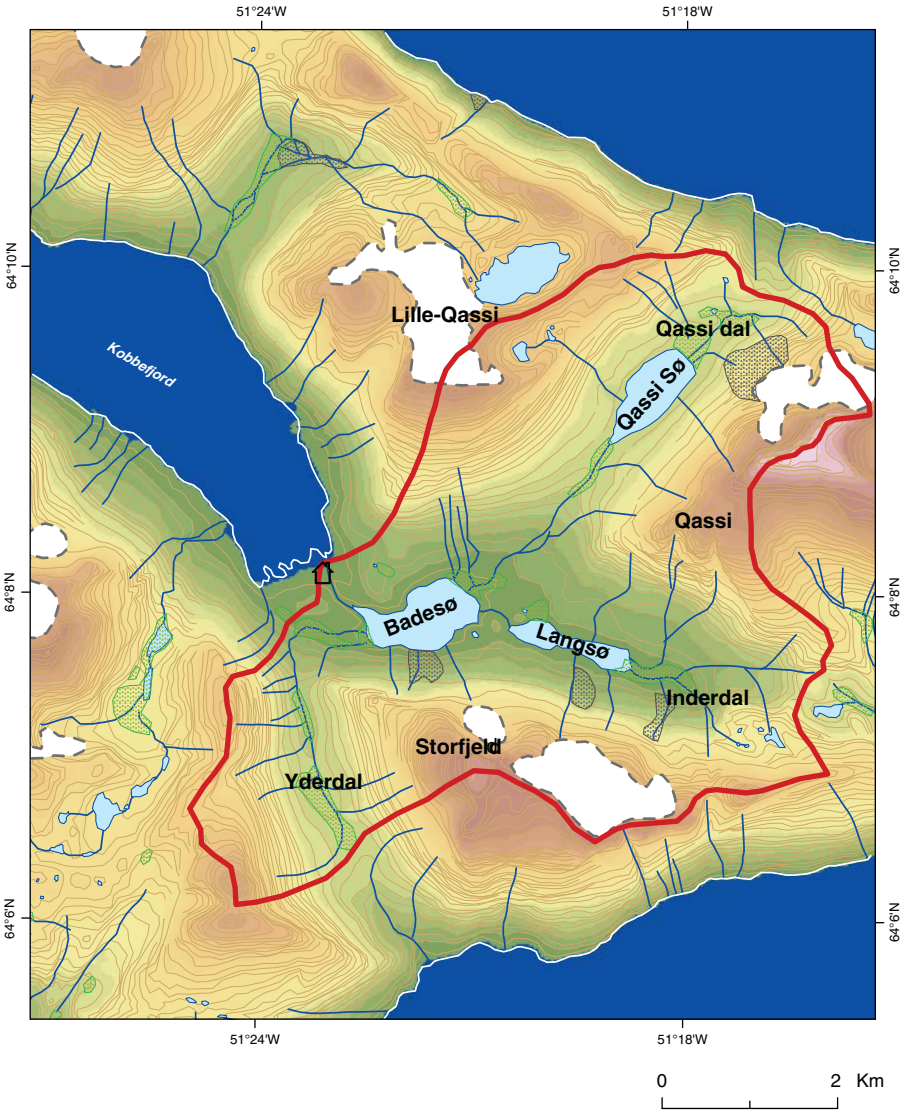


Figure 2. Close-up of the Bio-Basis study area

2 Detailed manual

Appendix 1 gives an overview of the activities during the monitoring season in Kobbefjord. In the following procedures are described in detail for all monitoring elements.

2.1 Plants

2.1.1 The NERO line

The NERO line was established in 2007. It is described in detail in Bay et al. (2008 - http://www2.dmu.dk/Pub/FR693.pdf). Next survey of the line is expected in 2012. The location of the line is shown in figure 3. See also Bay et al. 2008).

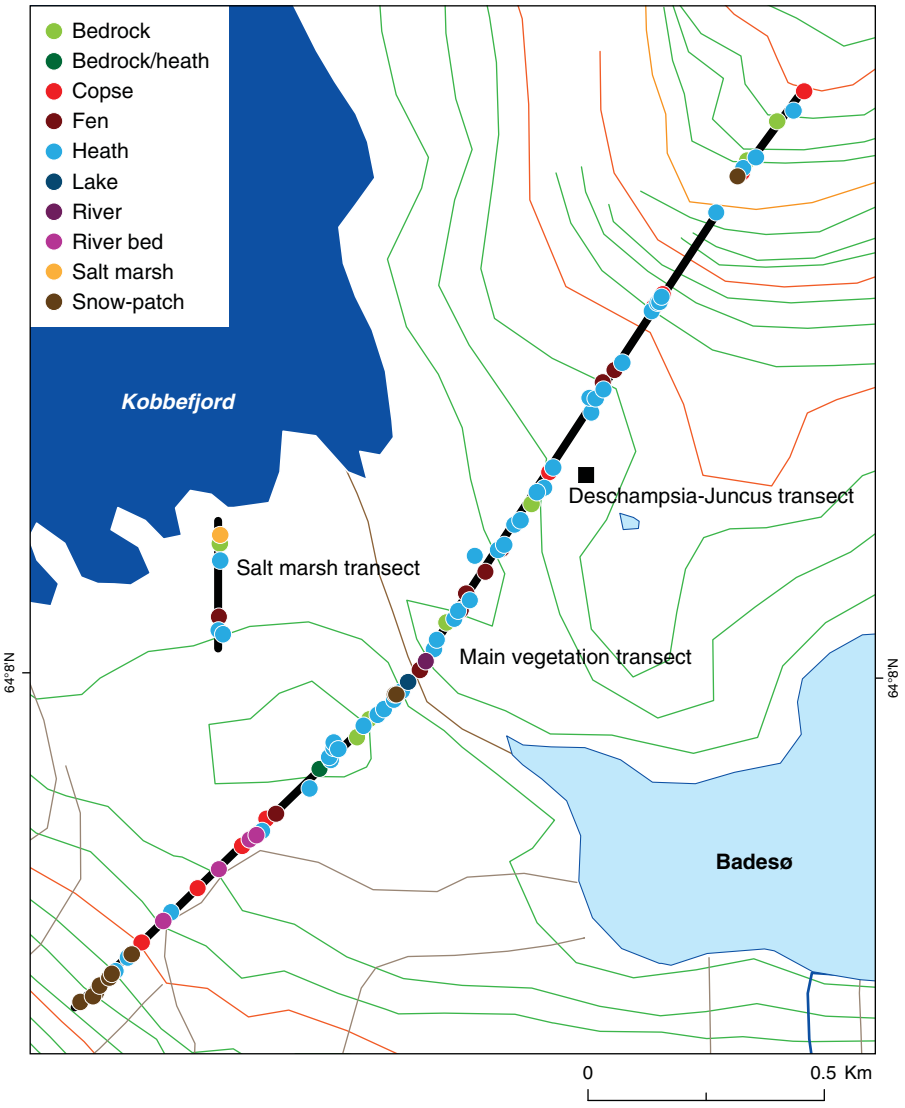


Figure 3. The NERO line. The dots show the positions of the pegs and the colour indicate the vegetation zone northeast of the peg. Numbering of the pegs starts in the south-western corner. The break in the long black line to the north-east marks a steep slope that was not analysed by Raunkjær analyses. The short line represents the coastal zone. The black square shows the position of the characteristic, but rare plant community dominated by *Deschampsia flexuosa* and *Juncus trifidus,* which is found on south facing, dry slopes. The map is based on GPS-positions (accuracy 5-10 m).

Input of data into database

Data from the Raunkiær circle analyses are entered into an Access data base with the columns Peg no., Plot no., Year, Month, Day, Observer, Vegetation type, species name, Raunkiær value, Uncertain species identifications have cfr. (=confer) added to indicate the need for further confirmation. Fertility is given by Flowering added next to the Raunkiær value.

Digital pictures are kept at the Greenland Institute of Natural Resources back up server.

2.1.2 Reproductive phenology

The monitoring consists of weekly counts of buds, flowers, and senescent flowers to monitor the proportion of buds, flowers and senescent flowers of the species: *Salix glauca*, *Loiseleuria procumbens*, and *Silene acaulis*.

Species to be monitored

Three species: Northern willow *Salix glauca* (Blågrå pil), Trailing azalea *Loiseleuria procumbens* (Kryblyng), and Moss campion *Silene acaulis* (Tuelimurt):

- Are commonly found in the area.
- Cover a spectrum of different growth forms (deciduous dwarf shrub, evergreen dwarf shrub, and cushion forming herb/dwarf shrub.
- Are comparable to species monitored by BioBasis in Zackenberg.

There are four plots for each species. The size of each plot varies (see table 2) depending on the abundance of individual flowering shoots of the species in question.

Frequency of sampling

Censuses of *Salix glauca*, *Loiseleuria procumbens*, and *Silene acaulis* are made at weekly intervals from May 1 to September 30 depending on the snow cover. Total counts of flowers are done once a year at peak flowering.

Equipment

- Map with position of study plots (+GPS)
- Data forms (Appendix 2A-C)/ Notebook.

Location and marking of study plots

The positions of the 12 study plots are shown in Figure 4. The plots are marked with angle iron pegs in each corner. The plots are divided into four sections (quarters A, B, C, and D separated by pegs at the centre where the diagonals cross and at the midpoint of each side – see Figure 5). The lettering starts at the corner with the plot-ID and continues clockwise around the centre. Co-ordinates, dimensions etc. appear from table 2.

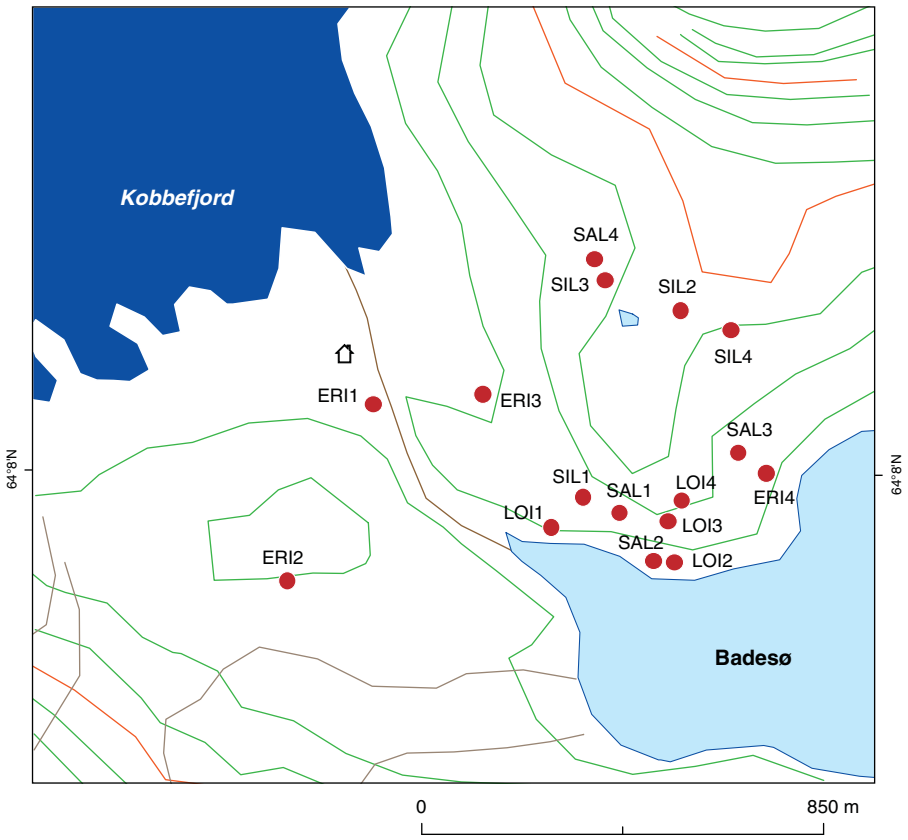


Figure 4. The locations of the plant reproductive phenology plots and plots for annual total counts of flowering shoots for *Salix glauca* (SAL1-SAL4), *Silene acaulis* (SIL1-SIL4), and *Loiseleuria procumbens* (LOI1-LOI4). Note that for *Eriophorum angustifolium* (ERI1-ERI4) only total counts of shoots are carried out. Coordinates can be found in table 2.

Table 2. Positions of plant reproductive phenology plots.

