

Zackenbergskeologiske Forskningsoperationer

BioBasis Manual

Conceptual design and sampling procedures of the biological monitoring programme within Zackenberg Basic



22nd edition



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BioBasis, Conceptual design and sampling procedures of the biological monitoring programme within Zackenberg Basic.

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Front cover illustration: Dwarf fireweed (*Chamerion latifolium*) at Zackenberg. Photo by Niels M. Schmidt.

Preface

BioBasis Zackenberg monitors key species and key processes across plant and animal populations and their interactions within the terrestrial and limnic ecosystems at Zackenberg, thereby documenting the intra- and inter-annual variation, resilience and long-term trends. Emphasis is on biodiversity, abundance and composition, phenology, reproduction and predation as important components in the structure and functioning of the high arctic ecosystems:

1. **Flora** (phytoplankton, lichens, mosses and vascular plants), including species diversity, community composition, abundance and phenology, herbivory, and greening patterns.
2. **Invertebrates** (zooplankton, arthropods) biodiversity, community composition, abundance and emergence phenology.
3. **Vertebrates** (fish, birds and mammals), including species diversity, community composition, abundance, breeding phenology, and predation.

Additional biotic (e.g. NDVI) and abiotic parameters (e.g. snow-melt patterns, soil moisture and water chemistry) related to biota are monitored, complementing other subprograms.

BioBasis aims at maintaining the integrity of the long core time series available from Zackenberg, whilst developing the monitoring to embrace new methodologies and questions. Field protocols for monitoring elements that are no longer part of the BioBasis monitoring programme can be found in previous versions of the manual.

Niels Martin Schmidt, April 2019.

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Superior notice on data handling and quality

It is of decisive importance that data obtained by BioBasis are handled with the greatest care. They all cost a lot of time, money and effort, and in many cases, they cannot be regenerated if lost.

While at Zackenberg, data files must be backed up each time new data have been added. Use two alternate discs, so that any transfer error does not destroy the copy already worked up.

At the end of the season, the entire set of data files must be copied on two separate sets of computers, portable hard disks or memory sticks. Each set is kept separate during the homeward travel, so that one e.g. goes with the shipped goods, one with the personal luggage and the computer itself is kept with the hand luggage.

Hand written notes and forms must be digitized as soon as possible in the field and at the return to the Department of Bioscience, Aarhus University. If still not digitized when leaving Zackenberg, these notes and forms must be kept in the hand luggage.

While at Zackenberg, all data must be typed into / transferred to the specific data files. Note that the use of PDAs in the BioBasis data collection is the primary tool during the field season. In addition, if possible, spatial data should be entered into GIS for further processing and validation. At home at the Department of Bioscience, Aarhus University, all data files are immediately copied to the main server, and these copies are updated each time the files have been supplemented or edited. The original data sheets from the field season are kept in separate places until the annual data set is uploaded to the database.

The quality of the data must be validated as soon as possible after collection, preferably the same day. In addition, each year in connection with the final editing and preliminary analysis following each season any data diverging from expected values must be checked carefully and discussed with the observer.

During the field season, keep a diary with daily notes on the weather conditions, work tasks done, problems, and ideas for improvements of techniques and procedures.

BioBasis

Conceptual design and sampling procedures of the biological programme of Zackenberg Basic

General outline of BioBasis

The long-term 'baseline studies' of BioBasis embrace the 38 elements listed below. They have been selected to cover a wide variety of trophic levels in the local ecosystem as well as for their suitability for standardised monitoring. Together, they aim to produce a coherent set of numerical and phenological data facilitating the understanding of the intricate dynamics of a terrestrial High Arctic ecosystem. In combination with the parallel climatological and physical geographical parts of Zackenberg Basic, Climate-Basis and GeoBasis, it will be possible to evaluate the effects of short- and long-term changes in the environment.

Overview of monitoring elements

Plants

- 1.1 In 48 plots, ranging from early to late snow-free plant communities, reproductive phenology (flowering) is recorded for *Cassiope tetragona* (4 plots + 10 ITEX plots), *Dryas* spp. (6), *Papaver radicum* (4), *Salix arctica* (7 + 10 ITEX plots), *Saxifraga oppositifolia* (3) and *Silene acaulis* (4) on a weekly basis during the growing season (late May – early September).
- 1.2 In the study plots mentioned under 1.1, the total number of flowers is recorded at peak of flowering each summer. The same applies to the four study plots with *Eriophorum scheuchzeri*.
- 1.3 An 8.8 km transect – the 'ZERO line' – running from sea level to 1040 m a.s.l. was established in 1992 and 1994. In connection with the establishment, all significant vegetation pattern changes along the line were recorded and marked, using numbered pegs. Future changes between competing plant communities can be measured by referring to these pegs. The transect is re-examined with five year intervals by a specialist.
- 1.4 One plant community plot (20 m x 20 m) is situated on a *Dryas* heath north of the east end of the airstrip along the ZERO line (see element no. 1.3). Within this plot, the distribution of plant species has been mapped in detail, so that even minute changes can be detected. A specialist will do the checking of each plot at five-year intervals, in connection with no. 1.3.
- 1.5 In 25 cryptogam plots along the ZERO line, the changes in the cryptogamic communities are examined by a specialist with five-year intervals.
- 1.6 In 18 cryptogam plots, changes in the cryptogamic communities are examined by a specialist with five-year intervals.

- 1.7 In nine ITEX point frame study plots, each with five frame plots in typical plant communities, the composition and growth is analysed by a specialist with five year intervals.
- 1.8 Five northern range plant species are monitored every fifth year. *Salix herbacea*, *Campanula giesekiana*, *Carex glareosa*, *C. lachenalii*, and *C. norvegica*.
- 1.9 In each plot, mentioned under element no. 1.1 and 1.2 and in four pitfall trapping stations (see element no. 2.1) weekly Normalized Vegetation Difference Index (NDVI) is measured.
- 1.10 Three NDVI transects are walked four times during the field season.
- 1.11 Annual NDVI analyses from satellite images are made for 14 sections in Zackenbergdalen.
- 1.12 Soil moisture at permanent plots
- 1.13 Vegetation composition in ITEX and UV plots is examined annually.
- 1.14 Vegetation composition in three GLORIA summits is checked every 5 years by a specialist.

Arthropods

- 2.1 Faunistic and phenological collections are carried out by means of yellow pitfall traps placed in five different plant communities. The traps are emptied on a weekly basis throughout the summer season (late May / early June – late August / early September). Arthropods are sorted into taxonomic groups, counted and preserved at the Museum of Natural History, Aarhus.
- 2.2 Two 'window traps' are placed at a pond near the station to monitor limnic insect emergence and aerial activity. The traps are emptied on a weekly basis throughout the summer season. Arthropods are sorted into taxonomic groups, counted and preserved at the Museum of Natural History, Aarhus.
- 2.3 Predation by larvae of the moth *Sympistis zetterstedtii* on *Dryas* flowers is recorded weekly in six plant plots (see element no. 1.1) during the summer season.

Birds

- 3.1 Populations of all breeding bird species are mapped annually during June-July in a 15.8 km² census area (0-600 m a.s.l.).
- 3.2 Breeding phenology (first egg dates, hatching, fledging) is monitored annually together with nest predation in the census area mentioned under element no. 3.1.
- 3.3 Barnacle goose broods in Zackenbergdalen are monitored during the fledging period.
- 3.4 Breeding common eiders, Sabine's gulls and Arctic terns are censused on Sandøen in late July, if feasible.
- 3.5 Other bird observations are recorded throughout the entire field season, including flocks of moulting geese.

Mammals

- 4.1 Winter nests of collared lemmings are examined and mapped annually in a 1.06 km² study area.
- 4.2 A complete census of musk oxen is performed within a 47 km² census area on a weekly schedule between 1 July and 30 August. All musk ox groups within this area are identified for age and sex composition.
- 4.3 All Arctic fox dens in Zackenbergdalen are checked regularly during the summer field season for occupation and pups.
- 4.4 Arctic hares present on the east facing slope of Zackenberg mountain are recorded every third day during July-August.
- 4.5 Walrus hauls out on Sandøen are counted, sexed and aged in mid/late July if appropriate.
- 4.6 Other observations of mammals are recorded throughout the entire field season.

Collection of wildlife samples

- 5.1 Tissue samples are taken from all wildlife carcasses encountered. Tooth samples are also taken from musk ox carcasses. Samples of hair and feathers are collected opportunistically, except for musk oxen and arctic fox that are collected annually.
- 5.2 Faeces from stoat and Arctic fox, and casts from long-tailed skua and snowy owl are collected at selected stones at the end of each season.

Lakes

- 6.1 Physical-chemical parameters, phytoplankton and zooplankton are monitored minimum four times per season in two lakes, one with and one without Arctic char.

Abiotic parameters

- 7.1 In each plot, mentioned under elements no. 1.1 1.2, 1.4, 2.1 and 2.2, the snow-cover is estimated at weekly intervals during snow melt.

Disturbance

- 8.1 Records are kept of activities (man-days and ATV trips) in the different sectors of the study area, particularly in the 'low-impact' study area east of Grænseelv and in the goose moulting area along the coast east of the old delta of Zackenbergelven. Records are also kept of aircraft operations in and around Zackenbergdalen, and of waste water and other discharges into Zackenbergelven etc. Finally, records, including UTM coordinates are kept of all manipulative research projects and of all studies involving take of organisms.

Detailed manual for BioBasis

1. Plants

1.1. Reproductive phenology

1.1.1. Species to be monitored

- White Arctic bell-heather *Cassiope tetragona* (kantlyng)
- Mountain/ Arctic avens *Dryas integrifolia/octopetala* (fjeldsimmer)
- Arctic poppy *Papaver radicum* (fjeldvalmue)
- Arctic willow *Salix arctica* (polar-pil)
- Purple saxifrage *Saxifraga oppositifolia* (purpur-stenbræk)
- Moss campion *Silene acaulis* (tue-limurt)

1.1.2. Frequency of sampling

At weekly intervals during late May/early June - late August/early September (see fixed dates in table 1.1.2). If inclement weather prohibits sampling, the work may be postponed to the following day.

Table 1.1.2. Sampling dates for weekly plant reproductive phenology and arthropod censuses. June, July and August cover the ordinary field season.

May	June	July	August	September
6	3	1	5	3
13	10	8	12	10
20	17	15	19	17
27	24	22	26	24
		29		31

1.1.3. Equipment to be used

- Map with position of study plots
- GPS
- PDA / dictaphone / notebook
- Knee pads
- Boardwalks
- Tally counters (*kliktællere*)

1.1.4. Location and marking of study plots

The position of the 48 study plots in the lowland is shown on fig. 1.1.4. Each plot is marked with angular aluminium bars in the corners and provided with a number plate. The plots are divided into four sections (quarters A, B, C and D separated by steel pegs) starting from the number plate and running either straight from there or

clockwise around the centre. UTM co-ordinates, dimensions etc. appear from table 1.2.2.

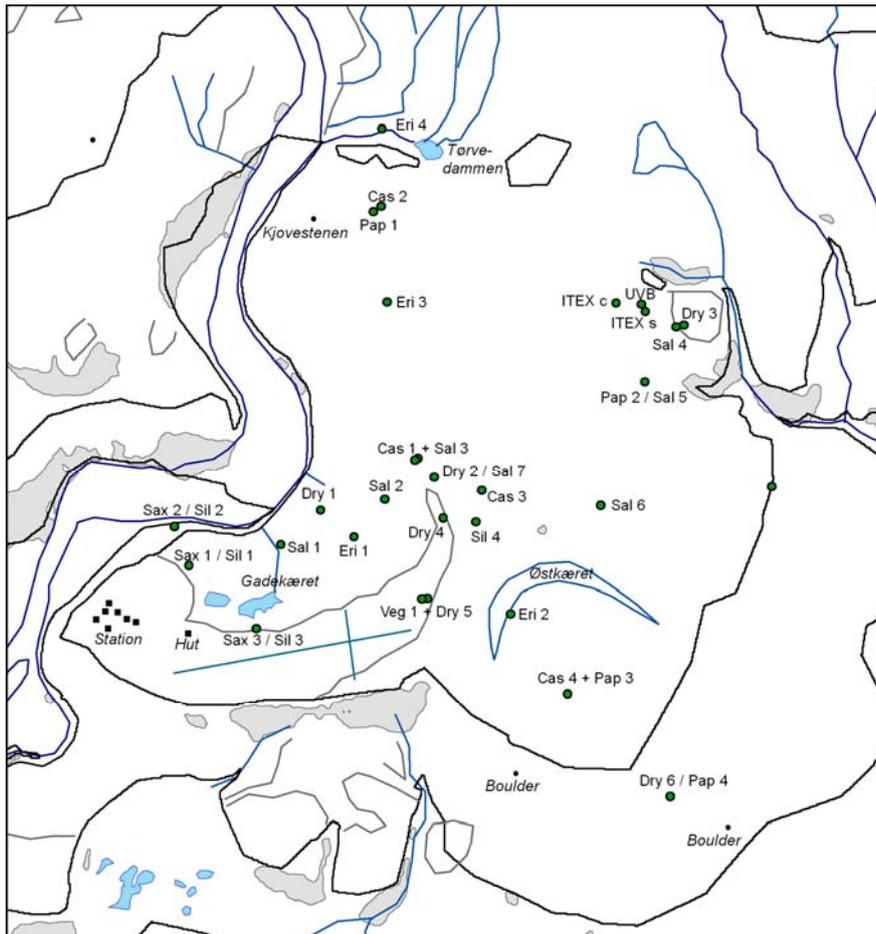


Fig. 1.1.4. Location of the plant phenology plots and the ITEX and UV plots. *Salix5* was closed in 2017.

1.1.5. Sampling method

During snowmelt in June, percent snow cover in each plot section is estimated at each sampling trip (including *Eriophorum* plots, and Veg 1 plot; see sections 1.2, 1.4). 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. Visit the plots every 2-3 days to estimate snow cover.

When the snow is gone, boardwalks are used at all *Cassiope* plots and *Dryas* plot 3. Carry the boardwalk some metres away, when not used and place it 10-20 m downwind (to the south) at the end of the season. Additional board walks are located at the station and can be used when needed.

At each visit, samples of a total of at least 50 flower buds, flowers and senescent flowers (or capsules with exposed seeds) are recorded within each plot section. In the *Salix* plots, a total of at least 100 buds, flowers and senescent flowers

are recorded. This is done by counting the different phenological stages within appropriate group sizes of individuals and dictating subtotals to the dictaphone concomitantly until a total of over 50/100 is achieved. Check now and then that the dictaphone is recording. Begin to the right in each section and count towards the left.

Finally, NDVI measurements are taken in each section of all plant plots (see section 1.9).

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 procedure applies in particular:

Cassiope

Buds are visible in both reddish and green shoots as small whitish shoots, located distally from the shoot apex. They are best seen when looking at the shoots straight from above. Vegetative shoots are green. Most petals fall off as one unit before they wither. The fruit bodies are still counted as senescent flowers.

Dryas

Apart from sampling of buds, flowers and senescent flowers, record also the number of buds and flowers where the reproductive organs have been partly or fully eaten by larvae (of *Sympistis zetterstedtii*). Such buds and flowers must still be included in the number of buds and flowers, respectively, i.e. the eaten buds or flowers appear twice in the spreadsheet.

Papaver

Petals either fall off or turn dark green and fold around the capsule (late in the season). Both types are recorded as senescent flowers. Open capsules with exposed seeds must also be recorded. In cases where the petals are folded around the capsule, it may be necessary to remove the top of these to check if the capsule is open. Finally, the capsules may fall off. Such 'flowers' can still be separated from last year's stems by their hairy appearance, and they must still be recorded among the 'senescent flowers with exposed seeds'.

If the plot has been grazed by e.g. musk ox, the number of eaten capsules (represented by a flower stem from this year without a capsule) must be recorded. This only happens while the capsules are still green.

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 when no stigmas or anthers are visible, and as flowers, as soon as

stigmas or anthers are visible (they are both red in the early stages). Buds cannot be sexed, so male and female buds are pooled.

The transition to senescent flowers and fruits is not recorded. Both types continue to be recorded as 'flowers' until they are recorded as having exposed seed hairs from the time of exposure of the first hairs on top of the splitting capsules.

Silene

A problematic species, since one or a few individuals may dominate the sample. Therefore, several individuals (preferably the same) must be sampled each week. Flower buds are reddish. Senescent flowers are defined as flowers with faded petals and empty pollen anthers.

Saxifraga

Flower buds are reddish. Senescent flowers are defined as flowers with faded petals. The number of open capsules (seeds exposed 5-8 weeks after senescence) is part of the sampling. New buds and even flowers may develop in late autumn. These are counted, but they are not included in the calculation of reproductive phenology presented in the annual reports.

1.1.6. Laboratory work

None.

1.1.7. Input of data into database

The data from the weekly registrations in the plots are entered into Excel data files with columns relevant for each species. The basic data are: Date (YYYY-MM-DD), Observer, Plot (e.g. Dry1), Sample (sector A, B, C and D), Snow (percent per section), Buds (give actual numbers counted, not percent), Flowers, Senescent flowers, Total (sum of buds, flowers and senescent flowers at the weekly count), and Remarks. Specific columns for individual species appear from the database files.

1.2. Total flowering

1.2.1. Species or taxonomic groups to be monitored

As in 1.1, plus Arctic cotton-grass *Eriophorum scheuchzeri* (polar-kæruld) and 'dark' cotton-grass *Eriophorum triste* (mørk kæruld).

1.2.2. Frequency of sampling

Once per season (table 1.2.2). The optimal time is when most or all flower buds have bursted. All sections of a plot are counted on the same day.

Table 1.2.2 Position, dimensions, orientation and approximate counting period for all flowering study plots. The different counting periods within the species are determined by snowmelt. *Salix 5* was closed in 2017

Plot no.	UTM E	UTM N	Size (m)	A corner	Counting period
Cassiope 1	513,300	8,264,809	1x2	SE	Early-late July
Cassiope 2	513,230	8,265,290	1x3	SE	Mid-late July
Cassiope 3	513,420	8,264,746	1x2	E	Early-late July
Cassiope 4	513,583	8,264,358	1x3	NE	Early-late July
Dryas 1	513,116	8,264,708	1x4	N	Early July
Dryas 2	513,330	8,264,771	6x10	E	Late July-August
Dryas 3	513,802	8,265,062	1x2	N	Early-mid July
Dryas 4	513,348	8,264,694	2x3	N	Early-mid July
Dryas 5	513,317	8,264,541	2x3	NW	Late June-mid July
Dryas 6	513,776	8,264,161	7x13	S	Mid July-August
Papaver 1	513,215	8,265,280	7x15	W	Mid July-early Aug
Papaver 2	513,729	8,264,955	10x15	E	Mid July-early Aug
Papaver 3	513,575	8,264,353	9x10	W	Mid July-early Aug
Papaver 4	513,776	8,264,161	7x13	S	Mid July-early Aug
Salix 1	513,041	8,264,643	6x10	W	Mid-late June
Salix 2	513,237	8,264,729	15x20	W	Early-late July
Salix 3	513,294	8,264,805	6x6	S	Mid June-mid July
Salix 4	513,788	8,265,058	10x15	E	Late June-mid July
Salix 5	513,729	8,264,955	10x15	E	Early-mid July
Salix 6	513,645	8,264,718	10x15	NE	Early-mid July
Salix 7	513,330	8,264,771	6x10	E	Early-mid July
Saxifraga 1	512,867	8,264,603	2x3.5	W	Mid June
Saxifraga 2	512,839	8,264,677	2x3	NE	Mid June
Saxifraga 3	512,994	8,264,433	2x5	NW	Mid-late June
Silene 1	512,867	8,264,603	2x3.5	W	Late June-mid July
Silene 2	512,839	8,264,677	2x3	NE	Late June-mid July
Silene 3	512,994	8,264,433	2x5	NW	Late June-late July
Silene 4	513,409	8,264,686	1x1	NW	Mid July-mid August
Eriophorum 1	513,179	8,264,657	3x5	N	Mid July-late August
Eriophorum 2	513,475	8,264,510	3x5	NW	Mid July-late August
Eriophorum 3	513,241	8,265,106	2x3	E	Mid July-late August
Eriophorum 4	513,232	8,265,437	2x4	SE	Mid July-late August

1.2.3. Equipment to be used

- Map with position of study plots
- Pieces of cord totalling 100 m
- Flower sticks
- PDA / dictaphone / notebook
- Knee pads
- Tally counters (klikktællere)

1.2.4. Location and marking of sampling plots

See fig. 1.1.4 and table 1.2.2. Each plot is divided into four sections, denoted A-D clockwise or straight, starting from the number plate.

1.2.5. Sampling method

Tighten a cord around each section of the plot. In large plots, subsections are established by placing two additional cords with about 1 m intervals from one end of each section, whereupon the number of **flower buds**, **flowers** and **senescent flowers** are counted separately 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. Dictate the results to the dictaphone at least for every 100 recordings.

In *Pavaver* plots, white flowers should be counted and given under Remarks, but still included in the total number of flowers. In the *Salix* plots, male and female catkins are counted separately. Catkins that have been grazed, but can still be sexed, are included. In the *Eriophorum* plots, both the number of *Eriophorum scheuchzeri* and *Eriophorum triste* inflorescences must be recorded, and they must be separated into fertile and infertile flowers. Infertile flowers have poorly developed white hairs and the stem turns brown long before the stems of the fertile flowers.

1.2.6. Laboratory work

None.

1.2.7. Input of data into database

See section 1.1.7. Make sure to enter the individual numbers for buds, flowers and senescent flowers (and other relevant information) for each plot section.

1.3. The ZERO line

1.3.1. Species or taxonomic groups to be monitored

All vascular plants species. The nomenclature is according to Böcher et al., Grønlands Flora, P. Haase & Søn's Forlag, 1978. Species of mosses and lichens are referred to as 'moss' or 'lichen'. The ZERO line vascular plants are examined by an expert botanist.

1.3.2. Frequency of sampling

Every 5th year during the peak season from mid July to mid August.