Species	Plot	Latitude	Longitude	Plot dimensions (m)
Eriophorum angustifolium	ERI1	64,1346	-51,3837	4*10
	ERI2	64,1312	-51,3873	10*10
	ERI3	64,1348	-51,3789	12*17
	ERI4	64,1333	-51,3666	5*9
Salix glauca	SAL1	64,1325	-51,3729	7*11
	SAL2	64,1316	-51,3714	8*8
	SAL3	64,1337	-51,3678	6*9
	SAL4	64,1374	-51,3741	4*5
Silene acaulis	SIL4	64,1361	-51,3681	5*12
	SIL1	64,1328	-51,3745	5,5*7
	SIL2	64,1364	-51,3703	11*11
	SIL3	6,4137	-51,3736	7*11
Loiseleuria procumbens	LOI1	64,1323	-51,3759	1,8*3,85
	LOI2	64,1316	-51,3705	1,7*2,95
	LOI3	64,1324	-51,3708	1,6*2,6
	LOI4	64,1328	-51,3702	1,6*3,0

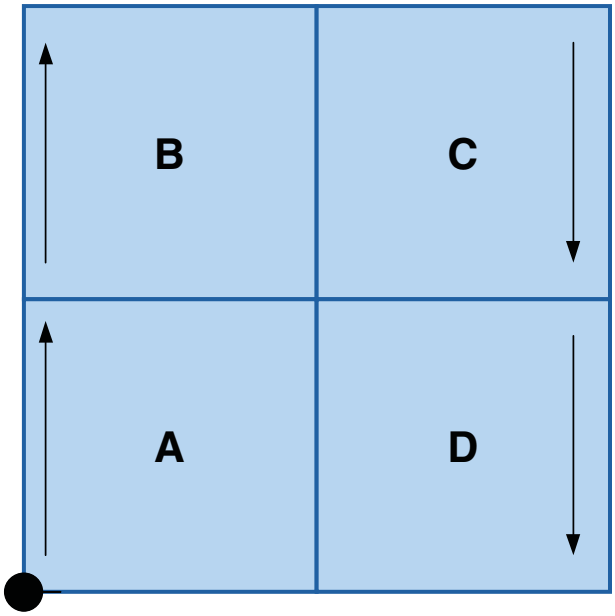


Figure 5. Lettering of subplots in plant phenology plots. The dot indicates the corner with the plot ID. Arrows indicate clock-wise round direction for NDVI-measurements.

Sampling method

The following observations and censuses are entered into the relevant data forms for all plots:

- Time
- Cloud cover
- Plot number
- Snow cover
- Number of buds
- Number of flowers/catkins – note that for *Salix* both male and female plants can be found in the plots
- Number of senescent female flowers/catkins with hairs (*Salix*)
- Number of senescent flowers (*Loiseleuria* and *Silene*)
- Total number of flowers (*Salix, Eriophorum*, *Silene* and *Loiseleuria*)
- Occurrence of larvae, fungi etc.

Data forms are found in Appendix 2A-C. The data from the weekly counts of the plots are entered into data files with columns relevant for each species. The basic data are: Year, Month, Day, Observer, Plot (e.g. Sal1), Section (sector A, B, C, and D), Snow (percent in sector), Buds (actual numbers counted, not percent), Flowers, Senescent (flowers), Total (sum of buds, flowers and senescent flowers), and Remarks. Specific columns for individual species appear from the database files.

During snow melt in May/June, percent snow cover in each plot section is estimated at each sampling trip. If any plant part is visible above the snow layer, the cover is given as 99%. If any ground/vegetation cover is free, no more than 98% can be stated.

When visiting *Silene*-plots, samples of a total of at least 50 flower buds, flowers or senescent flowers (or capsules with exposed seeds) are recorded within each plot section. In the *Salix* and *Loiseleuria* plots a total of 100 buds, flowers and senescent flowers are recorded. This is done by

counting the different phenological stages within appropriate group sizes of individuals concomitantly until a total of 50/100 is achieved. Begin to the right in each section and count towards the left. Avoid biasing the count by actively selecting a starting point other than the right corner.

In general, flower buds are defined as flowers not yet open, flowers are open giving insects access to the reproductive organs, and senescent flowers as flowers that have lost all petals or with all petals almost or fully faded or brown. In some of the final stages, flower stems from the preceding year may interfere with the counts. However, such old stems are always dry and stiff; stems of this year are soft and fleshy.

For each species, the following sampling procedures apply in particular:

Salix:
The sampling unit is catkins, not individual flowers. Most flowers from one catkin emerge the same day, and they also wilt at the same time. Hence, catkins are recorded as buds (Figure 6), when no stigmas or anthers are visible, and as male (Figure 7) and female (Figure 8) flowers as soon as stigmas (f) or anthers (m) are visible (they are often both red in the early stages, but the colour may vary).

Both senescent flowers and fruits are continued to be recorded as 'flowers' until they are recorded as having exposed seed hairs (Figure 9) from the time of exposure of the first hairs on top of the splitting capsules. Notice that fruits may be affected by larvae so that they expose seed hairs from the bottom of the capsules (excreta from the larvae are often visible among the seed hairs). These capsules must not be recorded as having seed hairs exposed, but should be recorded separately. In Kobbefjord there is no experience as to this issue yet.

Figure 6. *Salix glauca* buds. It is not possible to discriminate between male and female flowers at this stage.

Figure 7. *Salix glauca* male flowers.

Figure 8. *Salix glauca* female flowers.

Figure 9. Senescent *Salix glauca* female flowers with hairs.

Fruits infected by sponges (yellow and twisted) should be recorded separately (yet still included in the number for 'flowers', i.e. the infected fruits appear twice in the data forms). Also, infections by insects should be recorded.

Silene:

Silene acaulis grows in hummocks (Figure 10) and one or a few specimens may dominate the sample. Therefore, several individuals must be sampled each week.

Flower buds are reddish or light purple (Figure 11). Senescent flowers (Figure 12) have wilted petals or appear as empty “cups” (Figure 12). Senescent flowers are defined as flowers with faded petals and empty pollen anthers.

Figure 10. *Silene acaulis* hummock.

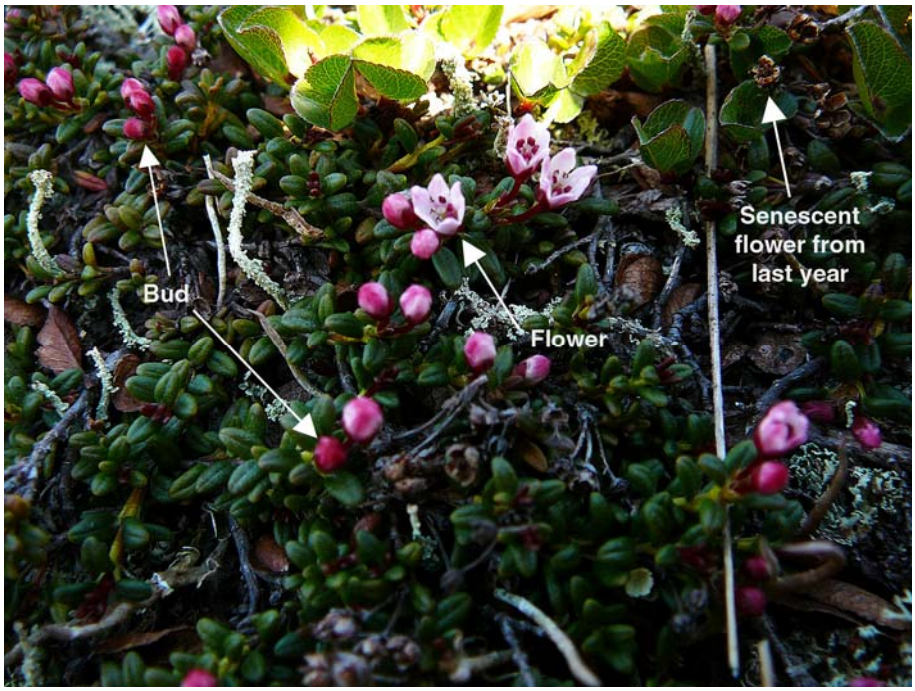
Figure 11. *Silene acaulis* – flowers in the foreground and buds scattered in the hummock. The buds in the background should be recorded as buds even though they are close to opening as flowers.

Figure 12. *Silene acaulis* – senescent flowers – in the middle still with wilted petals.

Loiseleuria:

Loiseleuria procumbens is a matted shrub with pairs of tiny, oblong, closely-set leaves and abundant clusters of small flowers, see Figure 13. In Greenland plants are not taller than 10 cm. The plant is creeping, much-branched, mat-forming, with 2-5 pink, bell-shaped flowers in terminal clusters and evergreen leaves with rolled edges.

Figure 13. *Loiseleuria procumbens.* Half open flowers, opening buds, closed buds and senescent flowers from last year.



Input of data into database

The data from the weekly registrations are entered into Excel data sheets with columns relevant for each of the three species. The basic data are: Year, Month, Day, Observer, Plot, Sample, Snow cover, Buds, Flowers,

Senescent flowers, Total, and Remarks. Specific columns for individual species appear from the data base.

2.1.3 Total flowering

Species to be monitored

Northern willow *Salix glauca* (Blågrå pil), Trailing azalea *Loiseleuria procumbens* (Kryblyng), Moss campion *Silene acaulis* (Tue-limurt), and Cotton grass *Eriopherum angustifolium* (Smalbladet kæruld). See previous section for descriptions of *Salix glauca, Loiseleuria procumbens*, and *Silene acaulis*. *E. angustifolium* is described below.

Eriophorum angustifolium

The flowers are monoecious (individual flowers are either male or female, but both sexes can be found on the same plant) and are pollinated by wind. There are two or more flowers on each stem. There are two or more fruiting heads per plant, which distinguishes it from the other common species, Arctic cotton grass. Figure 14-16 shows different stages of *Eriophorum* flower development.

Figure 14. *Eriophorum angustifolium.* Buds and young flowers.

Figure 15. *Eriophorum angustifolium.*

Frequency of sampling

Once per season. Total counts of *S. glauca*, *L. procumbens*, and *S. acaulis* are made at peak flowering. The optimal time for total counts of *E. angustifolium* is when most or all flower buds have reached senescence.

Equipment

- Map with position of study plots
- Pieces of cord totalling 100 m
- Flower sticks
- Tally counters
- Data forms Appendix 2A-D/ Notebook

Figure 16. *Eriophorum angustifolium.* Senescent flowers.

Location and marking of sampling plots

The plots are divided into four sections (quarters A, B, C, and D separated by steel pegs at the centre where the diagonals cross and at the midpoint of each side). The lettering starts at the corner with the plot-ID and continues clockwise around the centre. Co-ordinates, dimensions etc. appear from table 4.1. The plots are identical with the plant reproductive phenology plots shown in figure 4.

Sampling method

Tighten a cord around each section of the plot. In large plots, subsections are established by placing two additional cords with about 0.5 or 1 m intervals from one end of each section, whereupon the lumped number of flower buds, flowers and senescent flowers are counted between each cord. Move one cord at a time and repeat the process until the entire plot is covered. In small plots, sticks may be used instead of cords. In the *Salix* plots, male and female catkins are counted separately. Catkins that have been grazed, but can still be sexed, are included.

Input of data into database

The data from the yearly registrations are entered into Excel data sheets with columns relevant for each of the three species. The basic data are: Year, Month, Day, Observer, Plot, Sample, Snow cover, Total, and Remarks. Specific columns for individual species appear from the data base.

2.1.4 Normalised Difference Vegetation Index (NDVI) in plots and along the NERO line

The progression in the vegetation greenness is followed along the vegetation transect and in the plant phenology plots. The monitor measures the spectral reflectance of the plant canopy.

Species or taxonomic groups to be monitored

All vegetation types along the NERO line between VT001 and VT076.

All plants in the reproductive plant phenology plots.

Frequency of sampling

Along the NERO line: Monthly.

Plant phenology plots: Weekly in connection with the plant phenology censuses.

Equipment

- Map of vegetation transect and plant phenology plot positions
- GPS with positions of vegetation transect and phenology plot positions
- Crop Circle Handheld system. A handheld Crop Circle TM ACS-21 0 Plant Canopy Reflectance Sensor which calculates the greening index (NDVI). http://www.hollandscientific.com/CC_Handheld.html
- Notebook
- Digital camera.

Location and marking of sampling plots

The NERO-line crosses all the dominating vegetation types found in the study area. The NERO line is described in detail in Bay et al. 2008. The measurement is carried out 5 m north-east of the vegetation transect. Surveying rods mark the transect to be scanned. Figure 17 gives an impression of the vegetation greenness in July 2008.

Plant phenology plots : See Table 3 and Figure 4.

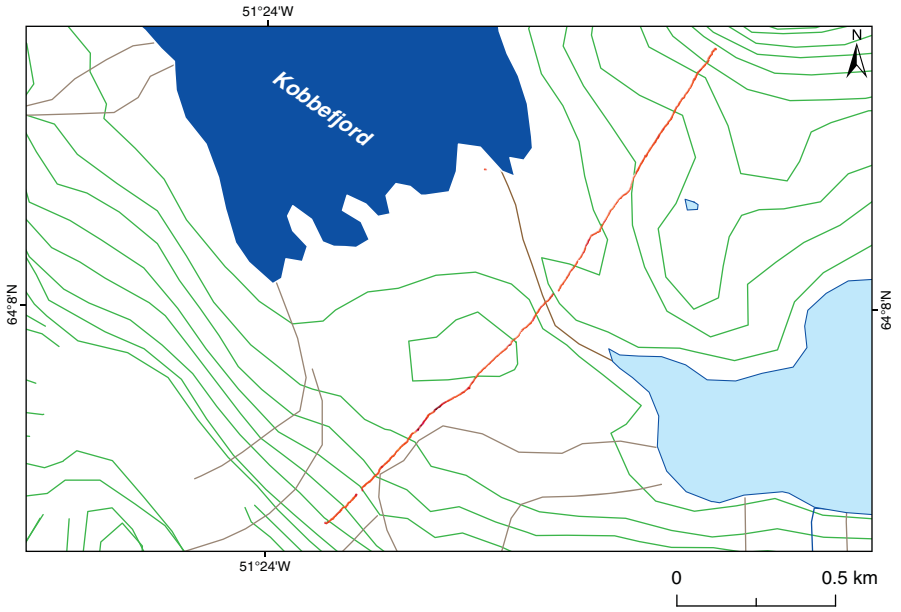


Figure 17. The NERO-line–vegetation greenness. Results of NDVI-scans July 8, 2008. The darker coloured the greener the vegetation.

Table 3. Sequence of phenology plots and NDVI.

ERI1	(Eriophorum 1)
EMP1	(Empetrum 1)
ERI2	(Eriophorum 2)
EMP2	(Empetrum 2)
ERI3	(Eriophorum 3)
EMP3	(Empetrum 3)
LOI1	(Loiseleuria 1)
SIL1	(Silene 1)
SAL1	(Salix 1)
SAL2	(Salix 2)
LOI2	(Loiseleuria 2)
LOI3	(Loiseleuria 3)
LOI4	(Loiseleuria 4)
ERI4	(Eriophorum 4)
EMP4	(Empetrum 4)
SAL3	(Salix 3)
SIL4	(Silene 4)
SIL2	(Silene 2)
SIL3	(Silene 3)
SAL4	(Salix 4)

Sampling method

NDVI is measured by **The Crop Circle Handheld System** which integrates a Crop Circle ACS-210, GeoSCOUT GLS-400 and a FieldPAK PS-12 into a single instrument (See Figure 18). Data is collected and stored on a SD flash disk.

Figure 18. Equipment for measuring NDVI. The Crop Circle Handheld System. Source: http://www.hollandscientific.com/CC_Handheld.html.

Please proceed as follows:

1. Insert an empty SD flash card into the card slot
2. Turn on the CropCircle system by pressing the ON/OFF button
3. Press the DISP button to select MAP mode. Then press OK
4. When ready press LOG and the CropCircle starts to measure NDVI
5. Use the trigger switch also connected to the CropCircle between each subplot (A, B, C, and D)
6. Turn the CropCircle OFF after each plot in order for the data to be saved on the SD flash card

Scans are conducted by moving the sensor steadily forward (ca. 1 meter per second) approximately 75 cm above the vegetation. This results in a measuring footprint of approximately 10 x 45 cm. Refer to the CropCircle manual for more information.

The sampling order shown in Table 3 must be applied. Also, always measure all plots in the order A-D (see Figure 5). At each visit, note under Remarks the presence of snow (snow in subplot; snow at plot edge) and if the vegetation is wet.

All measurements are conducted only on the AB and the CD sides of the plots (Figure 5). Place yourself at the plot number plate, just outside the plot. Hold the sensor app. 50 cm into the subsection at the subsection edge. Switch on the NDVI logger (switch on the left of the stage), and walk slowly (approximately 1m per second) along the sides indicated by arrows on figure 5. Use the trigger switch to pause the NDVI logger at

the next corner of the subsection. Repeat the procedure in the remaining subsections. Hence, four scans are made in each of the vegetation plots. Turn off the Crop Circle system between plots by pressing the ON/OFF button.

When measuring the NERO line always start at the top of the slope and walk towards the river.

Ideally all transects should be measured on the same day. If the vegetation is wet, the measurements must be postponed to the following day.

Input of data into database

Data are downloaded from the SD card from the CropCircle using a card reader. Crop Circle automatically names the files (e.g. ddmmyyAA.CSV; ddmmyyAB.CSV; etc.) Each file holds the following variables: Longitude, Latitude, Elevation, Fix Type, UTM Time, Speed, Course, SF1, SF2, SF3, SF4, SF5, and SF6. All Crop Circle data files are saved separately. In Excel, each data file is supplemented with the following columns: Year, Month, Day, DOY, Observer, Plot, Section, and Remarks. All files are merged into one sheet in one file. Please notice that if your computer is set with a Danish Office-version the ddmmyyAA-file is the last file in the file list since the AA is regarded as Å (but it is still the first one recorded).

Digital pictures are stored at the Greenland Institute of Natural Resources back up server (F:\40-59 PaFu\41 Vegetation\08 Nuuk-Basic_BioBasis).

2.1.5 CO_2 flux plots

The ratio between the uptake of CO_2 from photosynthesis, and release from decomposition of organic matter in the soil, and respiration is measured. The ratio is called Net Ecosystem Exchange (NEE).

Species or taxonomic groups to be monitored

The vegetation in the ITEX plots which is dominated by *Empetrum* heath with *Salix* as subdominant species. The reproductive phenology of Salix is followed in all plots. Soil moisture is measured in all plots. Temperature is recorded by TinyTags.

Frequency of sampling

Carbon fluxes are measured weekly. All plots should be measured between 10 AM and 3 PM, and on the same day.

Equipment

- ITEX-chambers incl. bolts and guy ropes
- TinyTag temperature data loggers and sensors
- EGM4 – see Figure 21
- Plexiglass measuring chamber (PMC) – measuring 33x33x34 cm (LxWxH).
- Theta-probe for soil moisture measurements
- Black plastic bag adjusted for the PMC
- Sticky Tack
- External 12V battery
- Ruler

- Digital watch
- Digital camera
- Data forms (Appendix 3A-C)/ Notebook.

Location and marking of sampling plots

30 plots are situated in a mesic dwarf shrub heath dominated by *Empetrum nigrum* and with *Salix glauca* as subdominant species. The heath is facing west.

Figure 19 gives an overview of the site and Figure 20 shows the relative positions of the plots.

Figure 19. Overview of CO_2 flux plot site with Hessian tents for shading and ITEX hexagons for increasing temperature.

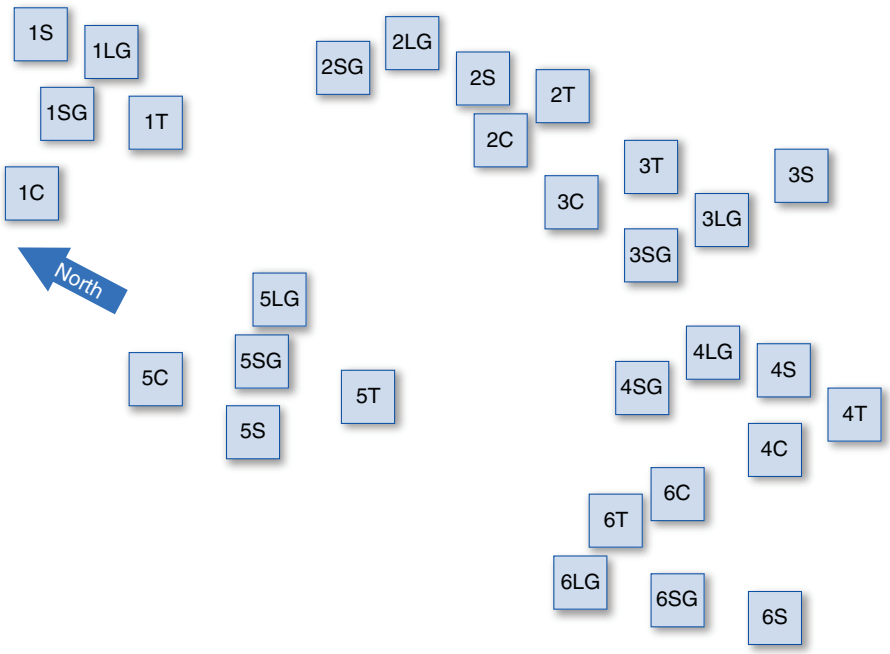


Figure 20. Detailed map showing the position of the CO_2 chamber plots. The position of the midpoint is 64,137˚N/51,38˚W. For explanation of the abbreviations please see text.

The setup consists of 5 treatments each replicated 6 times: Control (C), increased temperature (T, ITEX hexagons), shading (S, hessian tents), long growing season (LG, removal of snow during spring) and short growing season (SG, addition of snow during spring).

Temperature is enhanced at T plots by placing hexagonal open top ITEX chambers (OTCs), see Figure 22. This way temperature is expected to increase 1-2 ºC during the growing season (for further information, see Molau & Mølgaard, 1996). The shading treatment (S) imply erecting dome shaped sack cloth tents over the soil and vegetation causing an expected 60% reduction of incoming light (See Havström et al., 1993).

Short and long growing season will be implemented by respectively adding to and removing snow from SG and LG plots during snow melt at the spring causing plants and soils to be exposed earlier (LG) or later (SG) than in control plots.

In each of the 30 plots a metal frame of 35x35 cm has been inserted permanently into the soil. The frame is used for weekly measurements of ingoing and outgoing fluxes of CO_2 to the system by the closed chamber technique. The metal frames were placed at spots were *E. nigrum* and *S. glauca* dominated the vegetation. The metal frame is not to be removed by the end of the season.

Sampling method

The CO_2 flux plots are established as soon as possible early in the season (make note of the date). By the beginning of the season, check that the metal frames are level and adjust if needed. Do not adjust the metal frames later or in connection with the gas flux measurements!

The six plexiglas sides of each ITEX hexagon is bolted together in the field, and additionally secured with six guy ropes. Place a TinyTag temperature probe app. 2cm horizontally into the soil. Also place the TinyTag logger inside the plot. TinyTags are programmed to log the temperature every 30 minutes.

Before each measuring round, the EGM must be calibrated to the CO_2 level in the air (the level varies between app. 365 and 380 ppm). Also, in the lab replace the old Soda lime in the EGM with fresh Soda lime. See the EGM manual for further details (PP Systems 2003).

Before beginning the gas flux measurements in a plot take a digital orthophoto covering the entire area inside the metal frame, take three soil moisture measurements outside the metal frame but inside the plot using the ThetaProbe. See the ThetaProbe manual for further details, and measure the height (cm) of the upper edge above the ground of the metal frame on the four sides of the frame. Measurements of the chamber height is only done three times during the season (beginning, mid and end of season) in order to avoid unnecessary tear on the vegetation.

Measurement of carbon flux

1. While at the laboratory make sure the EGM4 is set to automatically take a measurement every minute (Figure 21). This is done by turning on the EGM4, press 2SET and then the 5RECD button. To change the recording from manual (M) into automatic (A) press 1REC. When

the recording type is changed into automatic the time interval (2INT) changes from 0 to 1 (1 minute) by default. The time interval can range from 1 to 720 minutes.

2. In the field (Figure 22) place the HTR-2 probe into the plexiglas measuring chamber (PMC), connect the probe and the tubes from the probe to the EGM4 (in = black tube) and back from the EGM4 to the PMC (out = clear tube). Seal the entrance to the chamber with sticky tack.
3. Turn on the EGM4 and press 1REC for record by pushing button 1. The EGM4 will have to heat up to approximately 50°C (it takes approximately 5 minutes depending on the surrounding temperature). Following the "heating-up" it automatically runs a calibration with the CO_2 in the air.
4. When the EGM4 runs the calibration (called "Counting Zero") place the PMC in the metal frame in the first plot to be measured. Make sure the PMC handle does not cast a shadow on the HTR-2 probe with the PAR measuring device. While measuring gas flux the EGM4 automatically measures the PAR (light intensity) and the temperature in the PMC.
5. When the first record is taken turn on the digital watch.
6. The EGM4 automatically takes a record every minute. Check the digital watch and when 4 minutes have passed (equalling 5 measurements: t = 0, 1, 2, 3, 4) lift the PMC off the frame. Aerate the PMC making sure the CO_2 level returns to that prior to measuring. Monitor the CO_2 concentration on the EGM4. Approximately 15 seconds before the next measure (at 5 minutes) place the PMC on the frame again and cover it completely with the black plastic bag. After yet another 4 minutes lift the PMC and aerate it while walking to the next plot to be measured.
7. Since the EGM4 automatically takes a record every minute you have 1 minute (or approximately 45 seconds) to aerate the PMC between light and dark measurements or to walk between plots.
8. The external 12V battery is connected to the EGM4 after approximately 2½ hours depending on the surrounding temperature.
9. When all plots have been measured turn off the EGM4.

Measurement of soil moisture

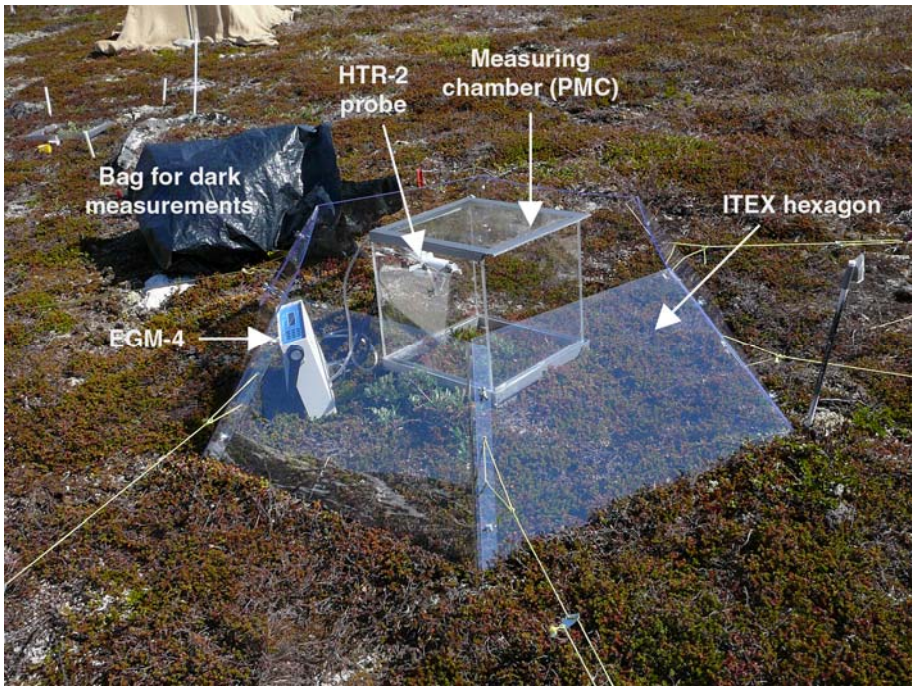
Take three measurements in each plot by sticking the sensor into the soil, read out the soil moisture, and enter data into the form Appendix 3A-B.