1.3.3. Equipment to be used

- A Raunkiær circling stick (metal angle)
- List of peg positions
- GPS
- Data sheets
- Knee pads
- Digital camera
- A hammer and extra aluminium tubes
- Notebook / dictaphone

1.3.4. Location and marking of the transect line

The ZERO-line consists of 130 numbered pegs running from the edge of the old delta to the top of Aucellbjerg. Each peg is placed at the borderline between distinct plant communities. Between each peg, 10 aluminium tubes denote the centres for Raunkiær circles. Five tubes (nos. 1-5) are situated with two metre intervals upwards from the peg and the last similarly before the next peg and again numbered upwards along the line (nos. 6-10). In plant communities less than 22 m wide, some tubes are placed on a right angle to the line, midway in the community and beginning on the western side. The exact locations of all pegs are given in the Excel data file named Plot positions and stored at Bioscience AU.

1.3.5. Description of the sampling method

The Raunkiær circling stick with marks indicating the radius of the three circles is placed in the tube and all vascular plant species within the three circles are recorded. A species gets 1 point if rooted (or if a dwarf shrub has its buds within) only the 1/10 m², 2 if found within the 1/100 m², and 3 if found within the 1/1000 m² circle. Thus, the maximum score in 10 circles is 30. In addition it is recorded if the species is sexually reproductive (buds, flowers or fruits) within the 1/10 m² circle. Photos are taken of all plots from the eastern side of the line and along the borderline between the plant communities. Specimens, which are impossible to identify due to lack of flowers, fruits or other diagnostic characters are given by the genus name or by cfr. (e.g. *Draba cfr. lactea*). Later recordings during summers with better conditions will reveal the species.

Take a digital orthophoto of each Raunkiær circling.

1.3.6. Description of laboratory work

None.

1.3.7. Input of data into databases

Data from the plots are entered into an Excel data file with the columns Peg no., Tube no., Date (YYYY-MM-DD), Observer and species names. Uncertain species identifications have cfr. (=confer) added to indicate the need for further confirmation at next survey. Fertility is given by an "f" in a separate column after the species column. See also: Fredskild & Mogensen (1997) and Bay (1998).

Digital pictures are downloaded and renamed to include Year, Peg number and Circle number (e.g. 2008_34_2).

1.4. Plant community plots

Originally, four 20x20 m plant study plots were established in 1992, but only one (Veg 1) was analysed (see Fredskild & Bay 1993). Since then, only snowmelt has been recorded each spring, until Veg. 2-4 were closed down in 2001. Now, only snowmelt in Veg. 1 is recorded annually during melt-off (see section 1.1.5 and 7.1).

1.5. Lichen analyses along the ZERO line

1.5.1. Species or taxonomic groups to be monitored

All lichen taxa. The nomenclature should be in accordance with Santesson 1993 and Hansen 1995. The ZERO line lichens are examined by a lichen expert.

1.5.2. Frequency of sampling

July-August every 5th year.

1.5.3. Equipment to be used

- A Raunkiær circling stick (metal angle)
- List of peg positions
- GPS
- Notebook / dictaphone
- Magnifying glass
- Digital camera
- Hammer and extra aluminium tubes

1.5.4. Location and marking of sampling plots

The exact location of all the pegs are given in the Excel data file named Plot positions. 10 aluminium tubes mark the centre of the Raunkiær circles. Five tubes are situated southeast of the central yellow tube and five northwest of it. The 10 circles are numbered from southeast to northwest.

1.5.5. Description of sampling method

The Raunkiær circling stick with marks indicating the radius of the three circles is placed in the tube and all vascular plants and lichens within the three circles are recorded. A species obtains 1 point if situated only in the 1/10 m², 2 if located within the 1/100 m² and 3 if situated within the 1/1000 m² circle. "Cfr" indicates that no precise identification of the plant has been possible. Take a digital ortho-photo of each circle.

1.5.6. Description of laboratory work

Only very small pieces of the lichens are sampled in order to avoid damaging the lichens. Standard TLC or HPTLC methods are used for identification of selected specimens of sterile crustose lichens such as, for example, *Lepraria*. The analyses are carried out at the Botanical Institute, University of Copenhagen.

1.5.7. Input of data into databases

Data from the ZERO line are entered into an Excel data file: Lichen ZERO line plots with the columns: Peg no., Circle no., Circle (1 to 10), Date (YYYY-MM-DD), Observer, Species, Points (1-3), Remarks.

Digital pictures are downloaded and renamed to include Year, Peg number, and Circle number (e.g. 2008_23_3).

1.6. Lichen square plots

1.6.1. Species to be monitored

All lichen taxa, both terricolous and saxicolous lichens. The nomenclature is in accordance with Santesson, 1993 and Hansen, E. S. 1995. The lichen square plots are examined by a lichen expert.

1.6.2. Frequency of sampling

July-August every 5th year.

1.6.3. Equipment to be used

- Folding ruler
- List of plot positions
- GPS
- Notebook
- Digital camera
- Magnifying glass
- Hammer and extra aluminium pegs
- Yellow paint

1.6.4. Location and marking of sampling plots

The positions of the plots are given in table 1.6.4. The 1 m² terricolous lichen communities are all marked with four aluminium pegs, while the 20x20 cm saxicolous lichen communities are marked with yellow paint.

1.6.5. Description of sampling method

The degree of covering is estimated using the following, modified scale of Hult-Sernander: 5=1/2; 4=1/2-1/4; 3=1/4-1/8; 2=1/8-1/16; 1<1/16; 0.1= just present, 100= dominating. The number of lichen thalli are counted, and the thallus diameter for the saxicolous lichens are measured. Take a digital ortho-photo of all plots. The plot position marks need to be painted again every fifth year.

1.6.6. Description of laboratory work

Standard TLC or HPTLC methods are used for identification of selected specimens of sterile crustose lichens (Culberson & Kristinsson. 1970 and Arup et al. 1993.). The analyses are carried out at the Botanical Institute, University of Copenhagen.

1.6.7. Input of data into databases

Data from the plots are entered into an Excel data file: Lichen square plots with the columns: Plot no., Date (YYYY-MM-DD), Observer, Species, Cover (1 to 5), No. of thalli, Max diam., Remarks.

Digital pictures are downloaded and renamed to include Year and Plot number (e.g. 2008_P3).

Table 1.6.4. Positions and dimensions of lichen square plots

Plot	UTM E	UTM N	Size (cm)
P1	517,299	8,269,876	200x580
P2	514,549	8,266,377	20x20
P3	514,528	8,266,322	20x20
P4	514,579	8,266,455	20x20
P5	514,590	8,266,463	20x20
P7	510,800	8,264,226	20x20
P8	511,645	8,264,946	20x20
P10	515,154	8,265,106	20x20
P11	515,100	8,265,087	100x100
P12	515,095	8,265,032	100x100
P13	515,065	8,264,940	100x100
P14	513,348	8,264,587	100x100
P15	510,962	8,264,118	20x20
P16	510,344	8,263,912	20x20
P17	513,460	8,264,760	100x100
P18	513,842	8,264,772	100x100
P19	513,834	8,264,726	100x100

1.7. ITEX point frame plots

1.7.1. Species to be monitored

All vascular plants species. The nomenclature is according to Böcher et al. (1978). Species of mosses and lichens are referred to as moss or lichen. The ITEX frame plots are examined by an expert botanist.

1.7.2. Frequency of sampling

Every 5th year.

1.7.3. Equipment to be used

- Map of plot positions
- GPS
- An ITEX point frame (stored at the Zackenberg Research Station).

- Data sheets
- A mm ruler
- Knee pads
- Digital camera
- Notebook / dictaphone

1.7.4. Location and marking of sampling plots

The position of the plots appears from table 1.7.4. Each plot consists of five point frame analyses of 70x70 cm, each marked by an aluminium tube and three large spears.

Table 1.7.4. Location of the ITEX point frame plots.

Plot	UTM E	UTM N
1	513,354	8,264,996
2	513,610	8,264,719
3	513,215	8,264,608
4	512,999	8,264,260
5	515,041	8,266,857
6	510,668	8,264,268
7	514,066	8,266,466
8	512,623	8,263,608
9	515,037	8,266,857

1.7.5. Description of sampling method

The frame is placed horizontally above the plot fixed with the four legs in the tube and on the spear heads marking the plot. The distance from the soil surface to the underside of the frame should be checked; following Bay 1998. For each of the 100 hits per plot the vascular species are identified and recorded and mosses and lichens is given as moss or lichen. In addition the distance to the plant is measured (± 0.5 cm).

An ortho-photo is taken of each ITEX point frame plot prior to vegetation analysis.

1.7.6. Description of laboratory work

None.

1.7.7. Input of data into databases

Data from the plots are entered into an Excel data file named ITEX point frame and holding the columns Plot, Analysis, Date (YYYY-MM-DD), Observer, NS location (1-10), WE location (1-10), Layer, Species, Status, Height and Remarks.

Digital pictures are downloaded, renamed to include year and plot name (e.g. 2008_1), and stored in a separate folder named ITEX_point_frames.

1.8. Northern range species

1.8.1. Species to be monitored

- *Salix herbacea* (S. her.)
- *Campanula giesekiana* (C. gie.)
- *Carex glareosa* (C. gla.)
- *Carex lachenalii* (C. lac.)
- *Carex norvegica* (C. nor.)

The Northern range species are examined by an expert botanist.

1.8.2. Frequency of sampling

Every 5th year.

1.8.3. Equipment to be used

- Dictaphone / notebook
- Digital camera

1.8.4. Location and marking of sampling plots

The positions are given in table 1.8.4. The plots are marked by aluminium tubes in each corner.

1.8.5. Description of sampling method

The species are low arctic species with their known northern distribution limit within the study area or the neighbouring areas just north of. In each plot, the number of buds, open flowers, senescent flowers and fruits are counted.

An ortho-photo of the plot as well as additional photos are taken.

1.8.6. Description of laboratory work

None.

1.8.7. Description of laboratory work

Data from the plots are entered into an Excel data file named Northern range species and holding the columns Species, Date (YYYY-MM-DD), Observer, UTM E, UTM N, Buds, Flowers, Senescent and Photo numbers.

Digital pictures are downloaded, renamed to include Year, Plot number and photo number (e.g. 2008_S_her_1_pic1), and stored in a separate folder.

Table 1.8.4 Location and size of the 24 permanent plots for northern range species examination.

Plot no.	UTM E	UTM N	Size
S. her. 1	512,652	8,266,331	25x30 cm
S. her. 2	512,652	8,266,331	25x30 cm
S. her. 3	514,220	8,266,309	30x50 cm
S. her. 4	515,036	8,264,993	30x40 cm
C. gie. 1	515,444	8,264,398	0.75 m ²
C. gie. 2	515,582	8,264,411	0.75 m ²
C. gie. 3	515,582	8,264,411	1.0 m ²
C. gie. 4	515,582	8,264,411	0.5 m ²
C. gie. 5	515,582	8,264,411	1.0 m ²
C. gie. 6	515,395	8,265,473	40x60 cm
C. gie. 7	515,400	8,265,475	40x100 cm
C. gie. 8	514,255	8,266,202	40x50 cm
C. gie. 9	514,255	8,266,202	40x50 cm
C. gla. 1	512,716	8,263,667	35x30 cm
C. gla. 2	512,691	8,263,674	50x50 cm
C. gla. 3	512,691	8,263,674	30x40 cm
C. lac. 1	515,212	8,264,756	30x30 cm
C. lac. 2	514,223	8,266,285	1.0 m ²
C. lac. 3	514,793	8,267,180	0.8 m ²
C. nor. 1	515,384	8,265,018	15x20 cm
C. nor. 2	515,378	8,265,058	20x50 cm
C. nor. 3	515,283	8,265,398	20x50 cm
C. nor. 4	514,309	8,266,081	40x120 cm
C. nor. 5	514,309	8,266,081	120x120 cm

1.9. Normalised Difference Vegetation Index (NDVI) in plots

1.9.1. Species or taxonomic groups to be monitored

All plots mentioned in section 1.1 plus Arctic cotton-grass *Eriophorum scheuchzeri* (polar-kæruld) and *Eriophorum triste* (mørk kæruld) (Eri1-4), arthropod plots 3-7 (see tables 1.2.2 and 2.1.4).

1.9.2. Frequency of sampling

Once a week, on the same day or the day after the weekly flowering phenology and arthropod sampling. If the vegetation is wet, the measurements must be postponed to the following day.

1.9.3. Equipment to be used

- Map of plot positions
- GPS
- CropCircle Handheld system
- PDA / dictaphone / notebook

1.9.4. Location and marking of sampling plots

Plots are shown in fig. 1.1.4 and 2.1.4, and tables 1.2.2 and 2.1.4.

1.9.5. Sampling method

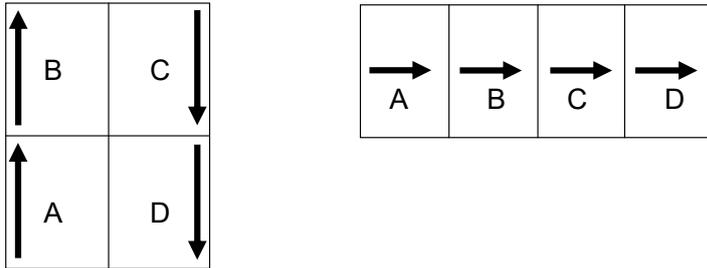
Insert an empty SD flash card into the card slot. Turn on the CropCircle system by pressing the ON/OFF button. Use the DOWN/UP arrows to select MAP mode. Then press OK. Press LOG and the CropCircle is ready to measure NDVI. NDVI measurements are conducted by scanning the vegetation. Scans are conducted by moving the sensor steadily forward (app. 1 meter per second) above the vegetation. The sensor **must** always be 75 cm above the ground when measuring. This results in a measuring footprint of app. 10 x 45 cm. Refer to the CropCircle manual for more information.

The following sampling order **must** be applied: SAX1/SIL1, SAX2/SIL2, SAL1, Dry1, Eri1, Sal2, Dry2/Sal7, Sal3/Cas1, Art3, Eri3, Pap1/Cas2, Eri4, Art7, Art5, Dry3, Sal4, Pap2/Sal5, Sal6, Sil4, Cas3, Dry4, Dry5, Eri2, Pap3/Cas4, Dry6/Pap4, Sax/Sil3. Also, always measure in the order A-D (see figure 1.9.5). At each visit, note under Remarks the presence of snow (snow in subsection; snow at plot edge) and if the vegetation is wet.

All measurements are conducted on the side of the plots (Fig. 1.9.5). Place yourself at the plot number plate, just outside the plot. Hold the sensor app. 50cm into the subsection at the subsection edge. Switch on the NDVI logger, and pause it immediately (using the metal switch on the downside of the stage), start logging (using same pause switch) and walk slowly (app. 1m per second) along the side of each subsection. 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 CropCircle system completely between plots by pressing the ON/OFF button.

In the arthropod plots, NDVI is measured just outside the plot to avoid measuring over the yellow pitfall traps (see figure 1.9.5). White sticks mark the location of the measuring area. One NDVI scan is made in each arthropod subsection (A-D) using the same procedure as for the vegetation plots. Turn off the CropCircle system between plots.

Vegetation plots



Arthropod plots

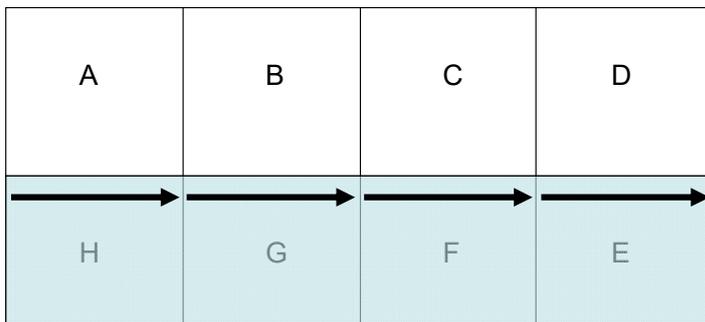


Fig. 1.9.5. NDVI scan lines in plant and arthropod plots. In the arthropod plots, the grey fill indicates the previously used pitfall squares that were closed in 2007.

1.9.6. Laboratory work

None.

1.9.7. Input of data into the database

Download data from the SD card using a card reader. CropCircle automatically names the files (e.g. DDMMYYAA.csv; DDMMYYBB.csv; etc. and in some cases AA.csv), and each file holds the following variables: : Longitude, Latitude, Elevation. Fix Type, UTC Time, Speed, Course, SF1, SF2, SF3, SF4, SF5, SF6. Save all CropCircle data files separately. In Excel, each data file is supplemented with the following columns: Date (YYYY-MM-DD), Observer, and Remarks. Rename the Plot values into the specific plot name (e.g. DRY1, following the specific order mentioned above), and Sample values into the specific section name (A, B, C or D). Merge all Excel files into one.

1.10. Normalised Difference Vegetation Index transects

1.10.1. Species or taxonomic groups to be monitored

Selected vegetation types along the ZERO line and in the valley lowland.

1.10.2. Frequency of sampling

Minimum four times during the season (5 June, 30 June, 25 July and 18 August, ...). Ideally, all transects are walked on the same day. If the vegetation is wet, the measurements must be postponed to the following day.

1.10.3. Equipment to be used

- Map of the transect routes
- Handheld GPS with transect waypoints
- CropCircle Handheld system
- Extra 12V battery
- PDA / dictaphone / notebook
- Digital camera

1.10.4. Location and marking of sampling plots

The three NDVI transects are shown in fig. 1.11.4. The first transect starts at the old delta and follows the ZERO line to app. 440m a.s.l. (ZERO Line mark 101). The second transect starts just north of Lomsø, covers the valley lowland and ends at the GeoBasis grid ZEROCALM_1 (110x110m), which is covered in the last transect. Transect UTM coordinates are given in table 1.10.4.

Table 1.10.4. Transect UTM coordinates for the three NDVI transects.

Transect	Way point	UTM E	UTM N
ZERO Line	SNZ1	512,730	8,263,610
	SNZ66	514,341	8,266,178
	SNZ91	514,927	8,266,903
	SNZ99	515,841	8,268,084
	SNZ101	516,037	8,268,342
Valley lowland	SNMBIO	513,600	8,264,050
	SNM3	513,471	8,264,686
	SNM4	513,538	8,266,089
	SNM5	513,641	8,267,092
	SNM6	513,193	8,265,685
	SNM7	513,365	8,264,855
ZEROCALM_1	SNM7	513,365	8,264,855

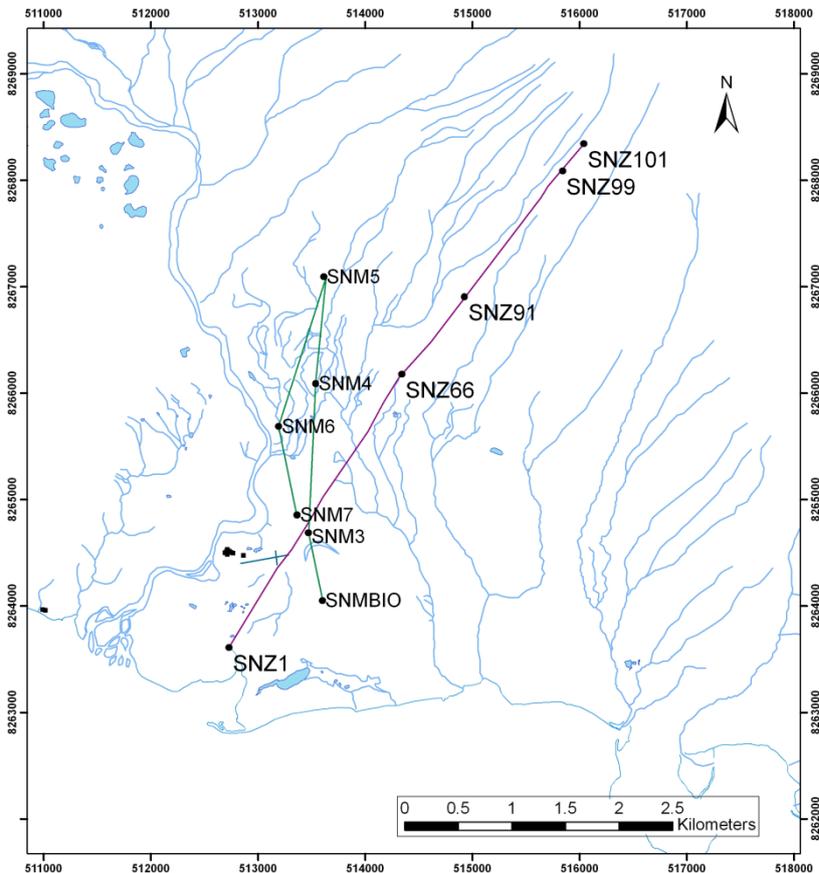


Fig. 1.10.4. Two of the three NDVI transect routes. The third, ZEROCALM_1, is located at point SNM7.

1.10.5. Sampling method

Basically, the same procedure as in section 1.9. Switch on the NDVI logger, and walk in a steady pace along the transect (at the ZERO Line app. 2-3 meter southeast of the aluminium markers), while holding the sensor to your right side and 75 cm above the ground. NDVI is logged continuously. In ZEROCALM_1, the grid is marked with small, white stones every 10m. The NDVI within the grid is measured continuously along transect routes marked by red and white stones (red stones mark the edges). Start in North-western corner (marked by red and white pole) and walk over the stones towards the North-eastern corner (marked by red and white pole) while measuring NDVI to your right. Here, turn 90 degrees south and walk towards the next transect line. Turn 90 degrees right and walk along the transect towards the west while measuring NDVI to your right (Fig. 1.10.5). Switch off the NDVI logger while having breaks. Switch off the CropCircle system now and then, and at the end of each transect, to save the CropCircle data in a file.

It is necessary to mark the transects with sticks to ensure that the transects are walked properly.

Take digital photos now and then to assist the interpretation of the NDVI measurements.

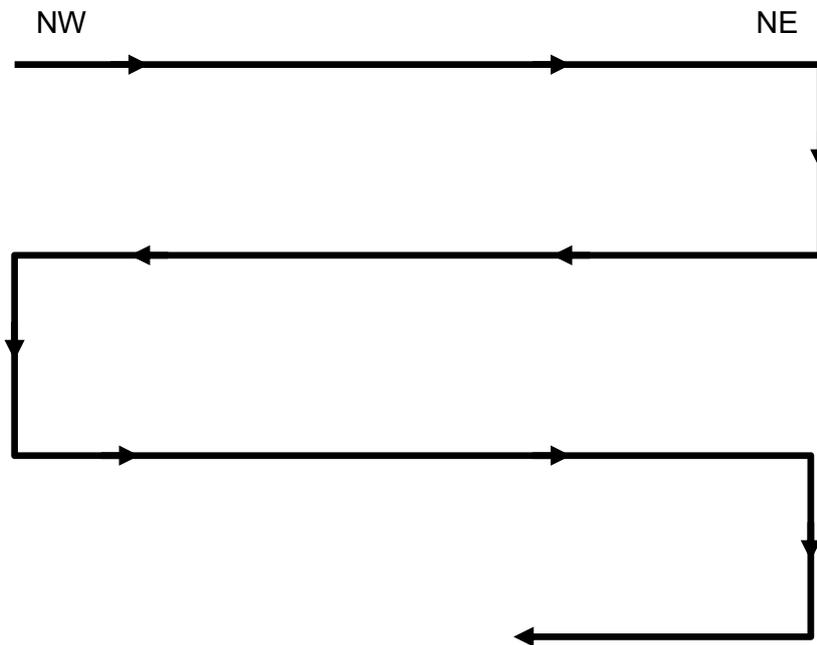


Fig.1.10.5. NDVI measuring route in the ZEROCALM_1 grid.

1.10.6. Laboratory work

None.

1.10.7. Input of data into the database

Download data from the SD card using a card reader. CropCircle automatically names the files (e.g. 220607AA.csv; 220607AB.csv). Each file holds the following variables: Longitude, Latitude, Elevation, Fix Type, UTC Time, Speed, Course, SF1, SF2, SF3, SF4, SF5, SF6. Save all CropCircle data files separately. Before editing, make all data files write protected. In Excel, each data file is supplemented with the following columns: Date (YYYY-MM-DD), Observer, Transect (ZERO_line, Valley_lowland, ZEROCALM_1), and Remarks. Merge all Excel files into one.

Digital pictures are downloaded and renamed to include transect name, date and picture number (e.g. Lowland_080605_pic1).

1.11. Normalised Difference Vegetation Index from remote sensing

Annual estimates of NDVI (around 1 August) inferred from satellite images and digital cameras mounted at Nansenblokken. Satellite images and digital camera images are analysed by GeoBasis or BioBasis. The 14 sections in which NDVI (and snow cover) is estimated are shown in fig. 1.11. See also special reports (e.g. Bøcker, C.A. 1999: Detaljeret analyse af NDVI for udvalgte vegetationszoner, Zackenberg 1998. – Asiaq).

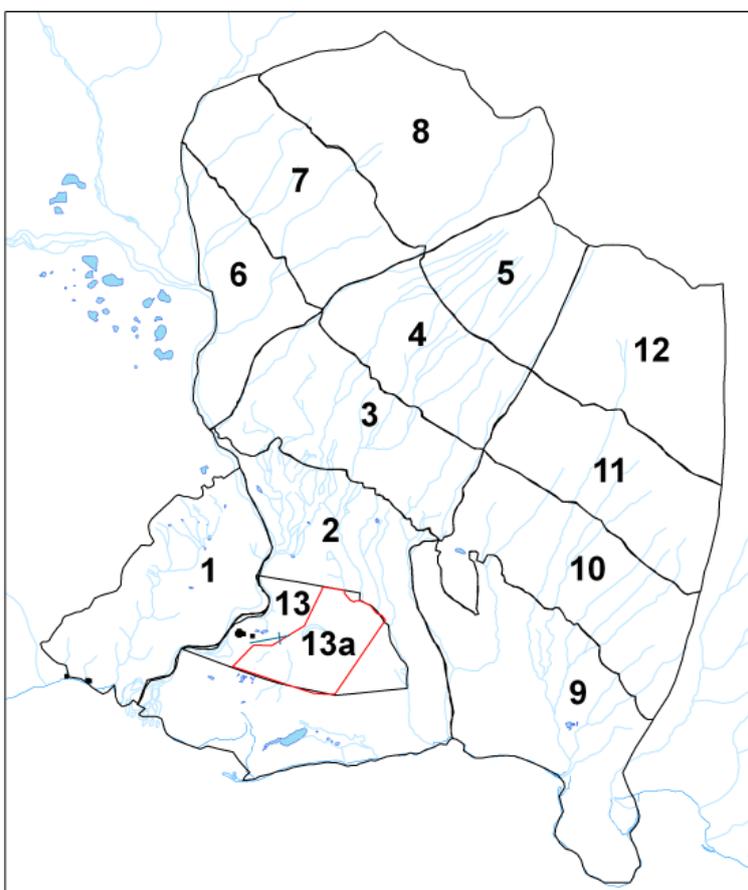


Fig 1.11. Map of Zackenbergdalen with the 14 snow- and NDVI-zones demarcated. Zone 13 is the old lemming census area, while 13a denotes the new, reduced lemming census area.

1.12. Soil moisture at permanent plots

1.12.1. Species or taxonomic groups to be monitored

Soil moisture at permanent monitoring plots in the valley lowland.

1.12.2. Frequency of sampling

Weekly.

1.12.3. Equipment to be used

A PR2 profile probe and an HH2 moisture meter readout unit set at Soil type =mineral soil, Field capacity=0.38, Display=% Vol. PDA for entering data in the corresponding Arcpad shape file (SoilMoistObs.shp).

1.12.4. Location and marking of sampling plots

Soil moisture tubes for the probe are installed at the permanent plant plots: Sal1, Dry1, Dry2Sal7, Sal3Cas1, Cas3, Dry5.

1.12.5. Sampling method

Remove the lid of the soil moisture tube. Lower the probe into the tube. Align the one of the screws on the probe handle with the black vertical line on the top of the tube. Press [Esc] on the HH2, then press [Read]. Shuffle through the measurement at 100mm, 200mm, 300mm and 400mm by using [UP] or [DOWN]. Enter the values for the different heights in the A column of the table in Arcpad. Then turn the probe 120 degrees to align the next screw on the probe handle with the vertical black line on the tube. Press [Esc], then [Read] and repeat the whole procedure for the B readings. Finally turn the probe a last 120 degrees to take the C readings. Remember to put the lid back on the tube as it may otherwise fill with snow or rain. If snow or rain has entered a tube there is a possibility it will freeze, unabling the probe to enter the tube. Ice in the tube can be carefully melted with warm water, making sure not to spill any on the outside of the tube. The melted snow or ice can then be sucked up with a flexible pvc tube.

1.12.6. Laboratory work

None.

1.12.7. Input of data into the database

Data files recorded in Arcpad and holding the columns OBSDATE, OBSERVER, PLOT, SNOWA, SNOWB, SNOWC, SNOWD, REMARKS, X, Y are read into the database.

1.13. Vegetation composition in ITEX and UV plots

1.13.1. Species or taxonomic groups to be monitored

Vegetation in ITEX and UV plots. The ITEX and UV plots are examined by an expert botanist.

1.13.2. Frequency of sampling

When possible in late July.

1.13.3. Equipment to be used

- Pin point counting frame (located in House 4)
- Digital camera
- Dictaphone / notebook
- Flora

1.13.4. Location and marking of sampling plots

See tables 2.12.4.1 and 2.12.4.2.

Table 2.12.4.1 Location of the ITEX plots

Site	Plot	Waypoint	UTM E	UTM N
Salix snowbed	NY-K-1-S	K-1-S	513,727	8,265,094
	NY-W-1-S	W-1-S	513,728	8,265,090
	NY-K-2-S	K-2-S	513,731	8,265,090
	NY-W-2-S	W-2-S	513,732	8,265,093
	NY-K-3-S	K-3-S	513,736	8,265,089
	NY-W-3-S	W-3-S	513,737	8,265,086
	NY-K-4-S	K-4-S	513,730	8,265,083
	NY-W-4-S	W-4-S	513,730	8,265,086
	NY-K-5-S	K-5-S	513,728	8,265,086
	NY-W-5-S	W-5-S	513,725	8,265,091
Cassiope heath	NY-K-1-C	K-1-C	513,687	8,265,106
	NY-W-1-C	W-1-C	513,690	8,265,109
	NY-K-2-C	K-2-C	513,683	8,265,107
	NY-W-2-C	W-2-C	513,681	8,265,110
	NY-K-3-C	K-3-C	513,661	8,265,104
	NY-W-3-C	W-3-C	513,663	8,265,101
	NY-K-4-C	K-4-C	513,670	8,265,101
	NY-W-4-C	W-4-C	513,676	8,265,099
	NY-K-5-C	K-5-C	513,664	8,265,096
	NY-W-5-C	W-5-C	513,658	8,265,096

Table 2.12.4.2 Location of the UV plots.

Plot	Waypoint	UTM E	UTM N
Control_1	CTRL1	513,717	8,265,100
Control_2	CTRL2	513,721	8,265,099
Control_3	CTRL3	513,724	8,265,107
Control_4	CTRL4	513,728	8,265,099
Control_5	CTRL5	513,728	8,265,103
Filter_1	F-1	513,716	8,265,097
Filter_2	F-2	513,718	8,265,102
Filter_3	F-3	513,721	8,265,105
Filter_4	F-4	513,724	8,265,099
Filter_5	F-5	513,727	8,265,105
UVB_1	UV-B1	513,715	8,265,100
UVB_2	UV-B2	513,719	8,265,099
UVB_3	UV-B3	513,723	8,265,104
UVB_4	UV-B4	513,726	8,265,097
UVB_5	UV-B5	513,726	8,265,102

1.13.5. Sampling method

Take a digital ortho-photo of each plot. Carefully place the pin point frame in the permanent markers in each plot. At each of the 100 intersections in the frame, a pin is moved vertically down. At each intersection, all plant species that are hit as the pin is moved towards the ground must be recorded, resulting in more than 100 recordings. All recordings of a given species in a given plot are lumped together. Take off in the record scheme from last year, and add new species when needed. The analyses are most efficiently conducted when the observer is assisted by a writer.

1.13.6. Laboratory work

None.

1.13.7. Input of data into the database

All data are entered into the excel file named Pin_point_YEAR_Itex_UV. Digital photos are downloaded, renamed to include Year and plot name (e.g. 2009_K1C), and saved in a separate folder.

1.14. Vegetation composition in alpine environments (GLORIA)

1.14.1. Species or taxonomic groups to be monitored

Vegetation composition on three summits in the valley. The GLORIA plots are examined by expert botanists.

1.14.2. Frequency of sampling

Every 5th year.

1.14.3. Equipment to be used

- Please refer to the detailed GLORIA manual available at www.gloria.ac.at.

1.14.4. Location and marking of sampling plots

Three GLORIA sites are found at Zackenberg: Kamelen (KAM, 90m a.s.l.), a moraine hill in the valley lowland, "Polemoniumbjerg" (POL, 470m a.s.l.), and "Little Aucellabjerg" (AUC, 605m a.s.l.) both located further up the slope of Aucellabjerg (table 1.15.4).

Table 1.15.4. The location of the three GLORIA summits at Zackenberg.

Plot	UTM E	UTM N
KAM	513,927	8,266,670
POL	515,621	8,269,150
AUC	516,580	8,268,930

1.14.5. Sampling method

Please refer to the detailed GLORIA manual available at www.gloria.ac.at.

1.14.6. Laboratory work

None.

1.14.7. Input of data into the database

Please refer to the detailed GLORIA manual available at www.gloria.ac.at.

2. Arthropods

2.1. Yellow pitfall traps

2.1.1. Species to be monitored

All taxonomic groups of arthropods.

2.1.2. Frequency of sampling

The traps are emptied weekly on fixed dates (see table 1.1.2). If bad weather prohibits proper handling of the samples, the traps may be emptied on the following day. Trap stations are open from snowmelt until 1 October (or when frozen).

2.1.3. Equipment to be used

- PDA / notebook
- Map showing position of study plots
- GPS
- 50 yellow (Pantone no. 108U) plastic cups, 10 cm in diameter and 8 cm deep
- A thermos
- A garden trowel with sharp edge
- 1, 5 and 10 l containers for water
- Odour-free detergent (Tween 20 from VWR / Bie & Berntsen, +45 4386 8788)
- Salt (NaCl) – not from the kitchen!
- 50 metal pegs
- Knee pads
- A small aquarium net with the outer 10 cm of a lady's stocking as bag (make a new one each year and clean it in fresh water after each sampling day)
- A pair of pointed and angled tweezers
- 350 ex. 10 cl plastic containers with lids
- 30 l of 96% alcohol
- An ear syringe (with a rubber bulb and tube)
- Alcohol resistant labels
- Alcohol resistant pens
- Alcohol resistant speed marker

2.1.4. Location and marking of sampling plots

The position of the study plots appear from fig. 2.1.4 (nos. 2-7) and table 2.1.4. Each plot measures 5 x 20 m² and is made up of four 5 x 5 m² squares marked with a white nylon stick (15 in total; see below) in each corner. Each plot is identified with a ID sign, and each section (with one trap each) is denoted A-D straight from the ID sign. E-H is the second row of sections going back towards the end with the ID sign.

On Station 2, the traps are marked with a nylon stick at each trap. Each stick is further marked with metal bands around the top (A-H):

- One band in the top: A
- Two bands in the top: B

- Three bands in the top: C
- Four bands in the top: D
- One band lower down: E
- Two bands lower down: F
- Three bands lower down: G
- Four bands lower down: H

Pitfalls E-H were closed in 2007, but sticks marking the four pitfalls are still present at Station 2.

Table 2.1.4. UTM-coordinates of the seven arthropod sampling plots.

Plot no.	UTM E	UTM N	A corner
1	512,966	8,264,532	-
2	513,083	8,264,591	-
3	513,320	8,264,844	SE
4	513,215	8,264,941	E
5	513,825	8,265,055	S
6	513,545	8,264,747	W
7	513,780	8,265,100	W

The vegetation types in pitfall plots are

- Art 2: Wet fen dominated by mosses and grasses/sedges (incl. Arctic cotton grass *Eriophorum scheuchzeri*), and with Arctic willow *Salix arctica* on the turfs.
- Art 3: Mesic heath dominated by lichens (an almost complete cover of organic crust) and white Arctic bell-heather *Cassiope tetragona*, and with scattered individuals of Arctic willow and Arctic blueberry *Vaccinium uliginosum*.
- Art 4: Mesic heath dominated by lichens (an almost complete cover of organic crust) and with scattered individuals of white Arctic bell-heather, Arctic willow, mountain avens *Dryas* sp., grasses/sedges and Arctic blueberry. This plot was closed in 2017.
- Art 5: Arid heath dominated by lichens (an almost complete cover of organic crust) and mountain avens and with scattered individuals of Arctic willow and Ballard's kobresia *Kobresia myosuroides*.
- Art 6: Snow-bed covered in a thick mat of lichens (an almost complete cover of organic crust) and poorly performing Arctic willow. The plot was closed in 1999, but reopened in 2016.
- Art 7: Highly exposed and arid heath dominated by lichens (an almost complete cover of organic crust) and mountain avens, and with scattered individuals of Arctic willow and Ballard's kobresia (more mountain avens and less Arctic willow than Art 5, which often is snow-covered in winter).

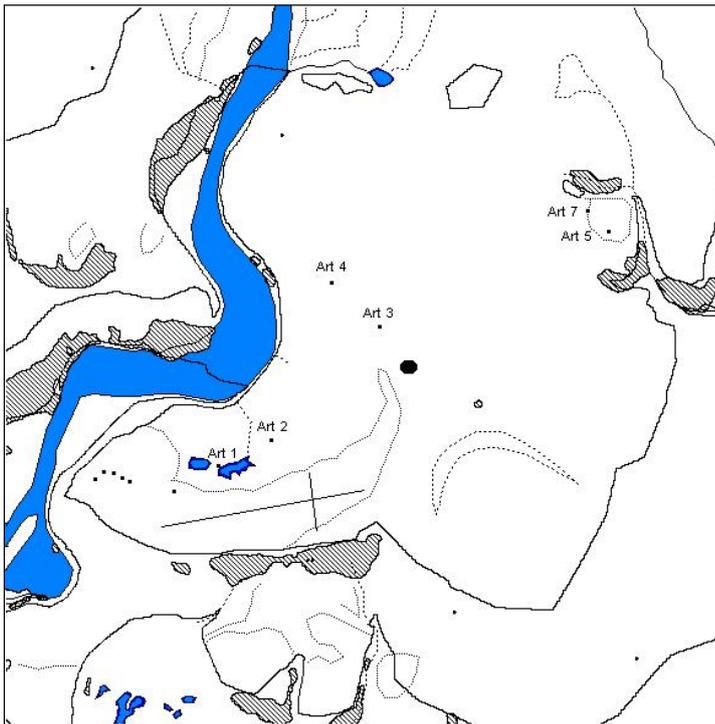


Fig. 2.1.4 Position of arthropod sampling 1-7 (1 = window traps, 2-7 = pitfall trap station) and the climate station (dot).

2.1.5. Sampling method

Starting the season

The pitfall traps are covered with a plastic lid and a stone during the winter (see below). At the start of the season (i.e. when the traps have appeared from the snow), new clean (washed with a little Tween 20) upper cups replace the 'overwintering' 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.

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.

The traps on Station 2 are positioned on the only 'elevated' mounds on the site that are not flooded during spring. The traps on this station are only made up of one cup, as they otherwise would float during the snowmelt. Still, they may need pegs to keep them in position.

If needed due to wear on the vegetation etc., new traps for the following season are established late each summer, when a set of four 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 turning your

back to the square and throwing an item over your shoulder. The trap is then dug down on the nearest reasonably level and 'elevated' site (so that it is not flooded during the snow melt) and carefully sunk into the soil, so that the upper rim levels exactly with the soil surface. Place the turf and the removed soil in the old hole.

Emptying the traps

Catches from each of the four traps are kept separate. Prepare 21 ex. 10 cl containers in advance by filling them app. half with 96% alcohol. Alcohol resistant labels (see figure 2.1.5) with date, plot numbers and sections (A-D) written with an alcohol resistant pen are prepared from home and kept in order by a paper clip. For ART1, the A sample denotes the window trap facing N-S, and B the trap facing E-W. Do not write on the containers.

NE GREENLAND, Zackenberg,
 74°28'N, 20°34'W
 Station: Date:
 Aarhus University, Dept. of Bioscience
 Museum of Natural History, Aarhus

Fig. 2.1.5. Text on the pre-printed labels.

When traps are emptied, the trap liquid is poured through the aquarium net into a spare cup, whereupon the liquid is poured back into the repositioned upper cup. Check the cup carefully for small arthropods before repositioning it. Mites, especially, often remain in the cups. Take care that liquid from the net does not fall on the soil around the trap by keeping the net over the cups all the time. The catch is then emptied into the 10 cl. container with alcohol by turning the net inside out in the container. All remaining invertebrates must be removed carefully from the aquarium net by the tweezers and put into the container.

Note the full hour of the day, when the traps in each plot are emptied. After emptying all traps, extra water must 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 or musk ox. 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 and new water must be established. Never touch the traps with mosquito repellent or suntan oil on your fingers!

At all visits at the arthropod stations during snow melt, the snow cover (%) is estimated for each section of the plot (see 1.1.5). Visit the arthropod stations every 2-3 days to estimate snow cover. At station 2, this only apply to the individual traps (i.e. the trap covered = 100%, the trap snow-free = 0%).

Finally, four NDVI measurements are taken in Arthropod 3-7 (see section 1.9).

Ending the season

At the termination of the catching season on 26 August or 3 September, the trap liquid must be collected from all the traps and poured into Zackenbergelven. Cover all traps with a plastic lid and a stone during the winter.

Arthropod samples are sent to Aarhus University. Place all containers from traps A and B in one zarges box, and all containers from traps C and D in another zarges box.

2.1.6. Laboratory work

Specimens are sorted to different taxonomic levels by expert entomologists at the Department of Bioscience, University of Aarhus.

Specimens from each taxa from each trap and date are kept in separate glass tubes with 70% alcohol. A pre-printed label written with an alcohol resistant pen stating date and year of collection, station and section no., and taxa (scientific name only) must be located inside the tube. Glass tubes with specimens are stored at the Museum of National History in Aarhus. The collection is organised in accordance with the instructions from the curator at the museum.

2.1.7. Input of data into database

After each round and emptying of the traps, the following data are entered into the Excel data sheets named Art2-7: Date (YYYY-MM-DD), Hour, Plot, Fieldworker, Snow A (percent in the section), Snow B, Snow C, Snow D; Days A (trap days since the last emptying for the trap in the sector), Days B, Days C, Days D, Taxon, and Remarks. Under Remarks, date of opening and closing together with relevant observations on the traps are stated. This include any disturbance that may influence the efficiency of the traps such as flooding, drying out, ice, dirt, faeces and vandalism by foxes.

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

2.2. Window traps

2.2.1. Taxonomic groups to be monitored

Same as in section 2.1.

2.2.2. Frequency of sampling

Same as in section 2.1.

2.2.3. Equipment to be used

- PDA / notebook
- Two window traps each with a 'window' of 20x20 cm²
- A cloth
- A bucket

- Otherwise same as 2.1.3

2.2.4. Location and marking of sampling plot

On an islet in the eastern pond of Gadekæret (Art 1 on fig. 2.1.4), two angular aluminium bars make up the holders for each trap. The traps are positioned with the windows in a right angle to each other, so that they catch in 'all' wind directions (i.e. one trap facing N-S, and one facing E-W).

2.2.5. Sampling method

The two 'basins' of each trap must be filled 3/4 with water, detergent (one spoonful in each of the four basins) and salt (12 tablespoons in each of the four basins). At each visit during ice melt in spring the ice cover (percent) on the pond must be estimated.

When emptying the traps, the aquarium net is used to 'fish' the catch from each basin. The catch from each of the two basins from one trap are emptied into a 10 cl. container. Refill the traps at each visit (5 l of water needed), and add a little salt in the middle of the season to compensate for loss. Keep the windows absolutely clean from salt water and salt.

Empty them together with the other traps by the end of the season. Use a bucket when you empty them. The traps must be stored in-house during winter. Arthropod samples are shipped to Aarhus University (see above regarding shipment of containers).

2.2.6. Laboratory work

As for the pitfall traps (section 2.1.6).