Reproductive phenology of Salix

Reproductive phenology of *Salix glauca* is followed in all plots according to the procedure described in section 3.1. Data are entered into the form Appendix 3C.

Figure 21. Upper figure shows the EGM4 which is used for measuring CO2 concentrations. The lower Figure shows the top of the instrument.

Figure 22. Measurement of CO_2-flux in an ITEX-hexagon open-top chamber with a measuring chamber (PMC) fitted with a HTR-2 probe connected to the EGM4. The black plastic bag is used for dark (respiration) measurements.



Laboratory work

None.

Input of data into the database

Data are downloaded from the EGM4 using the EGM transfer software, and the raw files are saved in a separate folder renamed to include the

date of the measurements (yymmdd.dat). In Excel, data are supplemented with the following columns: Year, Month, Day, DOY, Hour, Min, Plot, Treatment, Light, Photo_no, Recno, Cloud cover, Observer, Soil moisture, Chamber height, and Remarks. All files are merged into one for one season.

Download digital pictures and rename them to include plot name and date (e.g. ITEX_1C_090602). Save in a separate folder named Gas Flux.

Data from TinyTags brought back to the station by the end of the season are downloaded. Rename the individual files to include plot name and year (e.g. ITEX_5C_2008), and save in a separate folder named EGM/Temperature.

2.1.6 UV-B exclusion

The impact of ambient UV-B radiation on the vegetation is studied in a mesic dwarf shrub heath by placing filters approximately 10 cm above the vegetation. At the time of peak plant growth the chlorophyll a fluorescence is measured as an indicator of plant health.

Species or taxonomic groups to be monitored

A mesic dwarf shrub heath (facing WSW) dominated by *Empetrum nigrum* and with *Betula nana* and *Vaccinium uliginosum* as subdominant species. *Betula nana* and *Vaccinium uliginosum* are measured.

Frequency of sampling

Measurements of chlorophyll fluorescence of leaves of *Betula nana* and *Vaccinium uliginosum* is carried out three times with one week interval at the peak of plant growth (week 29 to 31).

Equipment

- HandyPea fluorimeter (PEA=Photosynthesis Efficiency Analyzer) – See figure 23 and further description below.
- 80 leaf clips
- Notebook
- Digital camera
- Frames with UV-B filter (Mylar film, 0.25 mm - with exclusion of UV-B)
- Frames of filter control (Teflon film - without exclusion of UV-B).

By the end of the field season, all equipment at UV plots are taken down and brought back to GNIR.

The Handy PEA chlorophyll fluorimeter consists of a control unit. The chlorophyll fluorescence signal received by the sensor head during recording is digitised within the Handy PEA control unit. Up to 1000 recordings of between 0.1 - 300 seconds may be saved in the memory of Handy PEA chlorophyll fluorimeter. Saved data can be viewed onscreen but shall be transferred to a computer for storage and further analysis.

Figure 23. Handy PEA with clips. Source: http://hansatech-instruments.com/handyPea.htm.

The sensor unit consists of an array of 3 ultra-bright red LED's optically filtered to a peak wavelength of 650 nm, which is readily absorbed by the chloroplasts of the leaf. The LED's are focused via lenses onto the leaf surface to provide even illumination over the area of leaf exposed by the leaf clip (4mm diameter).

Location and marking of sampling plots

The UVB plots are situated west of the CO_2 plots. Figure 24 and 25 gives overviews of the plots.

Figure 24. Overview of UV-B plots.

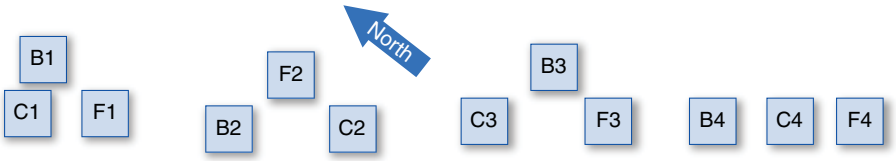


Figure 25. Schematic presentation of the location of UV-B plots.

There are three series of plots with five replicates:

1. Control - no treatment: C1-C5
2. UV-B filter (Mylar film, with exclusion of UV-B) : B1-B5
3. Filter control (Teflon film, without exclusion of UV-B) : F1-F5.

Each treatment plot measures 60 cm x 60 cm; the plots are marked with aluminium tubes at each corner and covered with a frame with the appropriate filter placed approximately 10 cm above the vegetation. During summer the vegetation may grow as tall as the filter which may then be lifted within the aluminium tubes.

Sampling method

Before establishing the UV plots, filters on frames are checked carefully, and changed if necessary. There are two filter types, Teflon (filter control; the thinnest and most flexible film) and Mylar (excludes UVB; thicker and less flexible). Frame positions are given by small sticks within each plot. UV plots are checked regularly during the entire field season, and repaired if necessary. Specifically, filters in the UV plots must be checked after heavy rain or wind.

1. Select five green, "healthy-looking" leaves of *Betula nana* and *Vaccinium uliginosum* in each plot.
2. Mount leaf clips on all leaves – preferably without removing the leaf from the branch. Mount on one species at a time. Make sure that the leaf is visible through the hole in the clip and push the shutter to cover the hole so the leaf material is in complete darkness.
3. Keep the shutters closed for at least 30 minutes. The closure time may be longer.
4. Switch on the Handy PEA, Open "main menu" and turn the arrow on the screen to "Measure".
5. Fit the sensor head to the clip; uncover the hole by pushing the shutter back. Start measuring by pushing "OK" or push the black button on the sensor head. During the measurement a number of parameters appear on the screen. Note that the "Fv/Fb" should be about 0.8. If something goes wrong step three must be repeated before you carry out a new measurement.
6. Accept to store the measurement. Note which measurement number corresponds to each plot.
7. Repeat the sampling now measuring on *Vaccinium uliginosum* leaves.
8. Take a photo of each plot at each measurement round.

Input of data into database

When all measurements have been completed, data must be transferred to a computer by use of the Handy PEA programme. Make sure that all data have been transferred to the computer before clearing the memory in the Handy PEA.

Data are downloaded from the HandyPEA using the PEA Plus software, and the raw files are saved in a separate folder named to include the date of the measurements (e.g. yymmdd.pcs). In Excel, data are supplemented with the following columns: Year, Month, Day, Observer, Species, Treatment, and File no.

2.2 Arthropods

Surface living arthropods are captured in yellow pitfall traps.

Species to be monitored

All taxonomic groups of arthropods.

Frequency of sampling

The traps are emptied weekly on fixed dates. If bad weather prohibits visits to the fjord or proper handling of the samples, the traps may be emptied on the earliest day of convenience.

Equipment

- For field work
- GPS
- 32 yellow (Pantone no. 108U) plastic cups, 10 cm in diameter and 8 cm deep. Cups have been placed permanently for the season. At the beginning of the season it is checked if all cups are placed properly.
- 8 window traps
- A thermos
- A garden trowel with sharp edge
- 1 x 2 L container for water
- Detergent: Odour free detergent (Coop Änglemark Bluecare Dish wash, concentrated, without perfume, colour and preservation agent).
- Salt (NaCl) without iodine and anti-caking agent
- 20 Metal pegs (to be used in the fen area)
- 1 lady's stocking per emptying bout
- A pair of flat tweezers
- 32 plastic containers with lids
- 1,5L of 70% ethanol
- Small bottle with tip (for rinsing the stocking with alcohol)
- Waterproof speed marker
- Disposable syringes for removal of surplus water
- Ethanol resistant labels
- Pencil
- Ethanol resistant speed marker
- Data form Appendix 4/note book.

Location and marking of sampling plots

The position of the study plots are shown on Figure 26. Each plot measures 10 x 20 m and is made up of eight 5 x 5 m squares marked with metal pegs in each corner. Each plot is identified with a number plate, and sections (with one trap each) are lettered A-H clockwise from the number plate, see figure 27.

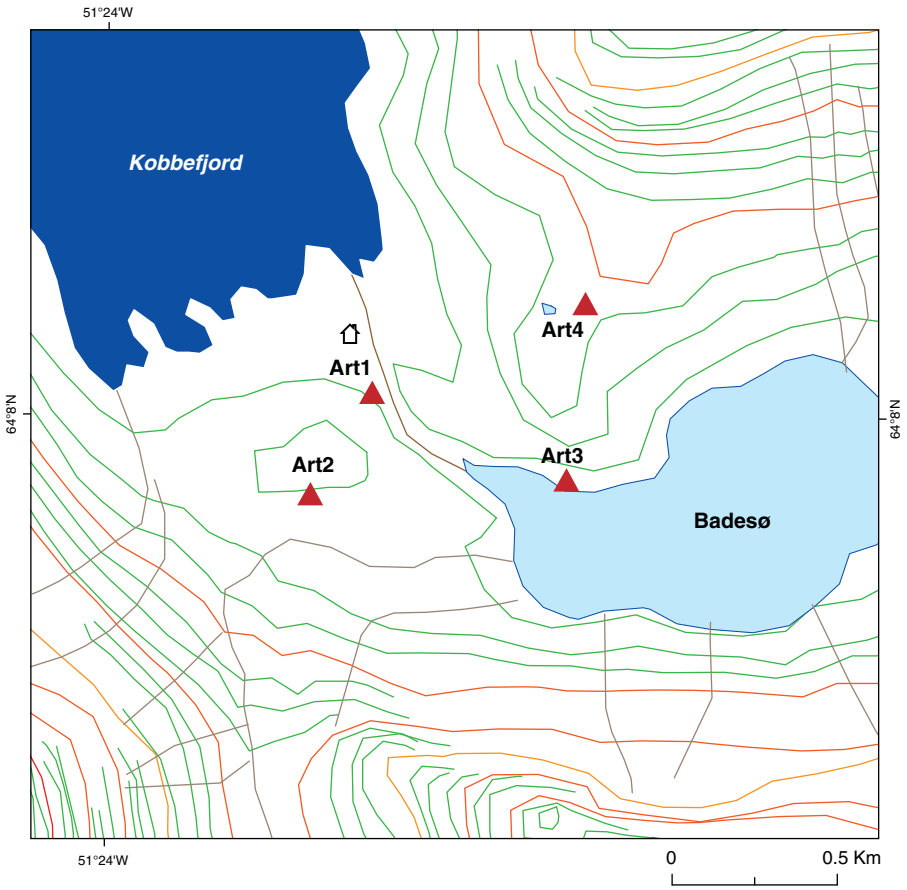


Figure 26. Location of pitfall traps in relation to vegetation type. Art1: *Empetrum nigrum* heath, Art2: Fen, Art3: *Betula nana*/*Salix glauca* heath, Art4: Abrasion.

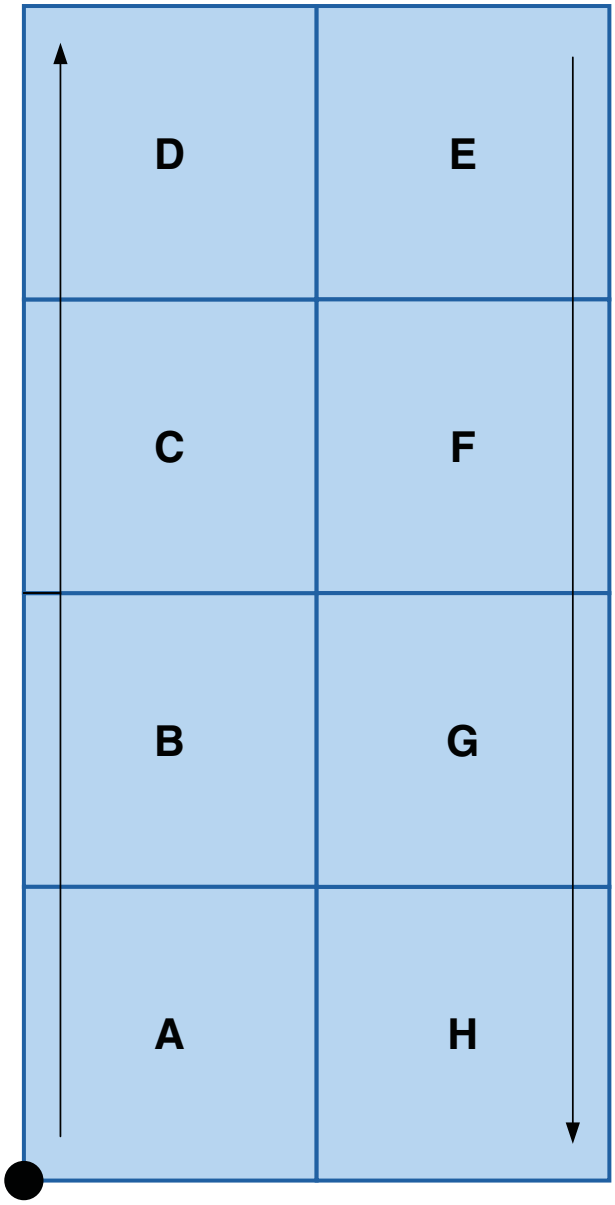
Sampling method

A set of eight pitfall traps are established in each plot. Each trap is composed of two plastic cups fitting into each other, so that the upper one can be lifted and emptied without disturbing the surrounding soil. The traps are positioned randomly within each of the 5 x 5 m2 squares by using the table with random numbers; see the Microarthropod section. The trap is then buried on the nearest reasonably level and 'elevated' site (so that it is not flooded during the snow melt or heavy rain) and carefully sunk into the soil, so that the upper rim levels exactly with the soil surface. Place the turf and the removed soil about a meter away from the trap. Do not disperse it, since it must be repositioned after the season, when the traps are removed.