2.2.7. Input of data into database

All relevant data are entered into the Excel data file named Art1 and holding the following columns: Date (YYYY-MM-DD), Hour, Plot, Fieldwork, Sorting, Ice (percent cover on surrounding pond), Days NS, EW (No. of trap days per chamber since last emptying. Note that since each trap consists of two chambers (e.g. N-S), one week of trapping equals 14 days), Taxon, No. NS, No. EW, and Remarks. Under Remarks, date of opening and closing together with relevant observations on the traps are stated. This includes any disturbance that may influence the efficiency of the traps such as ice, dirt and vandalism by musk oxen. After sorting, the total number of individuals per group is entered into the data sheets under Taxon and No. (no. of individuals in each trap).

2.3. Predation by larvae of *Sympistis zetterstedtii* on *Dryas* flowers

See section 1.1.5: *Dryas*.

3. Birds

3.1. Breeding bird census

3.1.1. Species to be monitored

All bird species breeding in the census area including

- Red-throated diver *Gavia stellata* (rødstrubet lom)
- King eider *Somateria spectabilis* (kongeederfugl)
- Long-tailed duck *Clangula hyemalis* (havlit)
- Rock ptarmigan *Lagopus muta* (fjeldrype)
- Common ringed plover *Charadrius hiaticula* (stor præstekrave)
- Red knot *Calidris canutus* (islandsk ryle)
- Sanderling *Calidris alba* (sandløber)
- Dunlin *Calidris alpina* (almindelig ryle)
- Ruddy turnstone *Arenaria interpres* (stenvender)
- Red-necked phalarope *Phalaropus lobatus* (Odinshane)
- Long-tailed skua *Stercorarius longicaudus* (lille kjøve)
- Northern wheatear *Oenanthe oenanthe* (stenpikker)
- Snow bunting *Plectrophenax nivalis* (snespurv)

3.1.2. Frequency of sampling

Annually during June and July.

3.1.3. Equipment to be used

- Binoculars
- Notebook
- PDA with GPS
- If working without PDA: Clip-board mounted topographic maps of census area sections
- Altimeter (when using printed maps rather than PDA with GPS)
- Skis
- Snowshoes

3.1.4. Location and marking of sampling plots

The borders of the 15.8 km² census area in Zackenbergdalen is marked on the field maps (fig. 3.1.4), and they generally follow visible features in the landscape.

Keep a careful eye out for nesting divers on the ponds and lakes in the census area. Incubating birds either stretch out on the nest, when a person approaches, or they leave it and stay on the water, where they may even hide along the shore, so check the entire shore and turfs carefully.

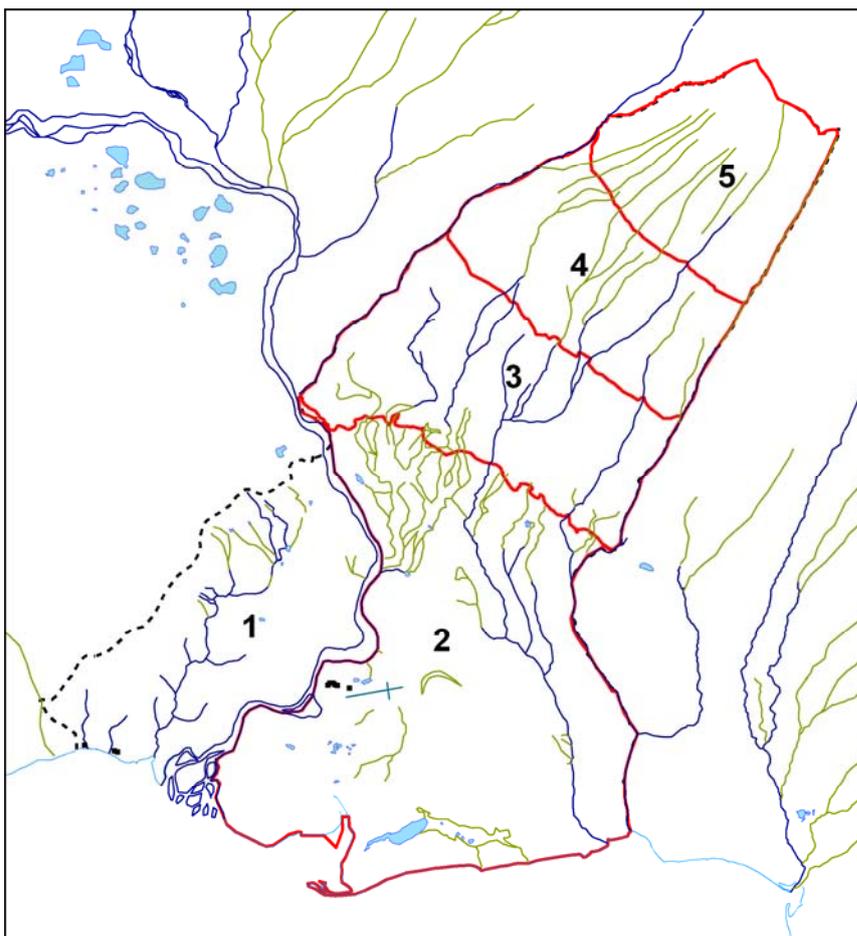


Fig. 3.1.4. Map of the breeding bird census area in Zackenbergdalen. Zone 1 west of the river is no longer censused.

3.1.5. Sampling method

The census area is surveyed during mid-late June. At that time, territories have been established, egg-laying has been initiated, and the birds are concentrated on the relatively limited areas of snow-free ground.

During the survey trips, the area must be covered in such a way that no snow-free spot is passed by the observer at a distance exceeding 100 m. Minor snow free spots may be searched carefully with the binoculars at a little longer distance. The surveys must be performed in fine weather. Avoid windy days and precipitation and start with the most important areas below 300 m a.s.l. In periods of prolonged overcast, pay close attention to the behaviour of the birds. After a few days of quiet, "normal, sunny-day behaviour" resumes; allowing to continue censusing as a usual "good condition" day.

On each survey trip, all bird observations are marked in GIS on the PDA (in case of equipment failure, use maps and specific symbols for each species and type of behaviour (see table 3.1.5)).

On the slopes of Aucellabjerg, it may be somewhat difficult to plot the observations on the right positions on the map/PDA. Should the GPS be malfunctioning or unable to get connection to satellites, the combination of rivulets,

snow drifts and the altitude provided by an altimeter gives the best clues. Remember to adjust the altimeter before leaving the station (35 m).

Until mid/late June or early July (varying from year to year), it is advantageous to use skis in most of the lowland, and snowshoes may be used to pass large snow drifts. During mid June, Rylekærene may be difficult to pass due to extensive water-soaked snow. The watershed south of Kamelen, along the western side of Kærelv, is then the best place to pass.

In GIS each observation is marked with species, sex and behaviour etc.

In case of equipment failure, use paper maps:

Observations of single individuals are plotted as open symbols, while pairs are filled. Nests are shown as a pair/individual (dependent on the presence of the parents) with a circle around, while broods are marked as a pair/individual with a dashed circle around. The number of eggs/young is stated at the symbol).

Breeding behaviour is given a symbol as follows

- s** Singing individual (or individual in a pair)
- ag** Aggressive individual/pair (towards con-specifics, other species or predators)
- v** Vocalisation other than song (e.g. alarm calling individual/pair)
- y** Distraction behaviour or other display that clearly indicate breeding (incl. nest building and food collecting in passerines)
- a** Alert (indicating nest or young)
- Purs** Flight pursuit between two or more birds

Two records of the same individual(s) on different positions are connected with a full line, while uncertain double-records are connected with a dashed line with a question mark on.

Be aware that it is already in the field that you decide, whether a record represents 'a pair/territory' or an uncertain 'pair/territory' by means of records of pairs, song or other vocalisation (see section 3.1.6).

Remember to estimate the ice cover on Langemandssø and Sommerfuglesø at each visit on the upper slopes of Aucellabjerg (see section 7.1).

3.1.6. Laboratory work

After each season the results of the field records are evaluated and concluded by a map of all territories found in the census area. It is recommended to do this as soon as possible, while the census is still fresh in memory! Ideally, it should be done at the end of or the day after the final leg of the census routes has been surveyed. All site claiming pairs/individuals are considered full members of the population, whether breeding or not. Hence, all records of pairs, singing (or otherwise territorial) and vocal individuals are considered as 'territories'. Records that does not fulfil these criteria, but still may indicate the presence of a territory (e.g. stationary single but silent individuals), are plotted as additional territories with a question mark. The same is done with possible double registrations.

For snow buntings (and other passerines), only pairs and males are considered representative of pairs/territories. Pairs and singing males are taken for 'territories', while non-singing males are taken as possible territories.

The initial census in mid-late June is considered the basis for the evaluation, and an initial evaluation is made after this survey. In case of equipment failure, two records of the same individual(s) on different positions are connected with a full line, while uncertain double-records are connected with a dashed line with a question mark on. Pairs may be added to these results if they are found in areas where no pairs were recorded nearby or in areas that were poorly covered during the initial census, or territories may be upgraded from a question mark if better records at the site are obtained later. Otherwise, later records of nests and small young together with stationary and clearly breeding pairs are just counterbalanced by repositioning a nearby 'territory' from the initial census. For the red knot, even pairs and singing individuals recorded during early June are included in the evaluation. Great care should be taken not to double record pairs and individuals that move from one area to another following snow melt. Territories, in which nests or broods have been found, are specifically marked (see section 3.1.5).

Table 3.1.5. Symbols for marking of bird observations on field maps.

Code	Species
	King eider <i>Somateria spectabilis</i>
	Long-tailed duck <i>Clangula hyemalis</i>
	Rock ptarmigan <i>Lagopus mutus</i>
	Common ringed plover <i>Charadrius hiaticula</i>
	Red knot <i>Calidris canutus</i>
	Sanderling <i>Calidris alba</i>
	Dunlin <i>Calidris alpina</i>
	Ruddy turnstone <i>Arenaria interpres</i>
	Red-necked phalarope <i>Phalaropus lobatus</i>
	Long-tailed skua <i>Stercorarius longicaudus</i>
	Snow bunting <i>Plectrophenax nivali</i>

Other species are written in full.

3.1.7. Input of data into database

The position of each territory is entered into an Excel data file named 'Bird territories' and holding the following columns: Species, Year, Observer, Serial no., UTM East, UTM North, Status, and Remarks.

Territories in which nests or young have been found, must be allocated the same serial number as in the bird nest and bird brood data files, respectively (see section 3.2.7). Such territories are given status as N for nest found or B for brood found. Other territories are given status T for territory or "?" for uncertain territory.

For territories in which nests have been found, the UTM co-ordinates must be the same as in the bird nest database. For territories with broods this only apply to brood finding places that are considered representative for the actual territory (and hence, the UTM co-ordinates of the finding place of the brood is given in both databases). Others are given separate UTM co-ordinates for territory and finding place, and the B in status is given in brackets.

Under remarks, you may note e.g. that this brood may originate from nest no. x, etc.

3.2. Bird breeding phenology

3.2.1. Species to be monitored

Sanderling, dunlin, ruddy turnstone and long-tailed skua, each of which data from between 10 and 15 nests/broods are collected. Remaining species from section 3.1 can be included if possible.

3.2.2. Frequency of sampling

Annually during June, July and August.

3.2.3. Equipment to be used

- White nylon sticks
- A thermos
- A 0.5 l vessel with a mm scale and protractor on the side
- Paper towels
- Electronic scales or 30, 100 and 500 g Pesola spring scales
- Fabric bags ("bird bags")
- Bird rings from the Zoological Museum, University of Copenhagen
- Ringing pliers
- Notebook and PDA with GPS
- Otherwise same as 3.1.3

3.2.4. Marking of sampling plots

Each nest found must be marked with a white nylon stick or a small stone cairn situated 10 metres from the nest in the direction away from the tall antenna of the Zackenberg station.

3.2.5. Sampling method

During the second half of June and the first half of July wader nests are spotted by keeping an eye on birds lifting from nests and by 'walking back' from intensively alarm calling or distraction displaying individuals. From a point with good overview, the bird is watched moving back to the nest. If both individuals of a pair are alarm calling, keep track of the least vocal bird usually located in the periphery of alarming individuals. It may also be productive just to sit down and search an area with your binoculars. Learning/reading up on the behaviour of the adults around the nest, is useful – at least for target species. Nests of other species are located opportunistically.

Eggs of waders and skuas are checked for incubation stage by floating two eggs of the clutch in the 0.5 l vessel 2/3 filled with approximately 25°C water from the thermos (cf. Hansen et al. 2011). The angle between the egg axis and the bottom of the vessel or the extent of the egg that is above water and the angle towards the water

surface is recorded. Use your watch to estimate angles. The eggs must be carefully dried with the paper towels before repositioned in the nest.

Do not visit nests again until about three days before the estimated date of hatching, as tracks to and from nests may guide predators to the nest. From then on, eggs are checked for cracks in the shell at each visit, i.e. whether they are 'starred' or 'pipped'. An egg is recorded as 'starred' when there are small hatching cracks in the shell, and as 'pipped' when there is a hole in the shell. An egg is starred 2-4 days prior to hatching and pipped within 1-2 days before hatching.

Nests found empty must be checked for signs of hatching (shell fragments in the bottom nest material) or predation (predated shells, smell of fox, faeces). Nest cups, from which pulli have left, are often broad and flat as compared to depredated nests. Cold unhatched eggs are collected and kept at the station in alcohol and refrigerated/frozen, and after the season, will be kept at Dept. of Bioscience. All wader and skua chicks found must be weighed to enable ageing. Furthermore, in skua young the wing is measured flattened and straightened on a ruler. Newly hatched wader chicks are weighed after they have been ringed (if weighed by spring scales, by 'hanging' them in the ring from the spring balance. Larger chicks are weighed in a fabric bag (remember to weigh the bag immediately afterwards and subtract the weight from the weight of the young). Keep all young of a brood in a fabric bag until they can be released simultaneously. Skua chicks cannot be ringed until 3-4 days old. Because the bird work demands handling of eggs and chicks, mosquito repellent is not allowed to be used by bird workers, and sun lotion on your hands should be avoided.

The goal is to find a combined total of 10-15 nests and broods in each of the species sanderling, dunlin and ruddy turnstone, besides as many of the long-tailed skua nests as possible.

Chicks that are only observed from the distance are 'aged' by comparing them with the state of development of chicks experienced at previous occasions. Newly fledged wader and skua young, still with downy head and neck and still accompanied by an adult, are recorded too. These can only be aged (cf. table 3.2.6.2) if no periods of inclement weather have prevailed during the possible fledging period!

Nests of divers, ducks and ptarmigan are checked from a distance to record hatching date, brood size and possible predation. Do not flush incubating waterfowl to check the nest. If incubating ducks are accidentally flushed from the nest, the eggs are counted, whereupon they are covered with the nest down. Observations of young of these species are similarly recorded.

Breeding phenology of passerines is monitored by records of calling young and observations of fledged juveniles.

Long-tailed skua young are followed until fledging or the adults leave the area, so that the young must be considered lost. If active skua families are still present when the bird observer leaves Zackenberg, a map marking the last position of these families and the expected date of fledging of the young must be provided to the remaining BioBasis personnel, so that they can be checked until the families have left the area.

Leave several nest markers scattered in the terrain and move them now and then to prevent foxes from discovering their connection to nests.

3.2.6. Laboratory work

The incubation stage of wader and skua eggs is estimated by using the criteria stated in table 3.2.6.1 or in figures 3.2.6.1-5 (see also van Paassen et al. 1984).

The age of wader and long-tailed skua young are similarly determined by means of species specific growth curves (figs 3.2.6.1-6). The estimated hatching date of the largest young in a brood is recorded as the hatching date of the brood.

First egg dates are consequently calculated using the egg laying, incubation and fledging periods given in table 3.2.6.2. Incubation periods for a number of other species are given in table 3.2.6.3.

After the season, ringing reports are mailed to Copenhagen Bird Ringing Centre, Natural History Museum (Zoological Museum), University of Copenhagen, Universitetsparken 15, DK-2100 København Ø, Denmark, or via email to Greenland Ringing at the Danish Ringing Centre: ringing@snm.ku.dk.

Table 3.2.6.1. Records of angle and float height of wader and skua eggs in relation to stage of incubation (given as days after start of incubation, but calculated from day of hatching).

Angle (°)	Floating above surface (mm)	Common ringed plover	Red knot	Sanderling	Dunlin	Ruddy turnstone	Long-tailed skua
Bottom							
0-10		0			0		0
10-20		3		0	0-1	0-2	2
20-30		2		2-4	2	1-4	2-4
30-40		3-4		2-4	3-4	1-5	3-6
40-50		4	5	4	3-5	6	3-6
50-60		4	5		4-6		6-9
60-70		5			6	4	5
70-80				6	6-8	6-8	5
80-90		9		6	8	5-9	10
Weightless				8		9	
Floating							
90	0-0.5	12		8	10	9	
80-90	0.5-1	12				9-11	13
70-80	1-2			12-15	13-16	10-16	15-17
70-80	2	14			15-17	14-16	
60-70	2-3			17	16-17	14-19	
60	3			19		19	
45	>3					18	

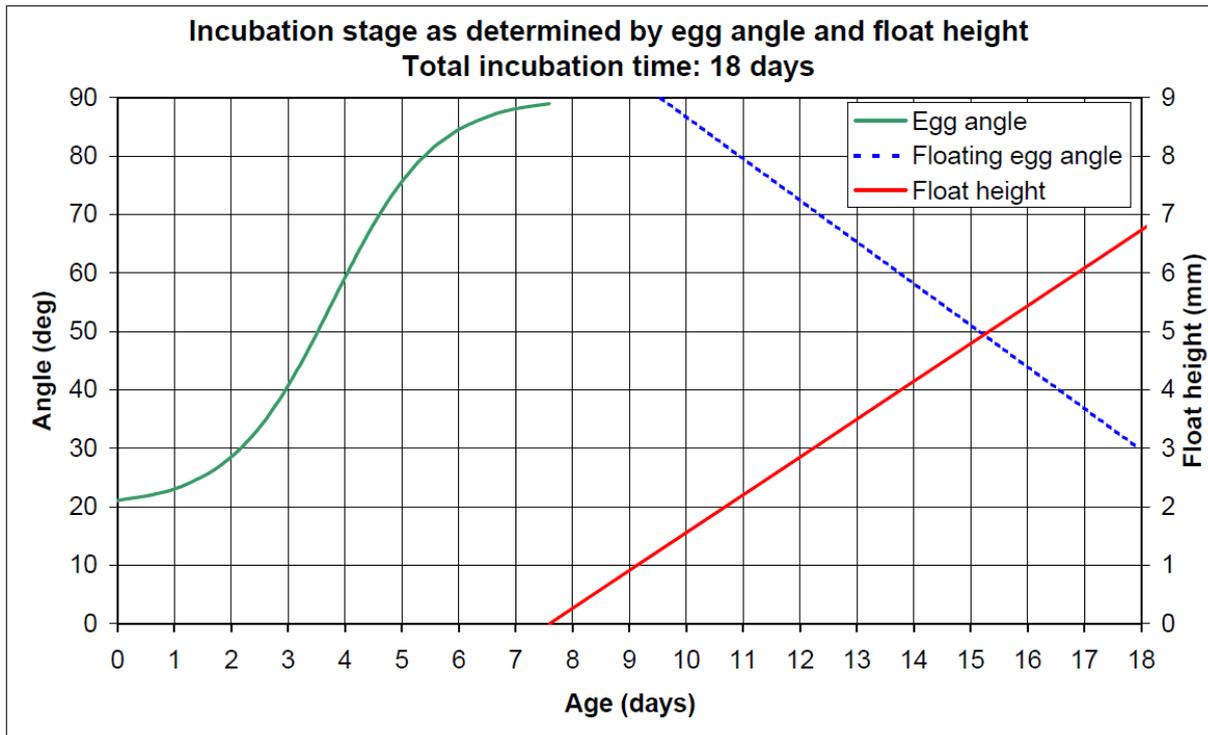


Figure 3.2.6.1 Stage of incubation at different flotation angles and height for species with 18 days of incubation. At Zackenberg, red-necked phalarope, while red phalarope has incubation of 18-19 days. From http://www2.dmu.dk/1_Viden/2_Miljoe-tilstand/3_natur/biobasis/egg_float_graphs.asp - derived from Meltofte et al. 2007.

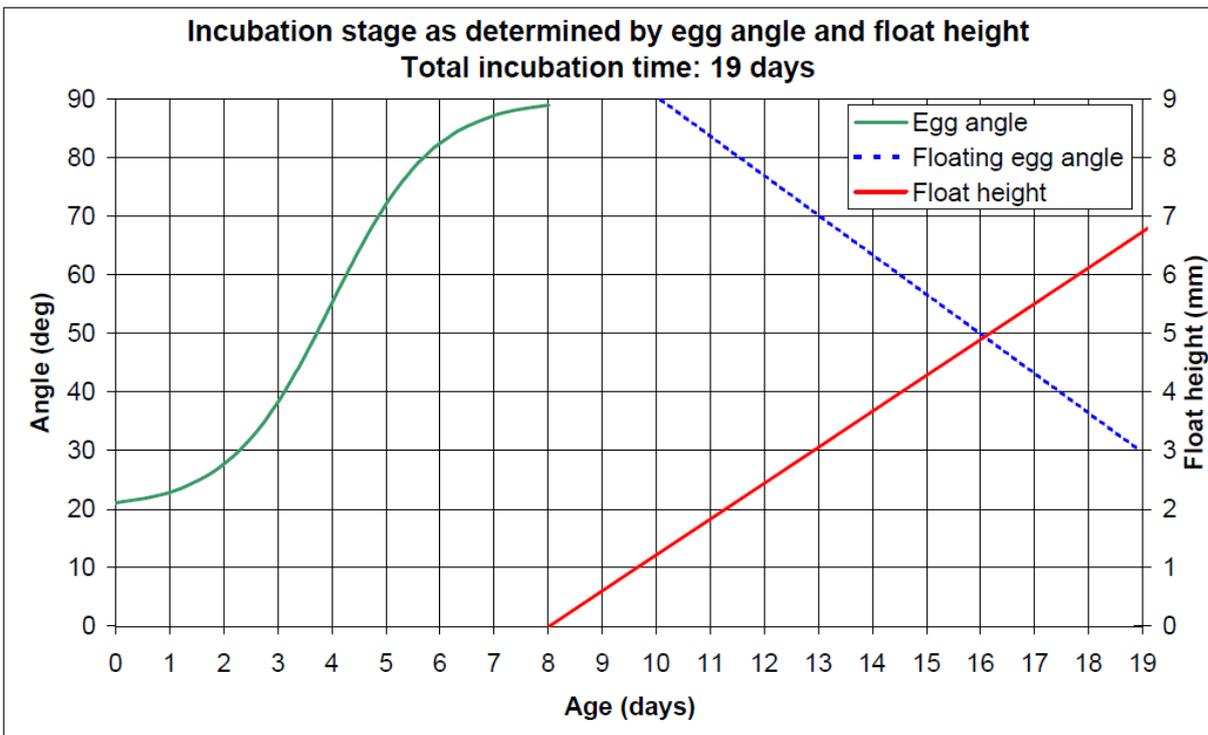


Figure 3.2.6.1 Stage of incubation at different flotation angles and height for species with 19 days of incubation. At Zackenberg, red phalarope has incubation of 18-19 days. From

http://www2.dmu.dk/1_Viden/2_Miljoe-tilstand/3_natur/biobasis/egg_float_graphs.asp - derived from Meltofte et al. 2007.

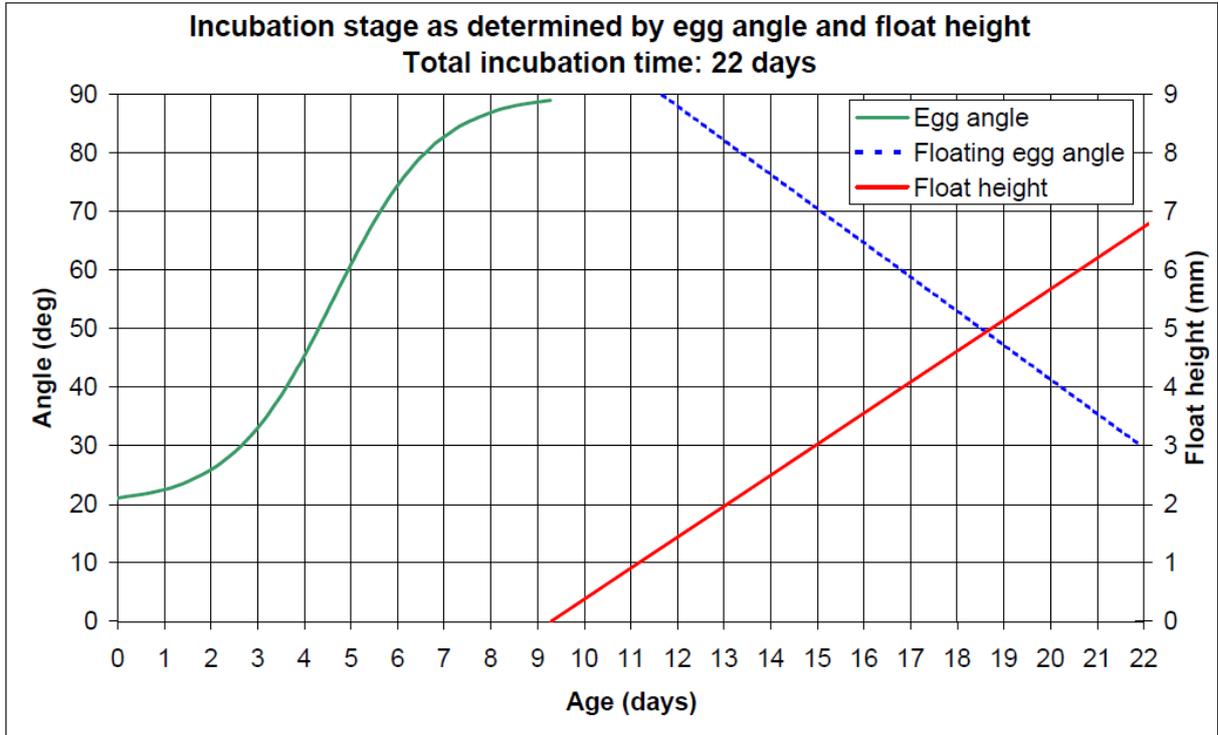


Figure 3.2.6.1 Stage of incubation at different flotation angles and height for species with 22 days of incubation. At Zackenberg, red knot, sanderling, dunlin and ruddy turnstone. From http://www2.dmu.dk/1_Viden/2_Miljoe-tilstand/3_natur/biobasis/egg_float_graphs.asp - derived from Meltofte et al. 2007.

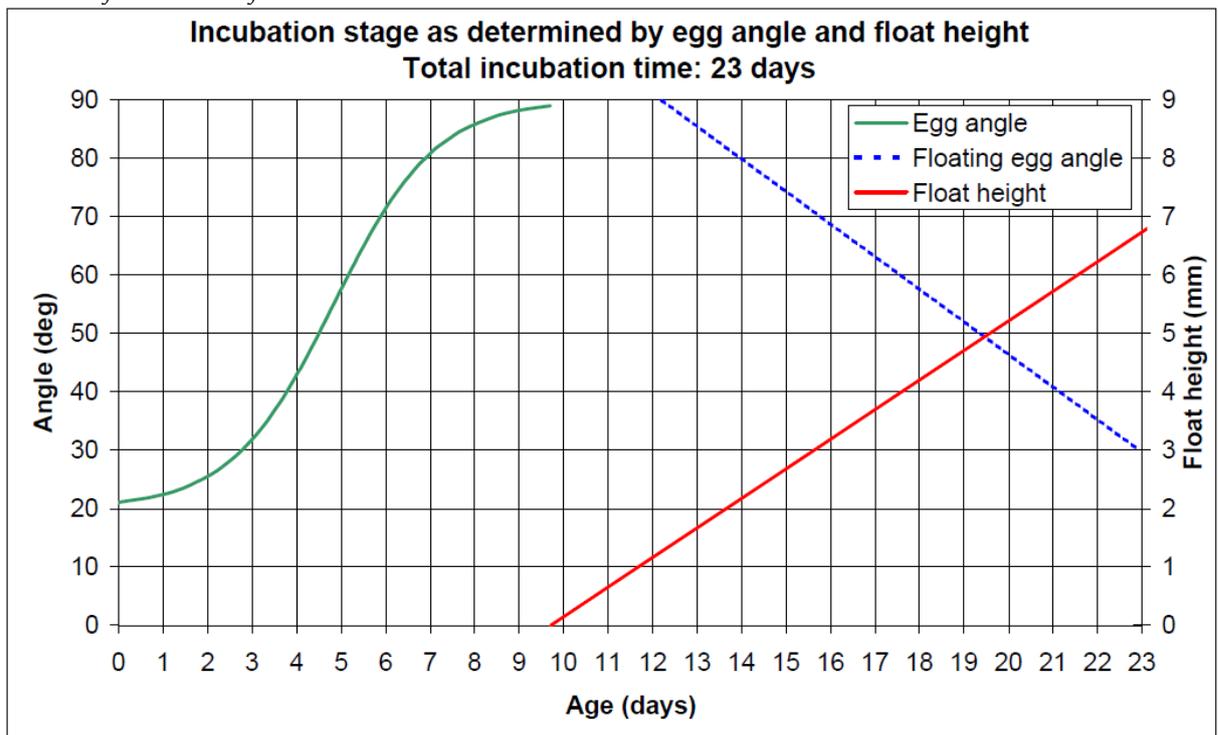


Figure 3.2.6.1 Stage of incubation at different flotation angles and height for species with 23 days of incubation. At Zackenberg, long-tailed skua. From http://www2.dmu.dk/1_Viden/2_Miljoe-tilstand/3_natur/biobasis/egg_float_graphs.asp - derived from Meltofte et al. 2007.

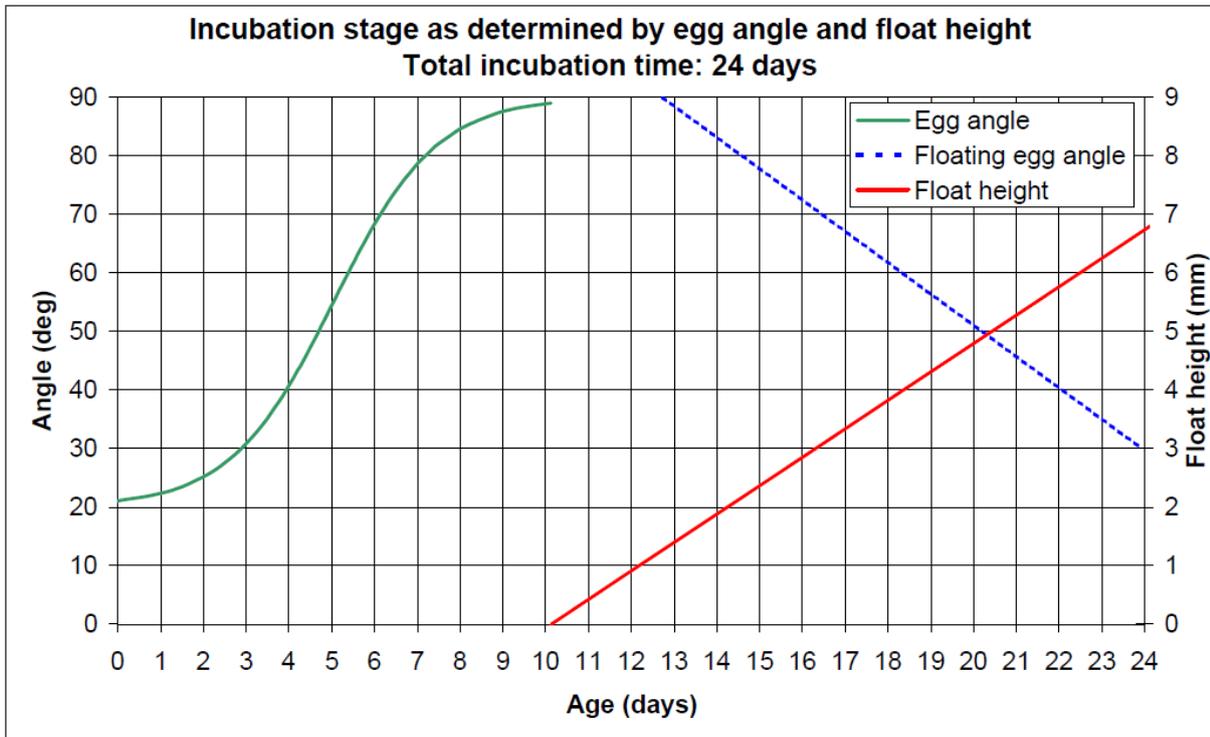


Figure 3.2.6.1 Stage of incubation at different flotation angles and height for species with 24 days of incubation. At Zackenberg, common ringed plover. From http://www2.dmu.dk/1_Viden/2_Miljoe-tilstand/3_natur/biobasis/egg_float_graphs.asp - derived from Meltofte et al. 2007.

Table 3.2.6.2. Egg laying, incubation and fledging periods (days) of waders and skuas. Add 1-2 days for fully fledged, but still downy-necked wader young.

Species	Egg-laying (4 eggs)	Egg-laying + incubation	Fledging
Common ringed plover	5	29	24
Red knot	4	26	19
Sanderling	4	26	17-20
Dunlin	4	26	17-20
Ruddy turnstone	4	26	17-20
Long-tailed skua	2 (2 eggs)	24 (1 egg)	25

Table 3.2.6.3. Incubation periods for other avian species.

Species	Days
Red-throated diver	25-27
Pink-footed goose	28
Barnacle goose	24-25
Long-tailed duck	26
Common eider	24-26
King eider	22-24
Rock ptarmigan	21
Red-necked phalarope	18
Red phalarope	18-19
Glaucous gull	27-28
Sabine's gull	21
Arctic tern	21-22
Snowy owl	32
Raven	21
Arctic redpoll	11
Snow bunting	13

3.2.7. Input of data into database

All observations and controls of nests and young are entered into the Excel data files "Bird nests" and "Bird broods", respectively. The file with bird nests gives the following columns: Species, Date (YYYY-MM-DD), Serial no., UTM East, UTM North, Observer, Nest building (yes, or no. of individuals), Bird incubating (0 or 1), Bird(s) attending (incl. neighbours attracted by alarm calling parents), No. of eggs, Bottom angle (for the most developed egg), Float height (for the most developed egg), Float angle (for the most developed egg), No. starred, No. pipped, No. wet pulli, No. dry pulli, Band no. I, Band no. II, Band no. III, Band no. IV, Weight I (of pullus), Weight II (of pullus), Weight III (of pullus), Weight IV (of pullus), Shell fragments (from hatching eggs: yes or no), Predated shells, Smell of fox, Fox tracks/faeces (yes or no; (yes) if in snow nearby), No. of cold eggs, 1st egg date, Accuracy, Eggs laid, Pulli hatched (give known or estimated number in Conclusion – see below), and Remarks. The serial number simply follows the chronology in which the nests and broods were found. Nests and broods are numbered consecutively, so that they can be entered into the Bird territories database with the same numbering. Broods that are too old to be associated with a specific territory, together with nests and broods that are encountered outside the census area, have 100 added to their number to indicate that they do not appear in the Bird territories database. The same apply to observations of newly fledged juveniles.

The "Bird broods" data base gives the following columns: Species, Date (YYYY-MM-DD), Serial no., UTM East, UTM North, Observer, Bird(s) attending (incl. neighbours attracted by alarm calling parents), No. of pulli, No. of juveniles, Band no. I, Band no. II, Band no. III, Band no. IV, Weight I, Weight II, Weight III, Weight IV, 1st egg date, Accuracy, and Comments. Under Comments you may e.g. note that this brood possibly could be the same as x brood (when not caught or only part of the possible number of young have been found).

In the "Bird nest" database, 1st egg date (YYYY-MM-DD), Accuracy and number of Eggs laid and Pulli hatched are filled in as a conclusion record after the dated records of each nest, including the fate of the nest. This is indicated by stating "Conclusion" in the "Observer" column. In the Accuracy column, codes 1-6 denote the kind of data that the 1st egg date estimate was based upon (table 3.2.7). The known/estimated number of pulli hatched from a nest e.g. includes all starred and piped eggs from nests that later were found empty with no sign of predation. Under "comments" you may e.g. note that this nest may have been a relay of x (failed) nest.

Table 3.2.7. Codes denoting accuracy categories of estimated first egg dates in waders etc. The codes represent the different kind of data that the estimates and calculation are based on.

Code	Description
1	Record of incomplete clutch
2	Record of starred and piped eggs or young still in nest
3	Record of pulli weights
4	Record of egg floating
5	Age estimates of chicks that were not caught and measured
6	Observations of newly fledged juveniles (e.g. with downy head and neck) that were still in company with alarm calling adults

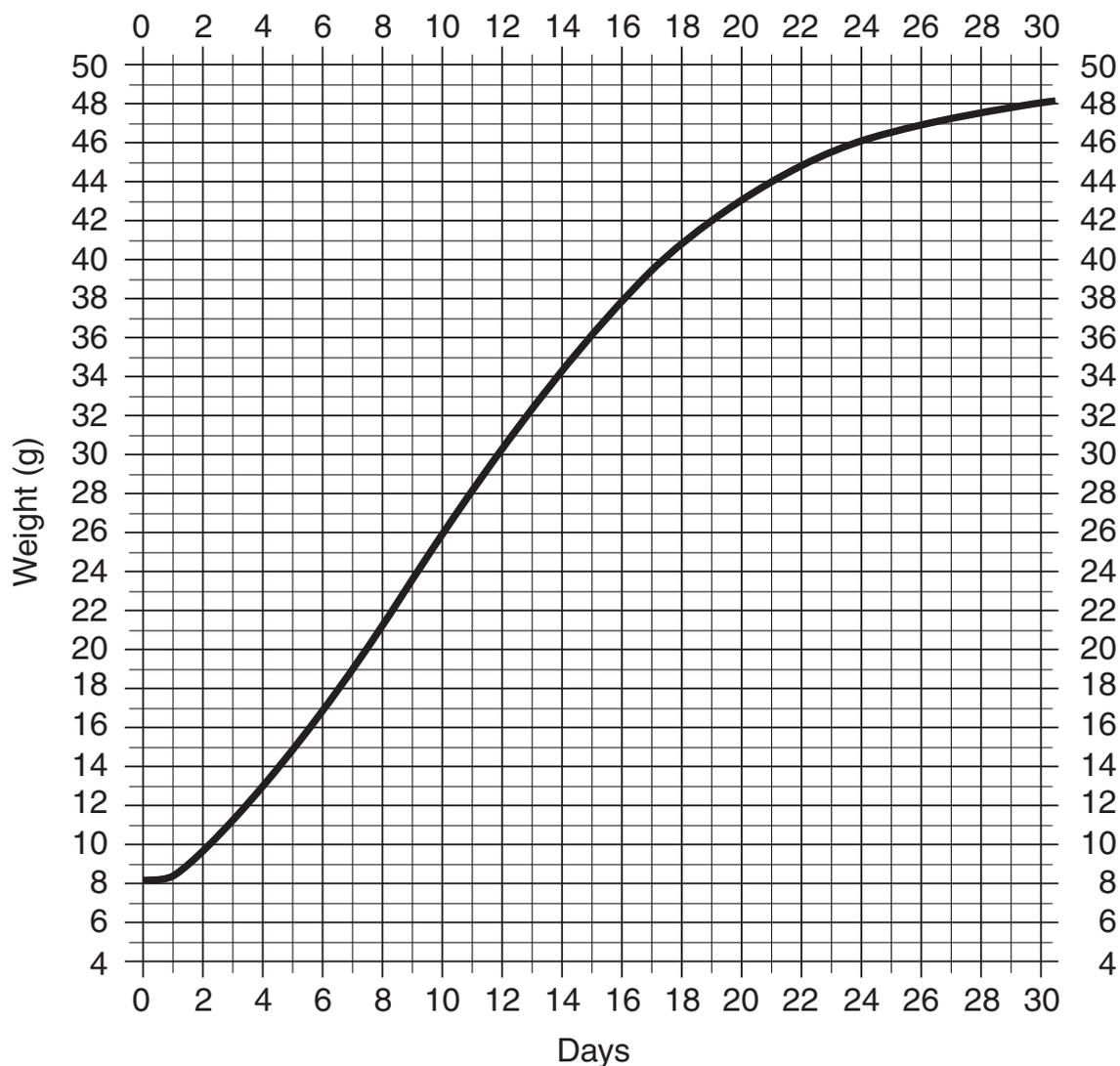


Fig. 3.2.6.1. Growth curve for common ringed plover chicks (modified from Hans Schekkerman in litt.).

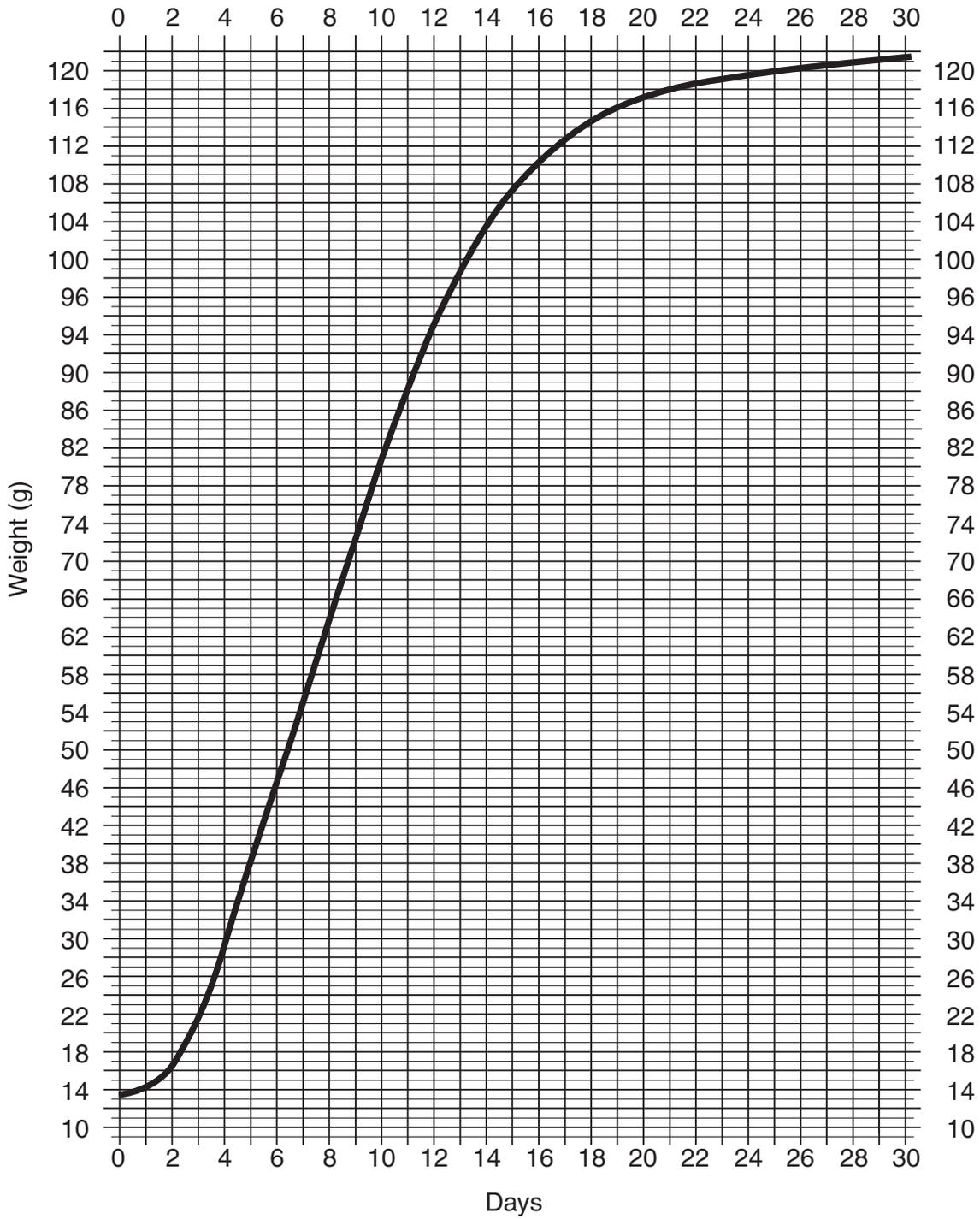


Fig. 3.2.6.2. Growth curve for red knot chicks (modified from Hans Schekkerman in litt.).