The new traps are placed upside-down during the winter. At the start of the season (i.e. on the round when the traps have appeared from the snow), new clean (washed with a little Tween 20) upper cups replace the 'wintering' ones. Bring hot water in a thermos in case the two cups are frozen solid.

If there is any risk that cups will float up due to water in the lower cup, two metal pegs must be placed along each cup to keep them in position.

Figure 27. Schematic diagram showing positions of Arthropod plots.



The upper cup of the trap is then filled 2/3-3/4 with water (1 l needed per station) added three drops of detergent and a spoonful of salt as killing agent, preservation and to prevent freezing.

Emptying the traps

Catches from each of the traps are kept separate. Before emptying a trap place the ladies stocking on a spare cup. Then pour the trap liquid through the stocking into the spare cup. Check the trap cup for remaining arthropods and flush with ethanol down into a 10 ml container should any still remain in the trap. Reposition the trap cup in the soil. The catch from the ladies stocking is now emptied into the 10 ml container by turning the stocking upside down on top of the container. Rinse the inverted stocking with ethanol from the tip of the small bottle. All remaining invertebrates must be removed carefully from the stocking using tweezers and put into the container. Plot number and Section (A-H) are written with an alcohol proof pen on the containers and Date, Plot number and Section (A-H) are written on a small water-proof piece of paper which is placed in the container.

After emptying all traps, extra water may be added to the traps to compensate for evaporation since last round (up to ½ l needed per station). In the middle of each season, a little salt and detergent must be added to compensate for loss during the season.

Bring an extra pair of cups on each round, together with equipment for setting up traps, in case a trap has been destroyed, e.g. by a fox. Any failures such as flooded or floating cups, fox faeces etc. must be recorded. This includes occurrence of fungi in the water. In that case a new cup with fresh water (+salt and detergent) must be established.

Note the full hour of the day, when the traps in each plot are emptied.

At all visits at the arthropod stations during snow melt, the snow cover (%) is estimated for each section of the plot.

Never touch the traps with mosquito repellent or suntan oil on your fingers!

Ending the season

At the termination of the catching season the trap liquid must be collected from all the traps and poured into the river. All the 'old' traps are gathered, and the turfs put back into the hollows. New traps are established at all stations. Arthropod samples are kept at GINR.

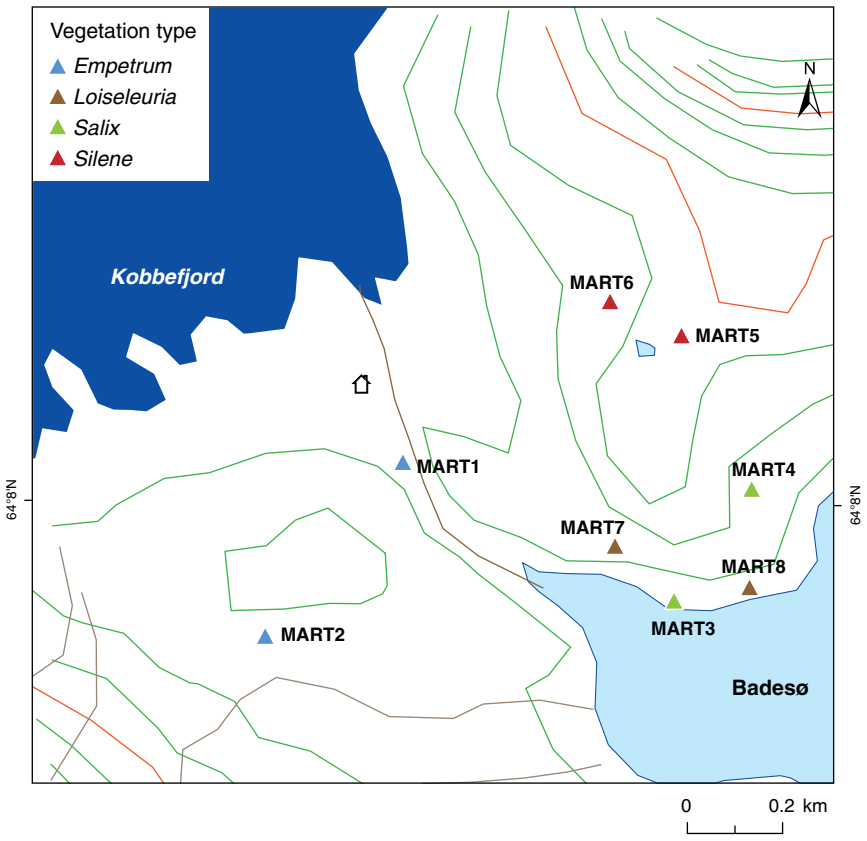


Figure 28. Location of plots for microarthropod sampling and litterbags.

Laboratory work

None.

Input of data into database

After the weekly emptying of the pitfall traps, the following data are entered into a Excel data sheet named Art1-4: Year, Month, Day, DOY, Hour, Plot, Fieldworker, Sorting, Snow A (% in the section), Snow B, Snow C, Snow D, Snow E, Snow F, Snow G, Snow H, Days A (trap days since the last emptying of the trap in the section), Days B, Days C, Days D, Days E, Days F, Days G, Days H, Taxon, and Remarks. Under Remarks, data of opening and closing together with relevant observations about the traps are stated. This include any disturbance that may influence the efficiency of the traps such as flooding, drying out, icing, dirt, faeces, and vandalism by mammals or humans.

After sorting, the total number of individuals per group is entered into the Excel data sheets according to Taxon and trap section.

2.3 Microarthropods and decomposition

2.3.1 Microarthropods

Species to be monitored

All microarthropods: Collembola at species level and mites at order level.

Frequency of sampling

Sampling is performed three times during the season corresponding to: spring (after snow melt), summer and autumn (before the snow appears). Extraction will be very slow in wet samples. To avoid this, sampling should be postponed until soil moisture is lower.

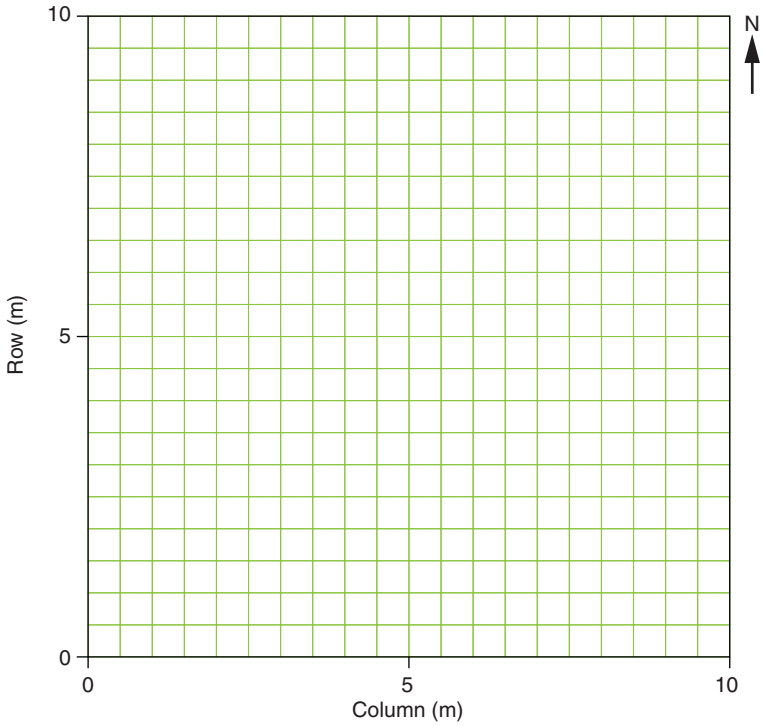


Figure 29. Microarthropods and litterbag sampling grid in 10x10 m plots with grid size: 0.5 x 0.5 m.

Equipment to be used

- Map/GPS with positions of plots
- Soil auger

- 64 microcosms tubes made of Plexiglas (height 5.5 cm/diameter 6 cm)
- 128 pieces DBIdut lids (size 89B)
- Tape measure
- Shears
- Knife to cut roots etc.
- Pre-printed labels, incl. Date, Plot Id, plant community (*Silene, Salix, Empetrum, Loiseleuria*), Replicate Id., Initials
- Transportation boxes.

Location and marking of study plots

The sampling programme consists of collecting microarthropod samples from:

4 habitats * 2 plots * 8 subsamples * 3 sampling occasion = 192 samples.

The sampling occasions may coincide with the three litterbag collections, if feasible. To ensure enough undisturbed sampling points for several years each plot is divided into a ½ meter square grid (Fig. 2).

The coordinates (x,y)=(0 m, 0 m) is the exact position of the iron corner stick with written label. An Excel table with random sampling points include these coordinates, for each subsample. The sampling points are sorted according to the x-coordinate. The random sampling Excel table with x (column) and y (row) coordinates include 10 subsamples to be used for the litterbags and of those 8 are used for the microarthropod soil cores. For practical reasons the same set of random numbers are used for all 8 plots at each sampling occasion.

Sampling method

1. The soil auger including two microcosm tubes is closed and ready for use.
2. The point of sampling is found using the random sampling table and a measuring tape.
3. The soil auger is placed vertically at the sampling point so it touches the soil surface.
4. At sites with dense vegetation it may be necessary to use a knife to cut around the soil auger before pushing it down into the soil or peat. Take care not to damage the soil/peat core.
5. Push the soil auger vertically 5.5 cm downwards so that the lowest tube is just filled with soil. The soil surface shall level the upper rim of the lowest tube. The soil auger is open in the top so that you can follow how the soil appears in the tube. The upper tube functions only to fix the lower tube. While pushing the soil auger down turn it from side to side thereby avoiding compressing the soil in the tube.
6. Tilt the soil auger from side to side loosen the soil core at the bottom and take care when you pull the soil auger including the soil core up.
7. Open the soil auger and carefully remove the tube including the soil core. Place a labelled DBIdut lid at the top immediately to avoid that organisms on the soil escape.
8. Turn the tube around and cut surplus soil away so the soil surface levels the bottom of the lower tube. Place a DBIdut lid in the bottom of the tube.
9. Place the tubes in a box with the top of the sample upwards.

Store the samples at low temperature in a shadowed place, and avoid bumping during transportation. On arrival to the lab the samples are stored in the dark at 5 ºC until extraction not later than two days after sampling.

Laboratory work

Extraction of microarthropods

The capacity for extraction is limited so it may be necessary to run the extraction more than once. To account for differences due to longer storage etc. between two extraction batches the principle of "blocking" is followed. Thus, a fraction of sub-samples, with a unique name e.g. *extraction block no. 1*, with e.g. half of the samples from a sampling plot, are randomly selected for the first extraction and the remaining other half, *block no. 2*, is stored at 5 ºC until extractors are ready. The blocking enables a statistically valid assessment of the possible differences between the blocks, i.e. the two extraction sessions.

Equipment

- Extractor with temperature sensor and data logger
- Insulation foam
- X number of soil samples
- X number of meshes with a mesh size of 1x1 mm
- X number of extraction cups
- Saturated benzoic 1 ml acid (14.5 g benzoic acid and approx 1 ml detergent per 5 L)
- Manual for extractor
- Detergent
- X number of lids for extraction cups
- Incubator
- 96% ethanol (may be denatured if pure ethanol is not available)
- Small cups for transportation of extracted organisms in extraction liquid and ethanol.

Extraction procedure

1. One day before extraction: Start the refrigerator connected to the extractor as the samples may not be stored at temperatures higher than 5 °C.
2. At the day of extraction: Bring the samples carefully from the storage room to the extraction room.
3. Fill all extraction cups with a saturated solution of benzoic acid (14.5 g in 5 L) + 1 ml detergent up to 0.5 cm.
4. For each sample: Take a tube containing a soil sample. Move the label from the lid to the extraction cup. Carefully remove the upper lid and place the mesh on the tube with the sample.
5. Place a suitable cup above the soil sample unit and turn the cup with the sample around.
6. Remove the DBIdut lid from the bottom and sweep surplus soil down into the cup.
7. Place the microcosm tube with a net on an extraction cup with the benzoic acid.
8. Pour the surplus soil into the soil sample.
9. Carefully place the microcosm tube with the soil surface facing downwards into the extractor.