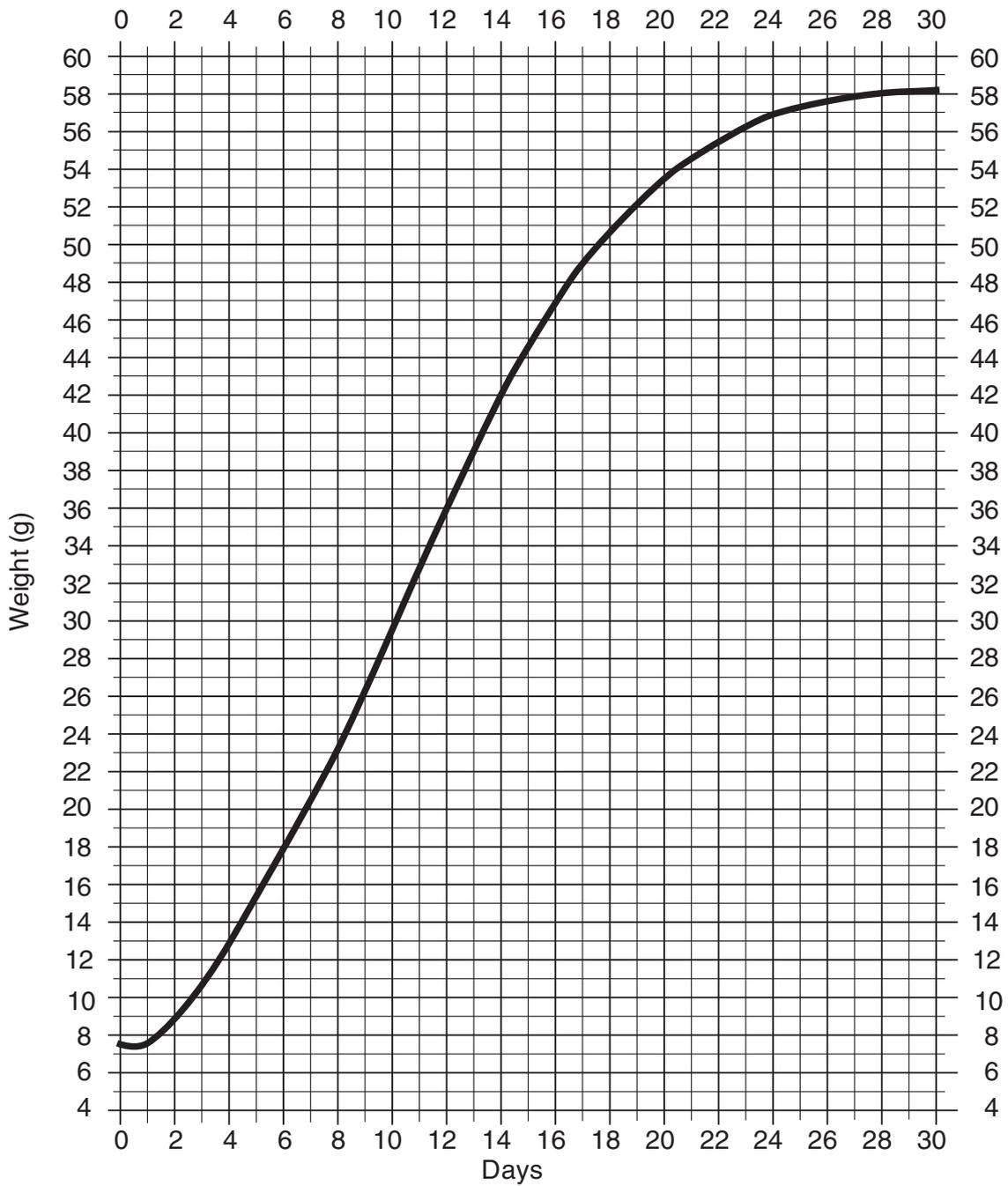


Fig. 3.2.6.3a. Growth curve for sanderling chicks (modified from Hans Schekkerman in litt.).

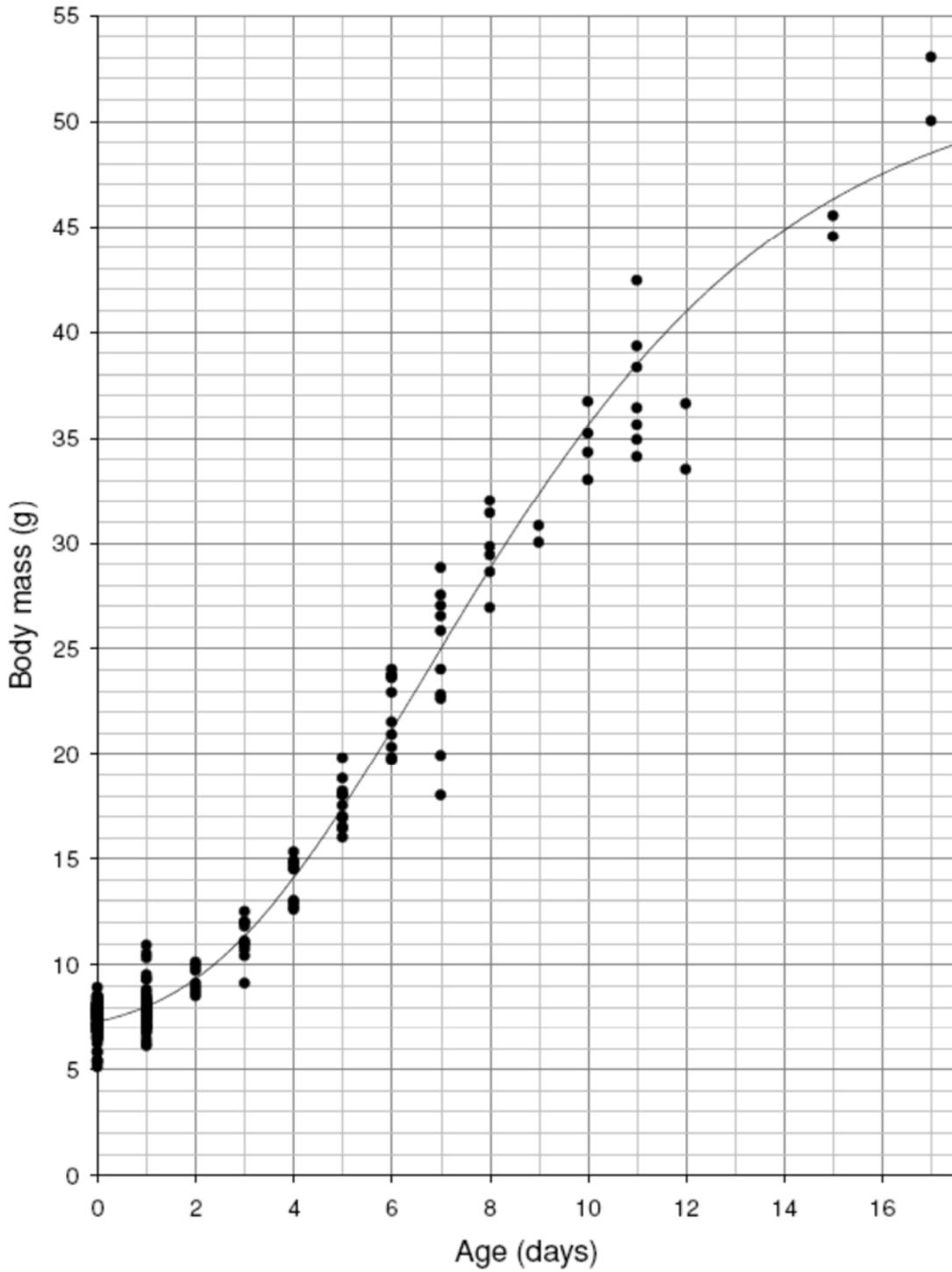


Fig. 3.2.6.3b. Growth curve for sanderling chicks from NE Greenland (J. Reneerkens, Uni. Of Groningen, unpubl.)

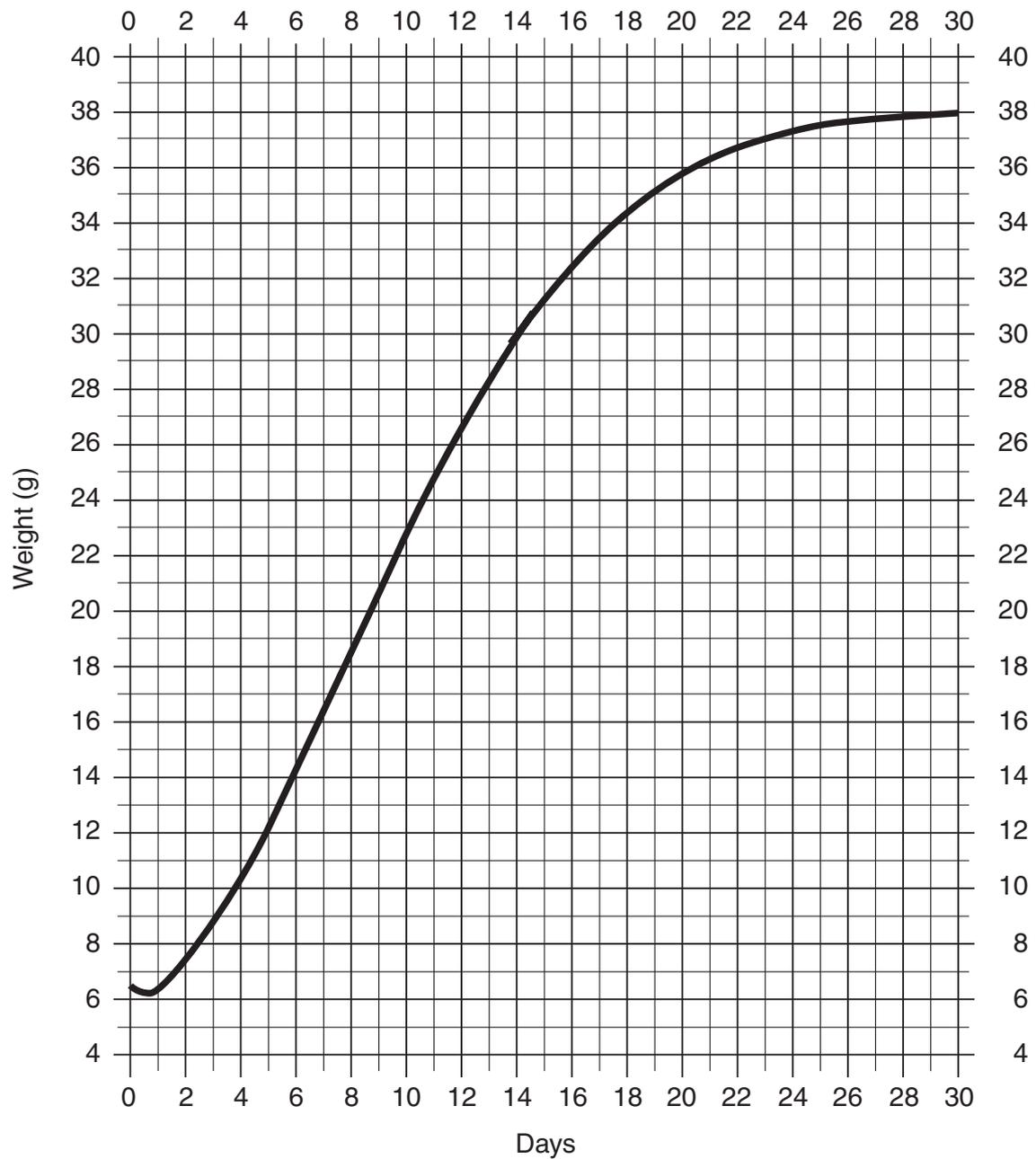


Fig. 3.2.6.4. Growth curve for dunlin chicks (modified from Hans Schekkerman in litt.).

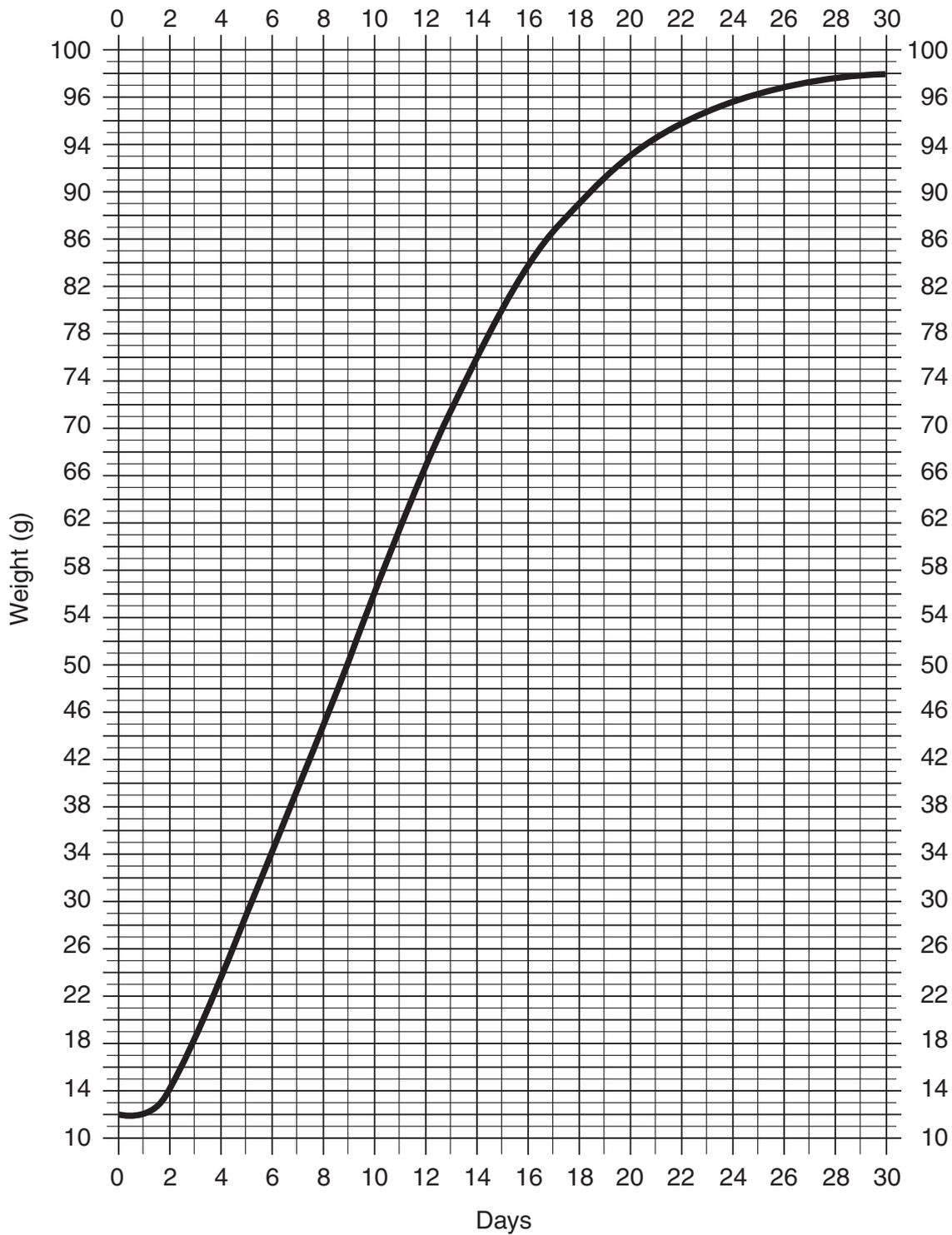


Fig. 3.2.6.5. Growth curve for ruddy turnstone chicks (modified from Hans Schekkerman in litt.).

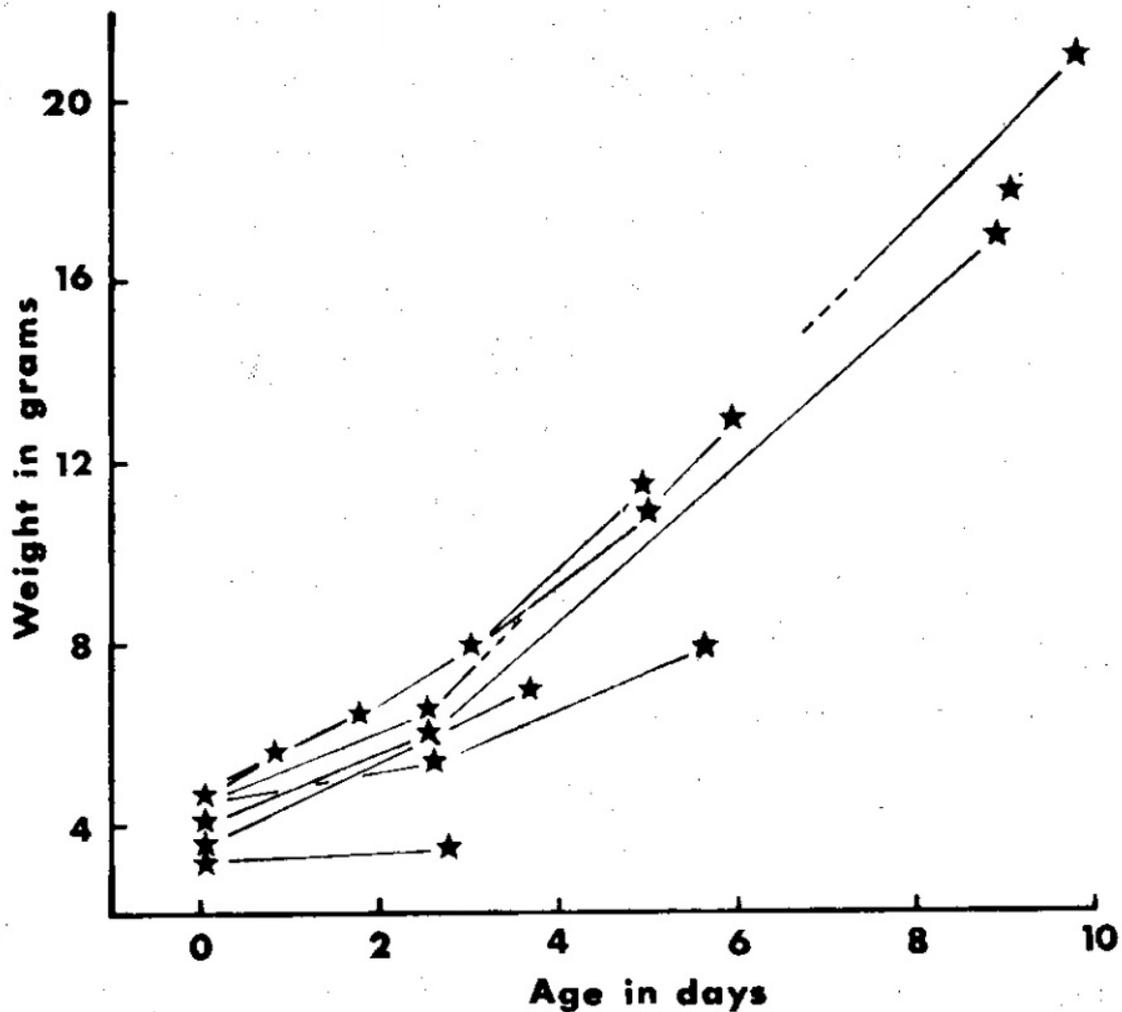


Fig. 3.2.6.6. Growth curve for red-necked phalarope chicks (from Hildén & Vuolanto, 1972).

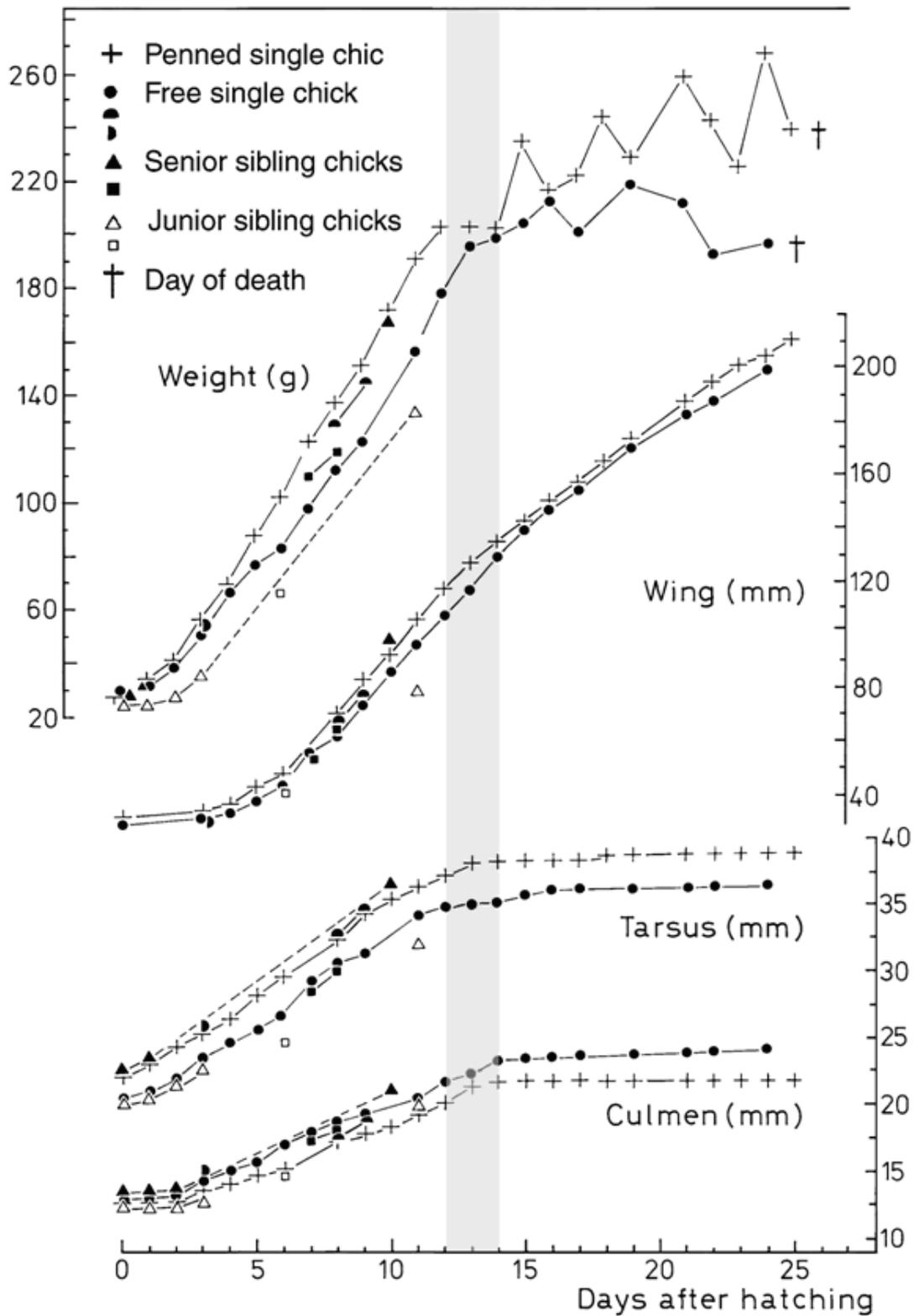


Fig. 3.2.6.7. Growth curves for long-tailed skua chicks (from de Korte 1986). Weights and measurements of eight chicks at Kårelo 1975.