10. Place the insulating material around the samples when all samples are in place in the extractor. The insulation around the tubes must be placed carefully so that no soil particles will drop into the cups.
11. Connect one temperature sensor in the extractor for regulation of temperature and connect three temperature sensors to a data logger to follow the temperature during the extraction in the benzoic acid liquid, just above the mesh and on surface of the soil sample facing the heater.
12. Close the extractor.
13. Turn on the extractor and press the green start button. The extractor will now heat the samples according to this schedule:
    - 30 °C for 48 hours
    - 40 °C for 48 hours
    - 50 °C for 48 hours
    - 60 °C for 24 hours, terminated manually by switching off the power supply,

    but it may be continued until all the samples are dry on the down-facing surface on the mesh.
    The cooling system should ensure that the temperature of the benzoic acid solution is minimum 4 °C and maximum 20 °C throughout the extraction.
14. Samples with high organic matter such as peat should be divided into two horizons, e.g. the lower 3 and the upper 3 cm, and extracted independently. The samples may be divided either from the beginning of the extraction or at the temperature, e.g. 50 °C, where the upper 2 cm has become completely dry. In the latter case, the upper 2-3 centimetre is cut off the sample and discarded provided they are completely dry. The sample is removed from the extractor during this operation, to ensure that no sample material will drop into the extraction beaker.
15. The extraction is stopped manually by turning the power off.
16. Check that the samples are dry on the surface facing downwards after termination of the pre-programmed extraction process. If some samples are still wet continue the extraction at 60 °C until the samples are dry.
17. Throw the soil away
18. Brush the nets clean. Wash the tubes.
19. Add a drop of detergent to all cups in the extractor to reduce the surface tension of the benzoic acid.
20. Take the cups up from the extractor and put lids on. If there are organisms on the sides of the cups then flush or move them into the benzoic acid with a brush.
21. Put all cups with lids on into a heating oven for 24 hours at 50 °C. The heat and the detergent ensure that all organisms sink to the bottom.
22. Pour the content from each cup into plastic cups and fill with 96% ethanol in a ratio of one part water to two parts of ethanol (resulting in approx 70% ethanol). If necessary to obtain this proportion divide the sample into two plastic cups.
23. Store the samples with lids closed tightly until filtering at NERI or GNIR.
24. Draw a graph (temperature as a function of time) of the extraction in Excel and save it on the server drives. The curves are used when evaluating the results.

2.3.2 Decomposition

Organic material used for monitoring

Filter paper is generally used for litterbags. A batch of litterbags with *Salix glauca* leaves available at GNIR is positioned together with the filter paper litterbags for the 2009 litterbag study only.

Frequency of sampling

Three times during the season depending on the stage of decomposition.

Equipment to be used

- Map/GPS with positions of plots.
- Litterbags filled with 2 g VWR filter paper corresponding to 4 round pieces of filter paper, 9 cm in diameter.
- Knife.
- Preprinted labels, incl. Date, Plot Id, subsample no., x-y coordinates, plant community (*Silene, Salix, Empetrum, Loiseleuria*), Replicate Id., Initials.
- Transportation boxes.

Litterbags

VWR filter paper is used as a surrogate for indigenous litter and filled into litterbags. Each litterbag (5 mm mesh and 10 by 10 cm) is filled with 2 g of filter paper (corresponding to 4 pieces of filter paper, 9 cm in diameter). Each plot holds 10 litterbags buried horizontally 3-5 cm into the soil/peat. Each bag is identified with a unique labelling embossed on a plastic tag and placed inside the closed bag including: sampling occasion (date), habitat, plot number, subsample no., x coordinate meter, y coordinate meter. The litterbags are left open in one end for ease of emptying and further processing. Marker sticks are used to locate the litterbags at the 3 sampling occasions to ease the idenfication and retrieval.

When employing a new batch of filter-paper the dry-weight (DW) is determined by taking 5 representative samples and drying them in an oven at 50 °C until constant weight. As the filter paper take up water from the surrounding air they should be stored in an exicator if they cannot be weighed immediately after drying in the oven.

Location and marking of sampling plots

During the 2009 sampling season each of the 4 habitats will be characterised concerning pH, texture and plant communities. About 0.5 kg soil is collected and send for analysis in Foulum, Denmark according to the soil sampling procedure.

Litterbags are placed according to a random sampling scheme in the field monitoring site each autumn and the last (third) batch is collected at the same time as the placement of the next year's set. The litterbags are placed in a manner ensuring good, natural contact with the underlying litter layer. The bags are covered by some of the surrounding litter if the habitat includes a natural litter layer. If the habitat consists of peat, the bags will be put into the peat layer at max 5 cm depth. In soil habitats they are covered by approx 3 cm soil. In this case a slit is made with a shovel and the litterbag is slided into the slit and covered by the soil.

The 30 litterbags in each plot are placed in a ½ m square grid. A stick is used to fix the litterbag to the soil for easy retrieval.

Sampling method

Three sets of litterbags will be collected and brought to the lab for measurement between spring and autumn. When roughly 50% of the original plant material has disappeared from the litterbags in autumn the 3rd set of bags are collected. If less than 30% has been decomposed in the autumn, another season may be added to the duration of the decomposition period to obtain a higher decomposition rate around 50%. Each set of litterbags consists of:

4 habitats x 2 replicates x 10 litterbags (subsamples) * 3 sampling occasions = 240 litterbags.

To check the state of decomposition in addition 10 extra bags are placed at each habitat, i.e. totally 40, to monitor the current level of decomposition. Only one replicate plot may be chosen for the monitoring purpose. Three of the monitor litterbags may be collected corresponding to each sampling occasion and measured before a final date for collection is decided. Decomposition should be terminated when the remaining dry-weight is about 30%, so the rate of decomposition of the first and second sampling occasion would be about 75% and 50%.

Laboratory work

After collection the filter paper is oven-dried in paper bags at 50 °C for 24 hours or longer to ensure the mass (DW) is constant. Any mosses, lichens, fine roots, or other plant parts that have grown into the bags should be removed prior to weighing.

Input of data into database

1. Labelling of the batch of filter paper with: batch no., date
2. Dry-weight of filter paper after decomposition
3. Daily temperatures and precipitation.

2.4 Birds

Monitoring of birds consists of two elements: Breeding phenology of small passerines on an ad hoc basis and weekly samplings of bird censuses at permanent points.

2.4.1 Breeding phenology of passerines

Species to be monitored

The passerine bird species Northern wheatear *Oenanthe oenanthe*, Snow bunting *Plectrophenax nivalis*, Lapland Bunting *Carduelis flammea*, and Common Redpoll *Calcarius lapponicus* are monitored in the study area indicated in figure 2 and from census points as shown in figure 30 an table 4.

Frequency of sampling

During June and July on an ad hoc basis. Nests of breeding passerines are located ad hoc and the located nests are followed as frequently as possible until the chicks have left the nest.

Equipment

- Binoculars
- GPS
- Data forms Appendix 5A/Notebook.

Sampling method

At all visits at located nests note:

- Species
- Date
- Number of eggs/chicks
- GPS position
- Take close up photo of the nest and chicks.

Input of data into database

The position of nests is entered into an Excel file named "Bird_nests.xls" and holding the following columns: Species, Date, Observer, GPS-position, Number of eggs, number of chicks, and Remarks.

2.4.2 Point sampling

The primary objective of this study is to monitor the birds in the Kobbefjord valley. It is, however, also a very good opportunity to watch for other kinds of wildlife. The main focus is on the small passerines.

Species to be monitored

All bird species (and mammals if seen).

Frequency of sampling

Weekly during the entire field season.

Equipment

- Binoculars
- Data forms Appendix 5B/Notebook.

Location and marking of sampling plots

The observation points are located by GPS (see Figure 30 and table 4).

Sampling method

Record flock size, sex and age, special behaviour, etc. Watch for 10 minutes at each point. In the records, indicate clearly whether the observation was done within the first or last five minute period. The first period represents birds having been disturbed; the latter period represents birds resident in the area.

Table 4. Positions of bird observation points.

POINTNAME	LAT	LONG
FUGLA	64,134685	-51,385105
FUGLB	64,135155	-51,391187
FUGLC	64,134592	-51,396234
FUGLD	64,13239	-51,39359
FUGLE	64,131052	-51,38916
FUGLF	64,129385	-51,37833
FUGLG	64,131761	-51,379398
FUGLH	64,132669	-51,374116
FUGLI	64,134509	-51,363874
FUGLJ	64,135639	-51,355553
FUGLK	64,133636	-51,344558
FUGLL	64,132841	-51,336278
FUGLM	64,131031	-51,326204

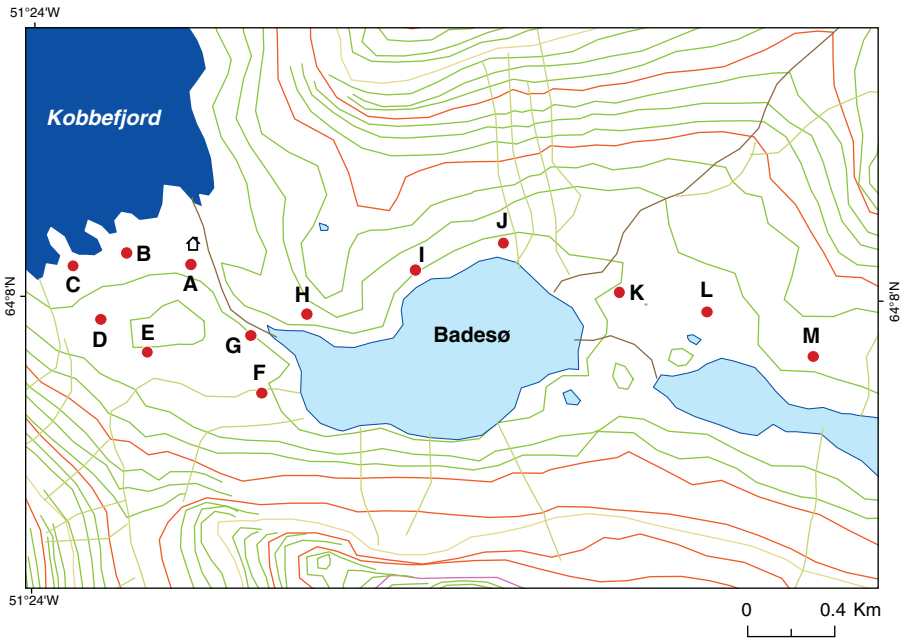


Figure 30. Bird census points. The field cabin is indicated by the symbol close to the bird census point A.

Input of data into databases

Number of birds observed at each observation point is entered into an Excel file named "Pointobservations.xls" and holding the following columns: Year, Month, Day, DOY, Time, Observation Point, Observation period, Cloud cover, Species, Age group, Gender, Number, and Comments.

2.5 Mammals

Species to be monitored

All mammal species.

Frequency of sampling

Ad hoc continuously during entire field season.

Equipment

- Binoculars (10x)
- Note book.

Location and marking of study plot

Entire activity area, see Figure 2.

Sampling method

Keep watch for everything with fur. Record geographical position, flock size, sex and age, special behaviour etc. All personnel must be encouraged to supply observations.

Input of data into data bases

Data are entered into Excel data file named "Random Obs." and holding the following columns: Year, Month, Day, Observer, Location, GPS-position, Species, Ad., Juv. and Remarks.

2.6 Lakes

Physical-chemical and biological (phytoplankton, zooplankton, fish and macrophytes) parameters.

2.6.1 Parameters to be monitored monthly

- Ice cover
- Water temperature
- Water transparency
- pH
- Conductivity
- Chlorophyll a
- Total nitrogen
- Dissolved nitrogen (nitrite/nitrate and ammonium)
- Total phosphorus
- Dissolved phosphorus (phosphate)
- Dissolved organic carbon (DOC)
- Phytoplankton (all taxonomic groups)
- Zooplankton (all taxonomic groups)
- Submerged macrophytes (all taxonomic groups). Note: only once per year.

Water samples

Frequency of water sampling

Five times per year, during June (from 50% ice cover), July, August, September and October. Preferably every 4 weeks. If the ice free period is shorter than 5 month, the period, between samplings, is reduced to fit 5 samplings into the ice free period.

Ice cover is recorded continuously via automatic cameras operated by GeoBasis.

Location and marking of sampling spots

The two sampling lakes are located in the Kobbefjord catchment area in the bottom of Kobbefjord (Badesø; Kangerluarsunnguup Tasia

64°07'48N, 51°21'23W and Qassi Sø, 64°09'11N, 51°18'18W. The sampling stations are the deepest spots in the lakes. The positions are marked with a buoy (floating during the ice free season and 2 m below the surface during the winter season) connected to the sediment traps (see text below). Positions of the sampling stations are saved in the GPS unit.

Sampling methods

At each sampling date, time, cloud cover (x/8), wind speed (m/sec.) and ice cover (% of lake area) are recorded. Use for data form Appendix 6a for recording data in the field.

Bring water sampler, tub, Secchi disc, depth sounder, sampling bottles for the sediment traps (only in August), oars and a filled outboard. Go to the sampling station and anchor the boat to the buoy (*Notice: the sediment trap is below the buoy – therefore use a long rope when tying up to the buoy and be careful not to disturb the sediment trap in calm weather*).

Before sampling, rinse all sampling gear and bottles with lake water. Measure the transparency with the Secchi disc and an accuracy of 0.1 m on the sun facing side of the boat. Keep the face close to the water surface; lower the disc until it disappears and pull it slowly upwards until it is just visible – this is the Secchi depth.

Take a depth integrated pooled water sample of approx 25 litres from 0.5 m below the surface to approximately 0.5 m above the bottom. Start from the top and avoid any sediment in the sample. If the lake sediment is disturbed and occur in the sampler a new sample shall be taken. Wait 10 minutes or move the boat away from the first sampling spot. Measure the water temperature.

On the shore, stir the water well and take 200 ml water sample for water chemistry, a 2 litre sample for Chl a, a 50 ml sample for phytoplankton enumeration (preserved with 1 ml lugol's solution) and a 15 litre 20 µm filtered zooplankton sample in a 50 ml bottle, preserved with 2.5 ml lugol's solution. All sample bottles are kept dark and cold.