3.3. Census of barnacle goose broods in Zackenbergdalen

3.3.1. Species to be monitored

Barnacle goose *Branta leucopsis* (bramgås).

3.3.2. Frequency of sampling

The presence of broods in Zackenbergdalen is monitored during late June, July and August.

3.3.3. Equipment to be used

- Binoculars
- Spotting scope
- Notebook/PDA with GPS

3.3.4. Location and marking of sampling plots

None.

3.3.5. Sampling method

Broods are recorded whenever they are encountered in Zackenbergdalen. The traditional brood rearing site at Lomsø must be checked regularly by spotting scope from a roof of a station building or from the gravel plateau SE of the runway.

3.3.6. Laboratory work

None.

3.3.7. Input of data into database

Data are entered into the general bird observation database 'Bird observations' (see section 3.5) giving as many details as possible on brood sizes, location, reactions to disturbance etc.

3.4. Census of breeding birds on Sandøen

3.4.1. Species to be monitored

- Common eider *Somateria mollissima* (ederfugl)
- Sabine's gull *Larus sabini* (Sabinemåge)
- Arctic tern *Sterna paradisaea* (havterne)
- Other waterbird species breeding in the colonies

3.4.2. Frequency of sampling

Annually in mid or late July, when the ice in Young Sund has broken up (only provided that relevant personnel is available, both logistical and scientific staff).

3.4.3. Equipment to be used

- Binoculars
- Bird rings from the Zoological Museum, University of Copenhagen
- Ringing pliers
- Notebook/PDA with GPS

3.4.4. Marking of sampling plots

Use sticks to mark appropriate sections or transects of the island. Each section/transect should not be wider, than it can be overlooked by one (or two) person(s) passing along and counting the nests. Sticks are moved consecutively in parallel rows, as the island is covered. Alternatively, if time does not permit a full census, census one or both of the two survey plots. If only one, prioritise the plateau plot (Fig. 3.4.4.1). The survey plots have the following positions. Plateau: N74 15 50.9 W20 09 26.5, N74 15 52.2 W20 09 22.9, N74 15 53.3 W20 09 27.4, N74 15 52.0 W20 09 31.0, and southern part: N74 15 44.9 W20 09 58.3, N74 15 43.6 W20 09 55.5, N74 15 44.3 W20 09 50.5, N74 15 45.8 W20 09 53.0.

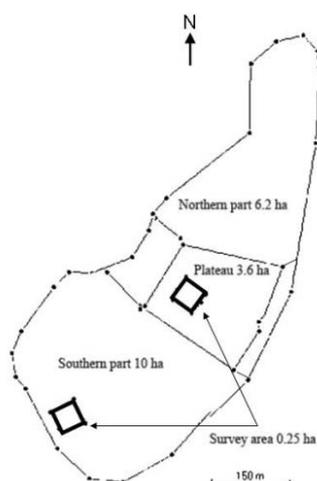


Fig. 3.4.4.1. Sandøen, with sub-areas and survey plots indicated. After Tøttrup & Levermann, in Klitgaard, Rasch and Caning 2007.

3.4.5. Sampling method

In mid or late July the number of nests of eiders, Sabine's gulls and Arctic terns on Sandøen are counted per sector. All nest cups that are or have been in use during the season are accepted as occupied nest cups. The number of eggs or young in each gull and tern nest is recorded. If incubating eiders are accidentally flushed from the nest, the eggs are counted, whereupon they are covered with the nest down. Egg identification is conducted following the description given below:

Sabine's gull

Eggs subelliptical, smooth and **slightly glossy; olive to buff-olive**, variably marked **darker olive-brown**, sometimes concentrated in zone around broad end. 44 x 32 mm

(41-47 x 30-33). Clutch: 2 (1-3). Incubation: 23-25 days by both parents. Young have much longer tarsi than Arctic terns. Fledging period: Unknown.

Arctic tern

Eggs subelliptical, smooth and **not glossy; pale buff to olive**, rarely brown, also variably blotched, spotted, and scrawled black and dark brown. 41 x 30 mm (36-46 x 26-33). Clutch 1-3. Incubation: 20-24 days. Young have so short tarsi that there is hardly room for a ring. Fledging period: 21-24 days.

All young of Sabine's gull and up to 100 young of Arctic tern are ringed.

3.4.6. Laboratory work

None.

3.4.7. Input of data into data bases

Count results are stored in the "Bird observations" database.

3.5. 'Random' observations

3.5.1. Species to be monitored

All bird species (plus foxes and lemmings).

3.5.2. Frequency of sampling

Continuously during entire field season.

3.5.3. Equipment to be used

- Binoculars
- Spotting scope
- Notebook/PDA with GPS

3.5.4. Marking of sampling plots

None.

3.5.5. Sampling method

Keep watch for everything with feathers. Record flock size, sex and age, special behaviour, geographical position etc.

Especially, Gadekæret, Sydkærene Kystkærene, Lomsø and the deltas should be checked every day. Furthermore, keep an eye on all potential nesting ponds and lakes for red-throated diver (see section 3.1.4).

Around 20 July, the moulting pink-footed and barnacle geese around Halvøen and along the remaining part of the coast off Zackenbergdalen should be recorded. Bring a spotting scope and make the count around Halvøen in the afternoon, if it is a

sunny day. Cover the lakes in Morænebakkerne and west of Lindemanselven in the same period.

Additionally, record all observations of foxes and lemmings together with other unusual occurrences during the bird census work.

3.5.6. Laboratory work

None.

3.5.7. Input of data into data bases

All records are kept in a journal (Excel files named "Bird observations") together with general information on snow and ice conditions, weather etc. The file should hold the following columns: Date (YYYY-MM-DD), Observer, Species, Observations, etc.

When series of observations concern different individuals, they are given with plusses (+) between them. When they concern possible re-sightings, they are given with commas between them. It is not necessary to enter observations of scattered breeding birds already plotted on the maps, but all special observations of e.g. pre-breeders, post-breeding flocks, migrating birds, immatures etc. should be stated.

3.6. Prioritised work schedule for the bird work

Pre-breeding/Nesting

Once snow has melted sufficiently, and birds are starting to settle (often mid-June), getting the 6 day long bird census done has the main priority. Following completion of the census, nest search is initiated.

Priority species are: sanderling, dunlin, turnstone and long-tailed skua. All other species are only followed if found opportunistically. Once at least ten nests or broods have been found of each of the focus species (bar long-tailed skua) has been found, other species may be sought for more intensively, if other BioBasis duties permit. For long-tailed skua, find as many nests as possible.

Hatching

During the hatching period, the main effort is put on visiting nests near or at hatching. A calendar of nests must be kept to ensure that no nests are missed. This calendar should be kept and followed strictly. Every evening, plan which nests to visit the following day.

Brooding

After hatching, the main focus of the bird work is search for fledglings. Date, time, state of plumage, location and whether or not the fledgling was accompanied by adult(s) is noted.

4. Mammals

4.1. Collared lemming winter nests

4.1.1. Species to be monitored

Northern collared lemming *Dicrostonyx groenlandicus*.

4.1.2. Frequency of sampling

Each year after snowmelt. It takes 70 to 100 hours to cover 60 transects within the study area, depending on the number of nests. Walking more than six transects per day is critical as your concentration drops. With four transects per day, it takes about 14 days to complete the survey.

4.1.3. Equipment to be used

- PDA, alternatively Observation sheet S1, Pencil, GPS, Notebook / Dictaphone
- Ruler
- White tray
- Tweezers
- Around 30 red and white poles to keep a straight transect

4.1.4. Location and marking of study plot

The study plot covers 106 ha between the station and Kærelv (see fig. 4.1.4). The northern and southern borders are marked by permanent white poles.

4.1.5. Sampling methods

East-west running transect lines are walked with 10 m intervals (10 one meter steps) throughout the study area starting along the north border. The southern part of the area within 400 m from the south boarder may not be walked before 10 August, when the snow has disappeared. Poles are placed with regular intervals to ensure a straight transects. At least two poles should be visible in a row from a given pole. Begin by setting up poles at the northern border and 10 m south of it. Walk through this "tunnel" from pole to pole looking continuously from side to side in front of you. When reaching a pole, move it 10 meters southwards to the next parallel transect leaving a straight row backwards. Continue the search on the original transect. When reaching the end, move the last pole 10 m to the south, and continue on the new transect. Place poles with smaller distance at the eastern end of the transects, since the area sloping down towards Kærelv is difficult to overview.

Winter nests are examined and data typed into the PDA template. In case of equipment failure, follow S1 and give UTM co-ordinates using the GPS. Turn the GPS on at arrival at a nest, and read the UTM co-ordinates at the end of the examination. Fresh and old winter nests must be separated. A typical fresh winter nest is characterised by a light yellow to light grey colour, a fresh smell of hey and a fluffy

appearance (fig. 4.1.5.1). Old nests are flat and dark and for these, only UTM-coordinates are taken (see below). The study plot has been examined since 1995 and is considered almost empty for very old nests, but unrecorded nest from previous years may be found. These should be recorded as old.

For each fresh winter nest the following data are recorded:

- Record no.
- UTM co-ordinates
- Nest diameter (1: ≤ 10 cm 2: 10-15, 3: 15-20, 4: 20-25 and 5: >25 cm)
- Position of nest (positioned into the vegetation or soil or loosely placed on top of the vegetation)
- Breeding expressed as the presence of small slender faecal pellets (length < 2 mm) in or close to the nest
- Signs of predation by stoat is recognised as a nest with large amounts of lemming fur inside the nest used for insulation when taking over the nest as stoat residence (fig. 4.1.5.2), or there may be stoat faeces or lemming bones nearby (app. within 2 meters from the nest).

Following examination, the nest is destroyed by spreading the nest material, so that it is not taken for an undiscovered nest the following year.

4.1.6. Laboratory work

None.

4.1.7. Input of data into database

Data from S1 are entered into the Excel data file named "Lemming nests" holding the following columns: Year, Observer, No. (running nest no. for each year), UTM East, UTM North, Nest type (1: Fresh; 2: Old), Nest size (1: <10 cm; 2: 10-15; 3: 15-20; 4: 20-25; 5: >25) , Nest position (1: Nest dug into vegetation/soil; 2: Nest upon vegetation/soil surface), Breeding (0: No breeding, 1: Nest has been used for breeding, showing slender pellets smaller than 2 mm), Predation (0: No predation; 1: Nest depredated by stoat), Remarks.

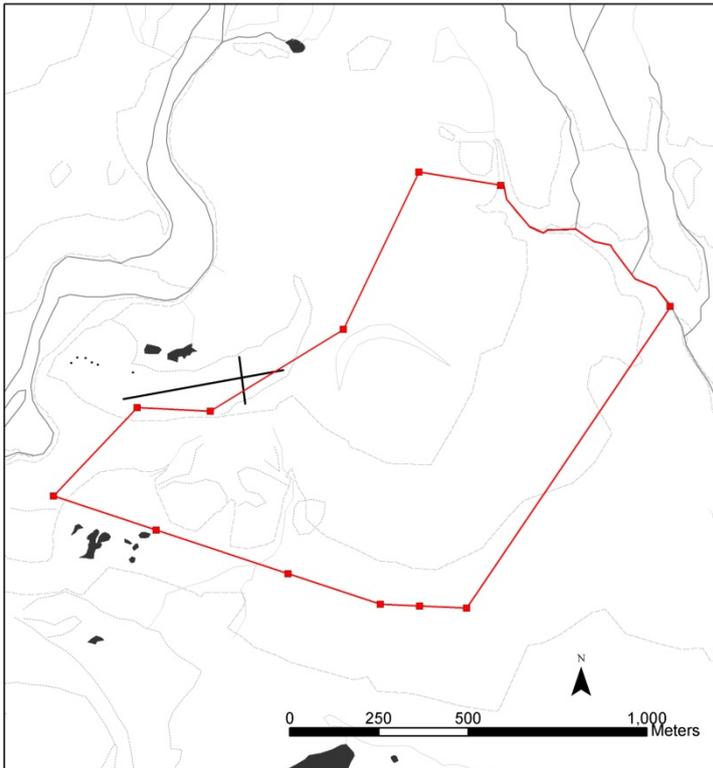


Fig 4.1.4. The lemming census area in Zackenbergdalen.



Fig. 4.1.5.1 Fresh winter nest.
Photo: Line Anker Kyhn.



Fig. 4.1.5.2 Fresh winter nest (opened) taken over by a stoat.
Photo: Aurora Photo/Thomas Bjørneboe Berg.

4.2. Weekly counts of musk oxen

4.2.1. Species to be monitored

Musk ox *Ovibos moschatus*.

4.2.2. Frequency of sampling

Total counts are made throughout July and August one or two days after the weekly flowering and invertebrate sampling dates (see table 1.1.2) or the first day thereafter with acceptable weather conditions (good visibility below 600 m curve). Additional censuses are conducted into the autumn.

4.2.3. Equipment to be used

- Binoculars (10 x)
- Spotting scope (30 x)
- Tripod
- PDA, alternatively Observation sheet S3, Field map M1, Pencil, GPS, Rangefinder
- Musk ox field guide
- Skis / snow-shoes if necessary

4.2.4. Location and marking of study plot

The musk ox census area is shown in fig. 4.3.4.

4.2.5. Sampling methods

Fig. 4.3.4 shows the basic route through the census area. The only fixed points on the route are the summit of Ulvehøj and selected fox dens.

From the top of Ulvehøj, the musk ox groups are roughly marked on M1. The route is followed eastward, looking up the individual groups. Whenever a group is found within the monitoring area it should be approached until an exact sex and age

determination can be made according to the PDA template. In case of equipment failure, use categories given on Observation sheet S3 (see Appendix 1 (Musk ox field guide)), and mark the group on M1 by the same observation number as on S3. If the encounter will disturb the group too much, the sex determination of yearlings can be dropped and the animals filed under yearling unspecified instead.

Note that the eastern border of the census area follows the ridge in front of Kuhnelv and that the route toward west does not turn until the two small lakes between Palnatokeelv and Lindemanselv can be seen. During snow melt, Aucellaelv may be too dangerous to cross, and in that case the route turns down along the east side of the river instead. Remember to specify if this detour is made. Before crossing the Aucellaelv the ice cover of Langemandssø, Sommerfuglesø, and, if possible, Store Sø in Store Sødal is estimated to nearest 10%. The total route takes nine to ten hours to complete.

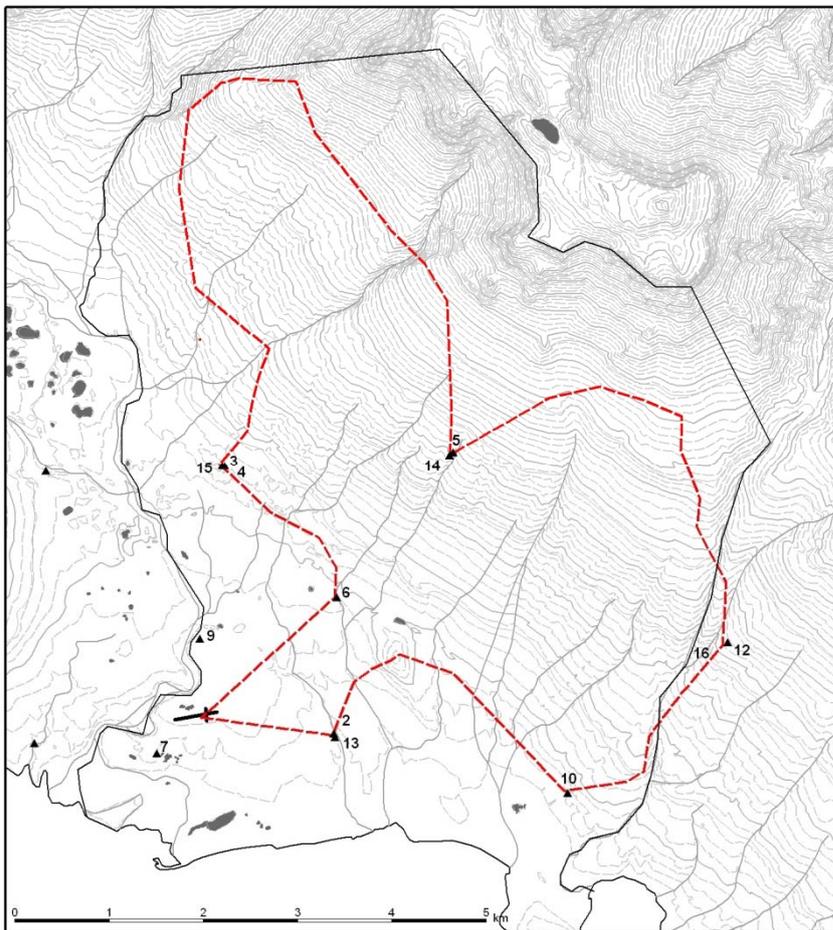


Fig. 4.3.4. The musk ox census area in Zackenbergdalen.

4.2.6. Laboratory work

None.

4.2.7. Input of data into database

Data from S3 are entered into Excel data file named "Musk ox Weekly Count" holding the following columns: Date (YYYY-MM-DD), Start (full hour), End (full hour), Observer, Cloud cover, Min. visibility, Max. visibility (km or unlim), Obs.no., UTM East, UTM North, Altitude and Group size is noted. Group composition with separate columns: M4+, M3, M2, M1, F4+, F3, F2, Unsp. Adults, F1, Unsp. 1, Calf, and Remarks. "M" indicates males, "F" indicates females, and "4+" represents animals of four years or older.

4.3. Arctic fox dens

4.3.1. Species to be monitored

Arctic fox *Vulpes lagopus*

4.3.2. Frequency of sampling

Automatic cameras are placed at all fox breeding dens (see Table 4.5.4) at the beginning of the season to verify breeding and determine litter size. Remaining dens are checked regularly for occupation and litter. Check of dens is discontinued after 15 August.

4.3.3. Equipment to be used

- PDA, alternatively Dictaphone, Map, GPS
- Binocular (10 x)
- Spotting scope (30 x)
- Tripod
- Automatic cameras

4.3.4. Location and marking of study plot

Several dens are known within Zackenbergdalen (table 4.5.4). Dens no. 7, 9, 13, 14, 15 and 16 consist of only one entrance. Den no. 9 is under the boulder Kjovestenen. Three dens are situated outside the central valley (dens no. 11, 12 and 16).

Table 4.5.4. UTM co-ordinates of fox dens.

Den no.	No. of entrances	Breeding den	Easting	Northing	Remarks
1	39	Often	511346	8264210	
2	50	Often	514505	8264281	
3	21	Often	513362	8267120	Dens #3 and #4 are regarded as one den.
4	28	Often	513338	8267074	
5	9	Rarely	515785	8267245	
6	2	No	514563	8265708	
7	1	No	512644	8264039	
8	6	?	511478	8267075	Uncertain. May be rare breeding den.
9	1	No	513104	8265270	
10	26	Often	517043	8263626	
11	13	?	520901	8260807	Den outside census area. Not visited.
12	13	Often	518701	8265230	
13	1	No	514541	8264211	
14	1	No	515759	8267237	
15	1	No	513277	8266958	
16	1	No	518685	8265260	
17	12	Often	515713	8270194	

4.3.5. Description of sampling methods

Camera traps are deployed at breeding dens as early as possible in the season. Cameras are mounted on alu-poles. Cameras are set to time-lapse mode with 1 picture every 5 minutes. Throughout the season, memory cards and batteries are changed as needed. Make sure to check the camera images now and then to ensure that everything is working properly. As long as there is snow, occupation is determined by tracks in the snow around the dens. At each visit, the entrances are checked for white or dark moulted hair, footprints and fresh digging. If present, the hairs are removed from the entrances and footprints wiped out at each visit. Also note prey remains, and if possible collect tissue samples of these (see section 5.1). When approaching a den, use your binoculars from the distance and every now and then as you get closer to check for animals present. Listen for sounds from the den and note the smell from the entrances.

Determination of presence of pups is made by 1) use of automatic cameras at the dens (see detailed camera manual for set-up instructions) or 2) extended observations from late June onwards. Fox dens nos. 2, 3, 4, 5, 10, 12, 13, 15 and 16 17 are checked on each weekly musk ox transect. If the den contains pups (actual sight or determined by footprints of pups), the litter size should be determined by a full day's observation in late June or early July. Check of dens is discontinued after 15 August.

Colour morph of all the individuals (adults and pups) must be recorded. The white colour morph is recognised during summer by a yellow-white belly, whereas the blue is brown all over. The colour morph of very small juveniles can be tricky to determine. Dark colour morph young are uniformly dark coloured, whereas white colour morph young has a bit lighter fur in the ears and under the belly.

Juvenile foxes reach adult size by October/November. Their tail differs from the adult by being less fluffy. Their size is clearly smaller than the adults during the whole field season. They may start exploring areas on their own away from the maternal den in late July or early August, and they may stay at other dens than the maternal den for some time. Take care not to record such an occupation as a new breeding den.