In August: Go back to the sampling station and empty the sediment traps (see text below) in the two plastic bottles. At the same time the Tid-bit temperature loggers are replaced with new ones with one-hour logging intervals (one on the buoy 2 m below the surface, named "lake no"-1 and the other one on the sediment trap, named "lake no"-2). The sediment traps are placed in their right positions again. The used Tid-bits are taken back to the lab and data are downloaded.

After each sampling the dinghy must be de-inflated and stored at Qassi Sø together with the other sampling gear to prevent damaging due to foxes etc. At Badesø the dinghy is taken back to the cabin. The outboard is taken back to the cabin/laboratory after each sampling.

Last sampling. After the last sampling in October the buoy on the lake surface must be lowered to 2 m depth to prevent ice damage.

Sediment trap

A sediment trap is set up at the main station in August 2007. The trap is emptied during the first and last visit every year. Check that lines and anchoring is OK.

StowAway Tidbit Temp Loggers are replaced every August (new Tidbits should log data every 1 hour)

Vegetation

In August submerged vegetation is monitored. The method to be used for the macrophyte studies is a transect investigation to obtain a relatively good overall description of submerged macrophyte distribution, density and diversity.

In each lake approx 15 transects and approx 150 observation points are included. If great spatial variability in plant cover occurs, more transects may be needed in order to provide an adequate description.

The transects must run in a straight line from one shore to the other (i.e. including emergent and floating-leaved macrophytes). The transects should be placed parallel at equidistant intervals to cover the whole lake area, i.e. different degrees of exposure, sediment type, slope etc. should be represented. Observation points are placed with equidistant distances on the transects (Table 1).

The first and the last observation point should be close to the shore. It is important to ensure that the total open water area is covered. Reference points and terminal observation points should be determined with GPS.

At each observation point, water depth, total macrophyte coverage (%) and the species/taxa are recorded (Appendix 6B). To describe the abundance of submerged macrophytes, only the total cover of all submerged plants is measured as percentage. Macrophytes occurring in scattered or open beds of reeds are also recorded. All observations are recorded in the field as in Appendix 6B. A table should be filled in for each transect.

In practice, the investigation is undertaken by two persons; one conducts all observations (degree of total coverage, dominating species, macrophyte height and water depth). The other person follows the transect, enters all observations in a standard data sheet (Appendix 6B) and saves the co-ordinates of all observation points into a GPS unit. Observations are made using a water glass, estimating coverage in a 2x2 m area or by using the plant rake 2-3 catches in each observation point and estimating the coverage using a 0-5 scale, where 0 = 0%; 1 = >0 -5%; 2 = >5 - 25%; 3 = >25 – 50%; 4 = >50 – 75% and 5 = >75 – 100%.

Example of basic analyses of data from the transect investigation

The following observations were obtained in a transect investigation of a 75 ha lake: 25*0%, 32*3%, 40*15%, 19*38%, 15*63%, 11*88% and 8*98%, i.e. a total of 150 observations.

The relative plant-covered area (RPA) is calculated as follows:
(25*0% + 32*3% + 40*15% + 19*38% +15*63% +11*88% + 8*98%) / 150 = 27%.

This requires that all observations have been made at the same equidistant intervals all over the lake. If relatively more observations have been made in dense near-shore macrophytes, the observations should be weighted before RPA is calculated.

Laboratory work

Before filtering for Chl a, absolute conductivity, temperature (°C) and pH is measured (accuracy 1 µS and 0.1 pH unit, respectively). This is done in the field. For chlorophyll measurements 2 litre in each lake is filtered (exact volume in ml is recorded) through a 47 mm GF/C filter. The filter is folded, placed in a test tube, wrapped in alufoil, put in a zip plastic bag marked with lake name, date and filtered volume (ml) and frozen (-20 °C) until analyses.

DOC. A 100 ml subsample of the filtered water is kept dark and cold (<5 °C) in an acid washed and well rinsed bottle. Use gloves to avoid contamination, when handling the water. DOC is analysed at the University of Copenhagen.

Water samples for water chemistry are frozen until analyses.

Zooplankton and phytoplankton samples are stored in darkness – not frozen. These samples are send to NERI, Silkeborg (Aarhus Universitet, Danmarks Miljøundersøgelser, Sø-sektionen, Vejlsøvej 25, 8600 Silkeborg) following the last sampling.

Equipment for monthly field work

- A rubber dinghy with oars, outboard and anchor
- Pump for the dinghy
- Plastic bottles for sediment (two per lake, only during the first and last visit)
- A water sampler
- 30 litre tub
- Zooplankton filter
- Plastic funnel for 50 ml zooplankton bottles
- Squeeze bottle
- 2 x 50 ml glass bottles including 2% lugols solution for zooplankton and phytoplankton samples
- Plastic tubs for water chemistry (200 ml) and chlorophyll a (2 litre)
- Secchi disc
- Optic shuttle and Tidbit Coupler for the StowAway Tidbit Temp Logger
- Depth sounder with thermometer
- Life jacket
- Survival suit
- Data form Appendix 6A.

Equipment for annual vegetation sampling

- Water glass
- Plant rake with robe
- Data form Appendix 6B.
- GPS navigator.

Equipment for laboratory work

- Whatmann GF/C filters (47 mm) for Chl a, if not filtered in the field.
- Alu foil
- Small plastic tubes for Chl a filters
- Zipper bags.

2.6.2 Parameters to be monitored every 5th year

- Fish (taxonomic groups, population and for isotopes)
- Macroinvertebrates (taxonomic groups, abundance and for isotopes)
- Phytoplankton and zooplankton for isotopes
- Sediment (paleo analyses of chironomids, diatoms and cladocerans).

Fish

In each lake a maximum of 9 (1.5 m deep) sinking Lundgren biological multi mesh gill nets are used for approx 16-18 hours. Gill nets are set in the littoral and in the pelagic (in the middle of the water column) and at the bottom (benthic nets) in the late afternoon. Nets are taken the following morning. The catch is treated per net and per net type (littoral, pelagic, benthic). Each fish is given a number, identified to species and sex, and length and weight is measured.

a. Tissue samples for isotope analyses (liver and dorsal muscle): For each species, samples are taken from approx 20 fish per lake considering all size classes of fish. The tissue samples are frozen in plastic vials and marked with lake name, date, fish number (same as above) and content.
b. From the above mentioned fish, stomach content is taken as well. Stomachs are preserved in 96% ethanol in vials or 100, 200 or 300 ml jars, depending on the size of the stomach. The container is marked with lake name, date, fish number (same as above) and content.
c. From the above mentioned fish, otoliths are taken as well. These are kept in paper and marked with lake name, date, fish number (same as above) and content.

Benthic invertebrates

8 sediment cores are sampled randomly from the profundal zone of the lake (depth between 70% and 90% of maximum depth) using the kajak sampler. A visual characteristic of the sediment colour is recorded (light grey / dark grey / brown / black / layered / plant material). Each core is emptied into a small bucket in the dinghy (before taking the next). Samples are kept separate (8 buckets). Based on the content of the bucket, a visual characteristic of the sediment type is recorded (silt/clay (< 0.06 mm), fine sand (0.06-0.6 mm), course sand (> 0.6 mm)). Samples are brought to the laboratory. The following day they are filtered as much as possible on a 212 µm sieve, gravel and plant remains are removed. If animals are not counted at that time, the sample must be preserved in 96% ethanol to a final concentration of 70%. Samples are kept separate.

Sediment/paleo samples

1) 5 sediment cores are sampled from the deepest part of the lake (depth > 80% of maximum depth) using the kajak sampler. Be careful not to disturb the sediment cores when unscrewing the core form the sampler. When the cores are taken in board there must be a sharp threshold be-

tween the sediment and the water phase. If this is not the case it indicates that the core may have tipped over at the bottom and the sample must be replaced.

On shore, surface sediment (0-1 cm) from the 5 cores are pooled in a 250 ml plastic jar and marked with lake name, date, number of cores and depth (0-1 cm).

Samples are kept cool and dark.

Samples are analysed at NERI, Silkeborg.

Isotope sampling and analyses

Samples of fish (see above).

a. Benthic invertebrates: Pelagic and littoral invertebrates are kept separate. Surface sediment is sampled with a sweep net (littoral and in macrophytes) or an Ockelmann-sledge. Samples are rinsed as much as possible in the dinghy before being put into bucket (several samples from one habitat can be pooled in one bucket – qualitative sample). On shore the samples are filtered through a 212 µm mesh and invertebrates are sorted into groups in small glass jars (~5 ml) and marked with lake name, date.
b. Zooplankton is sampled as two fractions (> 140 µm and > 500 µm). Both nets are pulled after the dinghy until enough material has been collected. Samples are put into 20 ml vials.
c. Phytoplankton: A large amount of water is filtered through first a 80 µm mesh followed by a 11 µm mesh and samples are put into 20 ml vials.
d. Benthic algae: The top 5 mm from one sediment core is put into a vial. If animals are present they should be removed.
e. Stones: Fist large stones are collected in the littoral and epiphytes are scraped into a vial.
f. Periphyton: Plants are selected randomly. Plants are washed in a 1 litre jar and periphyton is scraped off. The water including the periphyton is filtered on a 20 µm mesh. Animals are removed and the filtrate is put into a vial.
g. Macrophytes: A sample is taken of the dominating taxa. Periphyton is removed and roots are avoided. The sample is put into a vial.

All samples are kept frozen in small glass jars. Keep enough material for 3 replicates of all samples.

Laboratory work

Identifying and counting of benthic invertebrates

Samples are shipped to NERI, Silkeborg together with the plankton samples. The sample is transferred to a white dissection tray and distributed evenly. Animals are sorted, identified and counted to species or genus level according to table 5 (copepoda and daphnia are not included). Most groups are identified. For the identification, a stereo microscope and 10× magnification is used.

Table 5. Benthic invertebrates, level of identification.

GROUP	NERI-no	Sub-group	STADIE	CODE (Rubin)	Level of identification
Porifera	1000000			PORIFERA	Genus
Tricladida	6000000			TRICLADI	Genus
Nematoda	18000000			NEMATODA	
Bryozoa	76000000			BRYOZOA	Genus
Prosobranchia	65000000			PROSOBRA	Species
Pulmonata	64000000			PULMONA	Species
Schizodonta				SCHIZODO	Species
Heterodonta				HETERODO	Species
	66030100	Pisidium		PISIDIUZ	Genus
Hirudinea	22000001			HIRUDINE	Species
Oligochaeta	21000000			OLIGOCHA	Family
Hydracarina	24000001			HYDRACAX	Family
Aranea				ARANEA	Species
Entomostraca				ENTOMOST	Genus
Malacostraca				MALACOST	Species
	34000001	Ostracoda		OSTRACOX	Orden
	32000001	Cladocera		CLADOCER	Genus
Ephemeroptera	44000001			EPHEMERO	Species
Zygoptera	46000100			ZYGOPTER	Genus [1)]
Anisoptera	46000200			ANISOPTE	Genus [1)]
Plecoptera	45000000			PLECOPTE	Species
Heteroptera	47000000		Im	HETEROPT	Species
			La	HETEROPT	Genus
	47010000	Corixidae		CORIXIDX	Species
Megaloptera	52000000			MEGALOPT	Species
Coleoptera	49000000		Im	COLEOPTE	Species
	49020000	Haliplidae	La	HALIPLIX	Genus
	49030000	Noteridae	La	NOTERIDX	Genus
	49040000	Dytiscidae	La	DYTISCIX	Genus
	50010000	Gyrinidae	La	GYRINIDX	Genus
	50030000	Hydrophiloidea	La	HYDROPHX	Genus
	51010001	Scirtidae	La	SCIRTIDX	Species group
	51020000	Psephenidae	La	PSEPHENX	Species
	51030000	Elmidae	La	ELMIDAEX	Genus
	51040000	Dryopidae	La	DRYOPIDX	Genus
	51050000	Chrysomelidae	La	CHRYSOMX	Genus
	51060000	Curculionidae	La	CURCULIX	Genus
Trichoptera	53000000			TRICHOPT	Species
	51010000	Hydroptilidae	La	HYDROPTX	Genus
	54080000	Limnephilidae	La (small)	LIMNEPHX	Family
Lepidoptera	56000000			LEPIDOPT	Species
Diptera	57000000			DIPTERA	Family
	58020200	Chaoborus		CHAOBORZ	Genus
	59000000	Chironomidae		CHIRONOX	Genus
	61010300	Chironomus		CHIRONOZ	Species group

[1)]: Individuals can be very small; in that case they are identified to family level.
Im: Imago
La: Larvae

Isotope samples

All isotope samples (fish, invertebrates, plankton, periphytes, macrophytes) are prepared for analyses, i.e. each sample is freeze dried, ho-

mogenized and a 5 mg sub sample is weighed in a tin capsule (ready for analyses). Preparation for analyses is undertaken in Nuuk. For analysis they shall be shipped to: University of California, Davis Stable Isotope Facility, One Shields Avenue, Mailstop#1, Davis, CA 95616 (important to make an appointment in advance).

Equipment for every 5th year field work (first year 2008)

Fish

- Gillnets
- Floaters and robe
- Sinks and robe
- Poles for nets
- Buckets for fish
- Data forms
- Ruler, balance
- Gear for the isotope sampling (see below).

Sediment

- Kajak sampler
- Sediment cores and rubber stoppers
- Equipment to process sediment cores.

Isotopes

- Tweezers, scalpels,
- Rubber gloves
- Ethanol and squeeze bottles
- Phytoplankton net (prefer 11 µm net but 20 µm is OK)
- Zooplankton net, 140 µm
- Vertical net for large zooplankton, 500 µm
- Sweep net for invertebrates, 500 µm
- Sorting trays
- Plastic vials for invertebrates
- Coolers
- Vacuum pump and filtering equipment.

2.7 Disturbance

2.7.1 Parameters to be monitored

'Person-days' spent in the area, aircraft activity over the area, boat trips to and from the area, discharges (burning of waste, human discharges into the fiord). All of these can only be monitored in periods with personnel on the station.

3 Storage of data

Data collected during the season are downloaded at the Greenland Institute of Natural Resources when returning to Nuuk. All data are typed into or transferred to the specific Excel files immediately. At GINR the data on the server are security copied every night along with the general server back up. At the end of the season a DVD with a copy of all data is send to the National Environment Research Institute, Roskilde for storage. All original data are kept at the servers at GINR. Furthermore written material is stored at GINR along with collected specimens until processed at the appropriate facilities in Denmark.

4 References

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Other

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CO_2 Flux crop scan:
http://www.hollandscientific.com/CC_Handheld.html.

Soil moisture sensor – ThetaProbe:
ThetaProbe Soil Moisture Sensor type ML2x User Manual, v1.21, May 1999. Document Code: ML2x-UM-1.21 – can be downloaded from:
http://www.delta-t.co.uk/support-article.html?article=faq2005100703502.

EGM: EGM-4 Environmental Gas Monitor For CO_2. Operator's Manual. Version 4.11. PPSystems. 48 pp.

Appendix

Appendix 1

Date	May	June	July	August	September	October
1		*2.Pinsedag*	Gas flux		Flowering phen. & arthr. traps	
2		Flowering phen. & arthr. traps	NDVI transects		Gas flux	
3		Gas flux		HandyPea		
4	Reconnaissance trip	NDVI transects		Flowering phen. & arthr. traps		
5	Flowering phen. & arthr. traps			Gas flux		NDVI transect
6	Gas flux		Birds Total flower counts			Flowering phen. & arthr. traps
7			Flowering phen. & arthr. traps		NDVI transects	Gas flux
8		Birds	Gas flux		Flowering phen. & arthr. traps	Lake K2
9		Flowering phen. & arthr. traps	Lake K1+K2		Gas flux	
10		Gas flux			Lake K1+K2	
11	NDVI transects	Lake K1		Flowering phen. & arthr. traps		
12	Flowering phen. & arthr. traps			Gas flux		
13	Gas flux		Birds Total flower counts	Lake K1+K2		Flowering phen. & arthr. traps
14			Flowering phen. & arthr. traps		Blade	Gas flux
15		Birds	Gas flux		Flowering phen. & arthr. traps	Lake K1
16		Flowering phen. & arthr. traps			Gas flux	
17		Gas flux		Veg.tr.K2		
18	Reconnaissance trip	Lake K2		Flowering phen. & arthr. traps		
19	Flowering phen. & arthr. traps			Gas flux		
20	Gas flux		Handy Pea	NDVI transects		
21			Flowering phen. & arthr. traps		Establ. of new art.traps	
22		Birds, Total flower counts	Gas flux		Flowering phen. & arthr. traps	
23		Flowering phen. & arthr. traps	NDVI transects		Gas flux	
24		Gas flux		Veg.tr.K1		
25	Birds	Microarthropod		Flowering phen. & arthr. traps		
26	Flowering phenology & arthropod			Gas flux		
27	Gas flux		Handy Pea			
28			Flowering phen. & arthr. traps			
29		Birds	Gas flux		Flowering phen. & arthr. traps	
30		Flowering phen. & arthr. traps	Microarthropod		Gas flux	
31				Microarthopod		

Appendix 2A.

SALIX

BioBasis Nuuk 2009 Date: **Observer:**

Time	Cloud cover	Plot no.	Sample	Snow	Buds	M flowers	F flowers	Hairs	Fungus	Larvae	Total	Remarks
:		1	A									
:			B									
:			C									
:			D									
:		2	A									
:			B									
:			C									
:			D									
:		3	A									
:			B									
:			C									
:			D									
:		4	A									
:			B									
:			C									
:			D									

COUNT 100

Appendix 2B.

SILENE

BioBasis Nuuk 2009 Date: **Observer:**

Time	Cloud cover	Plot no.	Sample	Snow	Buds	Flowers	Senescent	Total	Remarks
:		1	A						
:			B						
:			C						
:			D						
:		4	A						
:			B						
:			C						
:			D						
:		2	A						
:			B						
:			C						
:			D						
:		3	A						
:			B						
:			C						
:			D						

COUNT 50

Appendix 2C.

LOISELEURIA

BioBasis Nuuk 2009 Date: **Observer:**

Time	**Cloud cover**	**Plot no.**	**Sample**	**Snow**	**Buds**	**Flowers**	**Senescent**	**Total**	**Remarks**
:		1	A						
:			B						
:			C						
:			D						
:		2	A						
:			B						
:			C						
:			D						
:		3	A						
:			B						
:			C						
:			D						
:		4	A						
:			B						
:			C						
:			D						

COUNT 100

Appendix 2D.

BioBasis Nuuk 2009 Date: ERIOPHORUM Observer:

Time	Cloud cover	Plot no.	Sample	Snow	Buds	Flowers	Senescent	Total	Remarks
:		1	A						
:			B						
:			C						
:			D						
:		2	A						
:			B						
:			C						
:			D						
:		3	A						
:			B						
:			C						
:			D						
:		4	A						
:			B						
:			C						
:			D						

ONLY TOTAL COUNTS AT PEAK-FLOWERING

Appendix 3A.

BioBasis Nuuk 2009 Date: EGM-4: A/B

		Record nos.				Soil_moisture			Chamber_height				Observer:
Time	Plot	Light	Dark	Cloud cover	Photo	1	2	3	1	2	3	4	Remarks
:	1C	-	-										
:	1SG	-	-										
:	1S	-	-										
:	1LG	-	-										
:	1T	-	-										
:	2SG	-	-										
:	2LG	-	-										
:	2S	-	-										
:	2C	-	-										
:	2T	-	-										
:	3C	-	-										
:	3T	-	-										
:	3SG	-	-										
:	3LG	-	-										
:	3S	-	-										
:	4LG	-	-										
:	4SG	-	-										
:	4S	-	-										
:	4T	-	-										
:	4C	-	-										
:	6S	-	-										
:	6SG	-	-										
:	6C	-	-										
:	6T	-	-										
:	6LG	-	-										

Appendix 3B.

Carbon Flux

BioBasis Nuuk 2009 Date: EGM-4:

		Record nos.				Soil_moisture			Chamber_height				Observer:
Time	Plot	Light	Dark	Cloud cover	Photo	1	2	3	1	2	3	4	Remarks
:	5T	-	-										
:	5S	-	-										
:	5LG	-	-										
:	5SG	-	-										
:	5C	-	-										
:		-	-										
:		-	-										
:		-	-										
:		-	-										
:		-	-										
:		-	-										
:		-	-										
:		-	-										
:		-	-										

Appendix 3C.

SALIX in ITEX-PLOTS

BioBasis Nuuk 2009 Date: Observer:

Plot no.	Snow	Buds	M flowers	F flowers	Hairs	Fungus	Larvae	Total	Remarks	Plot no.	Snow	Buds	M flowers	F flowers	Hairs	Fungus	Larvae	Total	Remarks
1C										6T									
1SG										6LG									
1S										5T									
1LG										5S									
1T										5LG									
2SG										5SG									
2LG										5C									
2S																			
2C																			
2T																			
3C																			
3T																			
3SG																			
3LG																			
3S																			
4LG																			
4SG																			
4S																			
4T																			
4C																			
6S																			
6SG																			
6C																			

Appendix 4.

ARTHROPODS

BioBasis Nuuk 2009 Date: Observer:

Time	Plot no.	Sample	Snow	Pitfall opened	Remarks
:	1	A			
:	1	B			
:	1	C			
:	1	D			
:	1	E			
:	1	F			
:	1	G			
:	1	H			
:	2	A			
:	2	B			
:	2	C			
:	2	D			
:	2	E			
:	2	F			
:	2	G			
:	2	H			
:	3	A			
:	3	B			
:	3	C			
:	3	D			
:	3	E			
:	3	F			
:	3	G			
:	3	H			
:	4	A			
:	4	B			
:	4	C			
:	4	D			
:	4	E			
:	4	F			
:	4	G			
:	4	H			

Appendix 5A.

BioBasis Nuuk 2009 Bird nests

GPS					

Date					**Remarks/Photo**

Appendix 5B.

BioBasis Nuuk 2009 Date: Bird countings

				Snow bunting			Lapland bunting			Red poll			Northern wheatear			Observer:
Time	Site	State	Cloud_ Cover	M	F	UK	M	F	UK	M	F	Uk	M	F	UK	Remarks / Other birds
:	A	Initial														
:	A	Obs														
:	B	Initial														
:	B	Obs														
:	C	Initial														
:	C	Obs														
:	D	Initial														
:	D	Obs														
:	E	Initial														
:	E	Obs														
:	F	Initial														
:	F	Obs														
:	G	Initial														
:	G	Obs														
:	H	Initial														
:	H	Obs														
:	I	Initial														
:	I	Obs														
:	J	Initial														
:	J	Obs														
:	K	Initial														
:	K	Obs														
:	L	Initial														
:	L	Obs														
:	M	Initial														
:	M	Obs														

When arriving at a new site, count the numbers you see within the first 5 min (Initial). Then start your observation (Obs) for 5 min.
If a bird is seen within the Initial period AND the Obs period let it only count in the Obs period

Appendix 6A.

BioBasis Nuuk 2009 Lake: Observer:

Date	Time	Ice cover	Cloud cover	Wind speed	Secchi depth	Depth	Temperature, surface	pH	Conductivity	Remarks
	:									
App. 25 litres of pooled water from 0.5m below the surface to 0.5m above the bottom							depth m/temperature C			
Samples taken from pooled water to:							/			
Water chemistry (200ml, wrap with tin foil)							/			
Chl a (1 litre, wrap with tin foil)							/			
Phytoplankton (50ml preserved with 1 ml lugol)							/			
Zooplankton(15 litre filtred through 20µm filter into 50ml bottle with 2.5ml lugol)							/			
							/			
pH and conductivity are measured on the pooled water							/			
							/			
							/			
							/			

Appendix 6B.

Data sheet used to record submerged macrophytes on a transect.

Country: __Cloud cover (x/8): _____ Wind speed (m/sec.): _____

Lake: Position (UTM): _____ Datum: _____

Date: Transect no.:

		Dominant species						Total	Height	Depth
Observation	**waypoint**							coverage %	m	m
	no.									
1										
2										
3										
4										
5										
6										
7										
8										
9										
10										
11										
12										
13										
14										
15										
16										
17										
18										
19										
20										

Species list: 01:00 2: 3:

04:00 5: 6:

07:00

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National Environmental Research Institute
Frederiksborgvej 399
PO Box 358
DK-4000 Roskilde
Denmark
Tel: +45 4630 1200
Fax: +45 4630 1114

Management
Department of Arctic Environment
Department of Atmospheric Environment (main address)
Department of Environmental Chemistry and Microbiology
Department of Marine Ecology (main address)
Department of Policy Analysis

National Environmental Research Institute
Vejlsøvej 25
PO Box 314
DK-8600 Silkeborg
Denmark
Tel: +45 8920 1400
Fax: +45 8920 1414

Department of Freshwater Ecology
Department of Marine Ecology
Department of Terrestrial Ecology

National Environmental Research Institute
Grenåvej 14, Kalø
DK-8410 Rønde
Denmark
Tel: +45 8920 1700
Fax: +45 8920 1514

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CONCEPTUAL DESIGN AND SAMPLING PROCEDURES OF THE BIOLOGICAL PROGRAMME OF NUUKBASIC

This manual describes procedures for biologic climate effect monitoring in Kobbefjord, Nuuk. The monitoring is a part of NuukBasic which is a cross-disciplinary ecological monitoring programme in low Arctic West Greenland. Biological monitoring comprises the NERO line which is a permanent vegetation transect, and monitoring reproductive phenology of *Salix glauca, Loiseleuria procumbens, Eriophorum angustifolium*, and *Silene acaulis*. The progression in vegetation greenness is followed along the vegetation transect and in the plant phenology plots by measurement of Normalized Difference Vegetation Index (NDVI). The flux of CO_2 is measured in natural conditions as well as in manipulations simulating increased temperature, increased cloud cover, shorter growing season, and longer growing season. The effect of increased UV-B radiation on plant stress is studied by measuring chlorophyll fluorescence in three series of plots. Arthropods are sampled by means of yellow pitfall traps and in window traps. Microarthropods are sampled in metal cores and extracted in an extractor by gradually heating up soil. The rate of decomposition is measured in three habitats. The avifauna is monitored with special emphasis on passerine birds. Only few terrestrial mammals occur in the study area. All observations of mammals will be recorded ad-hoc. Monitoring in lakes include ice cover, water chemistry, physical conditions, species composition of plankton, vegetation, bottom organisms and fish. Physical-chemical parameters, phytoplankton and zooplankton are monitored monthly in the period when the lakes are ice-free.

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