All new dens should be given exact UTM coordinates.

4.3.6. Laboratory work

The number of fox pups on the camera pictures are counted to estimate 1) the number of pups in each breeding den, and 2) the minimum number of young weaned (i.e. alive after 15 July) across all breeding dens.

4.3.7. Input of data into database

Data are entered into Excel data file named "Fox dens" holding the following columns: Date (YYYY-MM-DD), Observer, Den no., White ad., White juv., Blue ad., Blue juv., Hair in entrance, Tracks, Smell, Digging and Remarks.

4.4. Arctic hares

4.4.1. Species to be monitored

Arctic hare (*Lepus arcticus*)

4.4.2. Frequency of sampling

Every third day in July-August.

4.4.3. Equipment to be used

- Spotting scope (30 x)
- Tripod
- Observation sheet S2
- Pencil
- Field map M2

4.4.4. Location and marking of study plot

The census area on the east-facing slope of Zackenberg mountain (fig. 4.6.4).

4.4.5. Sampling method

Records are made from the roof of the ZERO building of the Zackenberg Station. Survey carefully the slopes of the Zackenberg mountain from the foothills to the top by means of the 30 x spotting scope. Each record of a group or a single animal is entered into the field template on the PDA. In case of PDA failure, use map (M2) and field sheet (S2). Observations of arctic hares outside the census area are recorded as random observation. All records should be given UTM coordinates obtained from the

map together with an estimate of altitude (field map M2). No counts should be made when clouds or fog covers the slopes of Zackenberg below 1000 meters a.s.l.

4.4.6. Laboratory work

None.

4.4.7. Input into data bases

Data from S2 are entered into Excel data file named "Arctic hare roof counts" holding the following columns: Date (YYYY-MM-DD), Full hour, Observer, Cloud cover x/8, Min. visibility, Max. visibility, Obs No. UTM East, UTM North, Altitude, Group size, and Remarks.

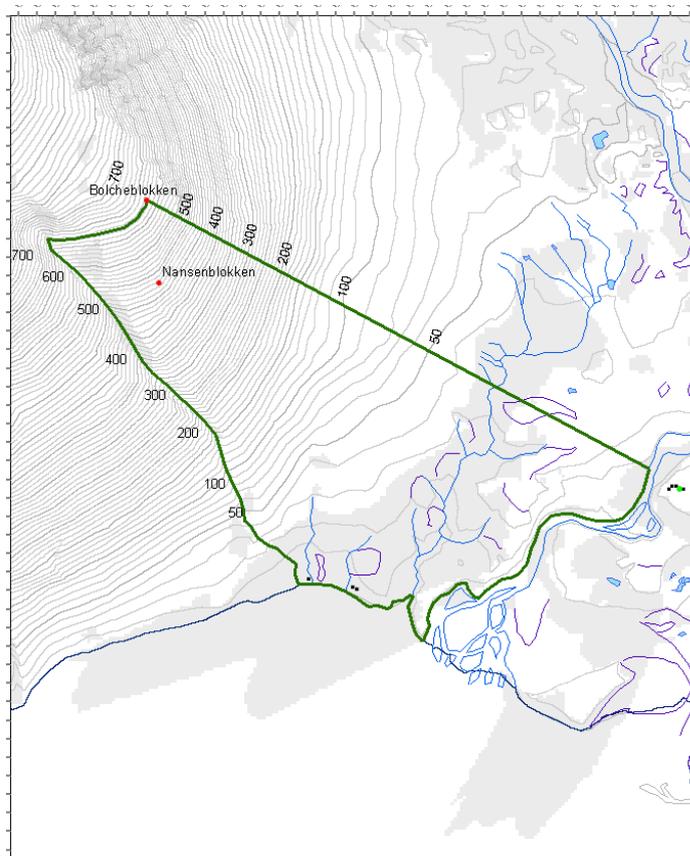


Fig. 4.6.4 The hare census area in Zackenbergdalen.

4.5. Walrus on Sandøen

4.5.1. Species to be monitored

Walrus *Odobenus rosmarus*.

4.5.2. Frequency of sampling

Annually in July/August when possible.

4.5.3. Equipment to be used

- Binoculars (10x)
- Notebook / PDA with GPS

4.5.4. Marking of sampling plots

Sandøen.

4.5.5. Description of the sampling method

The island is censused for walrus. If possible age and sex is determined.

Age estimation

Calves are less than 170 cm and have no visible tusks. At the age 1 year the animal is close to 200 cm and has around 3 cm long tusks. 2-year-old animals exceed 200 cm and have around 6 cm tusks.

Sex characteristics

Adult males have a broader nose, bigger and slightly convex shaped tusks. Male tusks are generally ellipse shaped in contrast to the more rounded shape of the female tusks. Old males have a knotty back. If the male is lying on its side, the penis opening is visible close to the navel.

4.5.6. Description of laboratory work

None.

4.5.7. Input of data into data bases

Data are entered into Excel data file named "Random Observations".

4.6. 'Random' observations

4.6.1. Species to be monitored

All mammal species.

4.6.2. Frequency of sampling

Continuously during entire field season.

4.6.3. Equipment to be used

- Binoculars (10x)
- Spotting scope (30 x)
- Tripod
- Notebook
- PDA with GPS. Observation sheet S6 in case of equipment failure

4.6.4. Location and marking of study plot

None.

4.6.5. Sampling method

Keep watch for everything with fur. Record geographical position, flock size, sex and age, special behaviour etc. All personnel and visitors must be encouraged to supply observations. For this purpose, S6 is displayed in the mess room. For musk oxen, only groups or individuals outside the census area should be recorded.

4.6.6. Laboratory work

None.

4.6.7. Input of data into the database

Data are entered into the Excel file named "Random observations" holding the following columns: Date (YYYY-MM-DD), Observer, Species, Number / track, locality, UTM East, UTM North, and Remarks.

5. Collection of wildlife samples

5.1. Tissue sampling of wildlife carcasses

5.1.1. Species or taxonomic groups to be monitored

All wildlife carcasses encountered.

5.1.2. Frequency of sampling

Ad hoc, whenever a carcass is encountered.

5.1.3. Equipment to be used

- Tweezers
- Disposable scalpels
- Gloves
- 2 Cryotubes per carcass
- 96% alcohol
- Alcohol and matches for cleaning tweezers
- GPS / field map
- Notebook
- Digital camera

5.1.4. Location and marking of sampling plots

None.

5.1.5. Sampling method

Use gloves when handling the carcasses. Tissue samples are taken using a new disposable scalpel for each individual. Tweezers are sterilized after each sampling. If possible, take enough tissue to fill 2 cryotubes, and label each tube with species code date and UTM coordinates using an alcohol resistant marker. If possible, take one molar from the carcass for age determination and put this in a small paper bag labelled with species date and UTM coordinates. Note the UTM position of the carcass, Species (see table 5.1.5), Sex, and estimated age (adult/juvenile; for musk oxen #years). For musk oxen, also note the bone marrow content (Absent, Liquid, Jelly, Solid). If however the bone is filled with fly pupae, bone marrow has been present, but has been eaten by the fly larvae. In this case, bone marrows should be indexed as Absent, and the information on fly pupae noted under Remarks. If possible, write the running number on the carcass. Take at least one photo of the carcass (for musk oxen, the photo should clearly show the horns for use in sex and age determination). For fish, also extract the otoliths.

Large carcasses are sampled in the field, while small carcasses may be brought back to the station for dissection.

5.1.6. Laboratory work

See above. Fill the cryotubes with 96% alcohol and store in the freezer. Frozen tissue and tooth samples are brought back to Bioscience, AU Roskilde.

Table 5.1.5. Species abbreviations used in the tissue collections. Please add more names and abbreviations if needed.

Class	Common name	Abbreviation
Fish	Arctic char	CHAR
Birds	Barnacle goose	BARN
	Dunlin	DUN
	Glacous gull	GLGU
	Long-tailed duck	LDUCK
	Long-tailed skua	LSKUA
	Pink-footed goose	PINK
	Raven	RAV
	Red knot	KNOT
	Red-throated diver	RTD
	Ringed plover	PLOV
	Rock ptarmigan	PTAR
	Ruddy turnstone	TURN
	Sanderling	SAND
Snow bunting	BUNT	
Mammals	Arctic fox	FOX
	Arctic hare	HARE
	Arctic wolf	WOLF
	Collared lemming	LEM
	Musk ox	MO
	Stoat	STOAT

5.1.7. Input of data into the database

In Excel the following data are entered into the carcass file: Date (YYYY-MM-DD), Observer, Running number, Location (even when exact coordinates exist), UTM East, UTM North, Species, Sex (Male, female, unknown), Estimated age (#years, or adult/juvenile), Bone marrow (0,1,2,3), Tooth sample (Yes, No), DNA sample (Yes, No), Otoliths (Yes, No), and Remarks.

Download the photos and rename to include Carcass ID and Year (e.g. PTAR006_2008).

5.2. Collection of faeces and casts

5.2.1. Species to be monitored

Faeces from stoat *Mustela erminea* and Arctic fox *Vulpes lagopus*. Casts from Long-tailed skua *Stercorarius longicaudus* and snowy owl *Bubo scandiaca*.

5.2.2. Frequency of sampling

Once a year, in the last week of August.

5.2.3. Equipment to be used

- App. 60 paper bags
- Pencil
- Pink spray painting
- Black permanent marker pen
- Large tweezers
- GPS
- Field sheet S5

5.2.4. Location and marking of study plot

29 stones between the airstrip and Kærelv, i.e. within and around the lemming study area, are marked by pink fluorescence painting on the west facing side of the stone and numbered with black numerals (table 5.2.4).

5.2.5. Sampling method

The area within 1 m from each of the 29 stones is examined carefully and all faeces and casts are collected in paper bags (one bag per sample, with reference to species, stone number and year). The painting and number are repainted if necessary. If non-marked stones with plenty of faeces or casts are found, these should be marked by pink paint and given the next serial number as well as UTM coordinates. Faeces and casts are illustrated on Fig. 5.2.5.

5.2.6. Laboratory work

The paper bags are stored open until the samples are completely dry. The material is brought back to Aarhus University.

5.2.7. Input of data into the database

Data are entered into Excel data file named "Faeces and Casts" holding the following columns: Year, Observer, Stone no., No. of fox faeces, No. of stoat faeces, No. of skua casts, No. of owl casts, Remarks.

Table 5.2.4. UTM co-ordinates
for the 29 marked stones

Stone no.	UTM East	UTM North
1	513,660	8,264,763
2	513,733	8,264,739
3	513,766	8,264,884
4	513,901	8,264,826
5	513,856	8,264,788
6	513,830	8,264,669
7	513,842	8,264,673
8	513,804	8,264,651
9	514,031	8,264,668
10	514,188	8,264,691
11	514,330	8,264,627
12	514,287	8,264,503
13	514,361	8,264,433
14	514,438	8,264,234
15	514,437	8,264,143
16	514,472	8,264,070
17	514,435	8,264,048
18	514,367	8,264,024
19	514,263	8,263,994
20	514,179	8,264,139
21	514,103	8,264,144
22	514,047	8,264,135
23	513,990	8,264,113
24	513,883	8,264,105
25	513,741	8,264,413
26	513,819	8,264,361
27	513,615	8,264,121
28	513,554	8,264,076
29	514,264	8,264,006



Fig. 5.2.5. Faeces and casts from relevant predator species in Zackenbergdalen.

6. Lakes

6.1. Physical-chemical parameters, plankton and submerged plants

6.1.1. Parameters and species to be monitored

- Ice cover
- Water temperature
- Water transparency
- pH
- Conductivity
- Chlorophyll
- Total Nitrogen (TN)
- Total Phosphorus (TP)
- Dissolved organic carbon (DOC)
- Phytoplankton (all taxonomic groups)
- Zooplankton (all taxonomic groups)
- Submerged macrophytes (all taxonomic groups)

6.1.2. Frequency of sampling

At least four times during July and August. The first sampling should take place at 50% ice coverage. Thereafter the sampling should take place with regular intervals of approximately 20 days (i.e. approximately 7 July, 1 August and 20 August if 50% ice cover is around 25th June).

Ice cover on the two lakes is estimated as percentage coverage whenever possible during June and July until the ice has melted, both at visits at the lakes and from the slopes of Aucellabjerg (using binoculars). Two automatic cameras are installed at the lakes. Early in the season, data from the SD in the 2 cameras placed at the mast between the lakes is downloaded and the cards replaced. If needed, the batteries are changed. The same procedure is conducted at the end of the field season. The camera runs year-round, and is set to take a picture every 12 hours (noon +1 h and midnight +1 h).

6.1.3. Equipment to be used

Equipment for field work

- A rubber dinghy with oars, rope and anchor*
- A pump for the rubber dinghy*
- A water sampler*
- A 30 l tub*
- A Secchi disc with a tape measurer*
- Two plastic containers with rope for buoys*
- A thermometer with a 0.2° scale
- Waders
- Life-jacket

- Six 250 ml new clean plastic bottles with water resistant labels (TP/TN + phytoplankton + zooplankton)
- One 1 l (plastic) measure
- Two 1.5 l plastic bottles per sampling with water resistant labels
- A bottle with acid Lugol solution (for preservation) and a 5 ml plastic pipette
- A 50 µm plankton net mounted with a collecting-bottle
- Funnel that fits into a 250 ml bottle
- A squeeze bottle
- A notebook and permanent marker pen
- Two automatic cameras**, incl. SD cards and batteries

Additional equipment for vegetation analysis at the last sampling date in august

- Water binocular
- Plant rake (Olsen rive) with rope
- Handheld echo sounder

*) permanently based in a metal box on the hilltop between the two lakes (see table 6.1.4).

**) Permanently installed on the mast on the hilltop between the two lakes (see table 6.1.4).

Equipment for laboratory work

Water chemistry: conductivity meter and pH meter (this equipment is already used by GeoBasis so please follow the instructions in the Geobasis manual and/or seek assistance for advice from the Geobasis personnel).

Chlorophyll: Glass fibre filters (Whatmann GF/C ø47mm) and a filtration funnel setup.

DOC: pre-prepared acid washed (or muffled) glass vials (or similar) with a tight lid (can be obtained from Kirsten S. Christoffersen kchristoffersen@bio.ku.dk), Freshwater Biological Laboratory, University of Copenhagen).

All other water chemistry analyses are conducted at laboratories in Denmark.

6.1.4. Location and marking of sampling plots

The two sampling lakes, Sommerfuglesø og Langemandssø, are the first two lakes north of Vesterport Sø, and hence the southeastern lakes in northern Morænebakkerne (table 6.1.4 and map p. 4). The sampling stations are in the deepest parts of the lakes. Sommerfuglesø is approx. 1.8 m and Langemandssø approx. 6 m deep. Cairns with red stones are situated on land to mark directions to the deepest points. At the first sampling each year, the deepest place in each lake is marked with a small plastic container tied to a rope and a stone for anchor. Note that at the first sampling at 50% ice cover it may not be possible to sample at the deepest place in the lakes, and samples are therefore taken as close to this place as possible.

Table 6.1.4. Lake UTM coordinates

Name	Easting	Northing
Sommerfuglesø	511,860	8,267,700
Langemandssø	511,830	8,267,970
Box with equipment	511,900	8,267,850

6.1.5. Sampling method

At each sampling date, the full hour, cloud cover (x/8), wind force (m/s) and per cent ice cover are recorded for each lake.

When still on land, inflated the boat and tie the anchor rope as well as the water sampling cord to the boat! Wash the tub and all bottles to be used with lake water. Bring the Secchi disc, the water sampler and the 30 l tub in the dinghy and row to the deepest place in the lake. When anchored on the sampling station, the transparency is measured with the Secchi disc on the shade side of the dinghy. Lower the disc in the water until it disappears, and then pull it gently up again until it is just visible. The depth of the disc is then taken from the tape measurer (to 0.1 m). Often the disc will be visible right to the bottom.

Collect a total of 20 l of water equally distributed from different depths of the lake. Start at the surface (0 m) and move downwards with intervals of 0.5 m (Sommerfuglesø) or 1 m (Langemandssø). The deepest samples should be taken 0.5 m above the bottom, as it is very important not to have sediments in the samples. If the lake sediment accidentally is disturbed, it is necessary to wait 5-10 minutes or move a little away before taking a new sample.

On the shore, stir the water and measure the temperature (to 0.2°) with the thermometer held in the water. Await a stabile reading (normally within 5 minutes). Take the following water samples from each lake and always remember to stir the water first:

- one sample of 150 ml into a 250 ml bottle (it is important only to fill 2/3 of the bottle) for total N and P
- one sample of approx. 245 ml into a 250 ml bottle, which is added approx. 5 ml lugol right away, for phytoplankton analysis
- one sample of min. 1.5 l for pH, conductivity and chlorophyll measurements (to be processed in the lab). It is handy for lab work to split this in two bottles per lake.
- filter 15 l of water from (use the 1 l measure) through the plankton net, reverse the plankton net and wash the content carefully down into a 250 ml plastic bottle using a squeeze bottle and a funnel (using approx. 100 ml of water) and finally add 3 ml lugol

NB! Write lake name (LS or SS), type of sample (TP/TN, Phyto, Zoo) and date on all labels with an alcohol resistant pen. The samples are hidged from direct sun exposure during procession and transportation.

A survey of the submerged vegetation is made at the last sampling date in August along 4 transects (2 North-South and 2 East-West) that are marked by white stones at the shore. Note that a proper survey is only possible in calm weather (in practice: no

wind!). Observation points are made for each 5 m along each of the transect starting at the shoreline. At each point record the following: water depth, total plant coverage (% of the observed area) within a 2x2 m area below the observer, mean height of plants and the species/taxa. The latter two parameters are estimated by using the water binocular and/or the rake. Preferential a subsample of each plant species is brought to the laboratory and frozen for later analysis. The %-coverage is divided into 6 categories: 0 = 0%, 1 = >0-5%, 2 = 5-25%, 3 = >25-50%, 4 = >50-75% and 5 = >75-100%.

After each sampling occasion the rubber dinghy must be de-inflated and stored together with the rest of the gear in the metal box to prevent foxes etc. from harming it. At the last sampling, the dinghy must be dried as much as possible with some cloth or kitchen paper, and the edge of the lid should be taped to the box to prevent snow from blowing into the box during winter. Cover the box safely with rocks to protect it against wild beasts!

6.1.6. Laboratory work

Conductivity and pH is measured to nearest 1 μ S and 0.1 pH unit, respectively. The pH-meter needs calibration with two buffers that have a pH around the value of the lakes i.e. pH 4 and 7 (see the specific manual for pH measurements).

For chlorophyll analysis, 1 l of water (record exact volume using a cylinder) is filtered through a 47 mm GF/C filter, whereupon the filter is folded once (with the Algal material turning inside), wrapped in aluminium foil (6x6 cm), put in a zip plastic bag and frozen at -20°C. Write lake name (LS or SS), date and filtered volume (ml) on each bag.

DOC analysis: Use a new piece of alufoil as work space. Be careful to keep everything clean. Two sample vials is prepared with ID label and 150 μ l 2M HCl. A 25mm (450°C treated) Whatman GF/F filter is mounted in the filter holder using tweezers (do NOT touch the filter with the fingers). A 60ml syringe is mounted with a 15cm hard Teflon tube, filled with 60ml sample water, and emptied again (this to rinse the syringe). The syringe is then filled with 50ml sample water, the Teflon tube is detached, and the filter holder attached. Push sample water through the filter until there is 40ml left in the syringe (this to rinse the filter and filter folder). Fill the two sample vials each with 15ml filter sample water through the same filter and tighten the vial lids tightly. Store the samples dark at room temperature (NOT freezer but refridgerated is fine). Dispose the filter. Syringe, Teflon tube and filter holder may be rinsed in carbon free water and left to air dry. Be very careful not to lose the clear rubber gasket of the filter holder.

Finally, make sure that all labels on the bottles are readable and well attached

Storage: the zooplankton and phytoplankton samples are stored in darkness (e.g., in a box in a cupboard (do not wrap in foil and do not freeze!). The 200 ml samples for total N and P are frozen at -20°C. The 10 ml of filtered water for DOC is refrigerated.

All samples are brought to the Department of Bioscience, Aarhus University at the end of the season. N and P samples must be kept frozen (or at least below +1°C) while

the remaining samples are kept at room temperature and darkness during the transport. The samples are sorted and analysed at relevant laboratories.

Images from the SD cards from the lake camera are transferred to a computer.

6.1.7. Input of data into the database

All recorded data are entered into separate Excel data sheets for each lake and year. After sorting of the samples, the results are added. Each sheet holds the following columns: Date (YYYY-MM-DD), Hour, Cloud (cover), Wind m/s, Lake, Ice, Temp, Secchi (cm; put the depth in brackets, if it is the bottom [always note Secchi depth even at bottom]), pH, Conductivity ($\mu\text{S}/\text{cm}$), volume of filtered chlorophyll sample (g), Chlorophyll (to $0.01 \mu\text{g}/\text{l}$), TN ($\mu\text{g}/\text{l}$), TP ($\mu\text{g}/\text{l}$), Taxon, No./l (to 0.1 individual per l), Biovolume/l (mm^3/l) and Remarks. All phytoplankton and zooplankton taxa should be stated for each sampling (including early data on ice cover), but “No sampling” should be added under Remarks, when the actual taxon was not sampled at that date. Data stated under phylum/family are unidentified taxa. The column Vol (g) is deleted, when the chlorophyll has been entered into the data sheets. Also add all data on ice conditions for the two lakes collected during the field season (section 7.1).

7. Abiotic parameters

7.1. General recordings of abiotic parameters

See 1.1, 1.4, 2.1, 2.2 and 6.1.2.

7.1.1. Parameters to be monitored

- Snow cover and snow melt
- Snow and ice on Sommerfuglesø and Langemandssø
- Start of flow in river
- Break up of fjord and sea ice

7.1.2. Frequency of sampling

Continuously during entire season.

7.1.3. Equipment to be used

- Binoculars
- Dictaphone / notebook

7.1.4. Sampling sites

All permanent plant and arthropod plots (sections 1.1, 1.4, 2.1 and 2.2). Also, keep record of snow and ice on Sommerfuglesø and Langemandssø in Morænebakkerne (see section 6.1), start of flow in Zackenbergelven and the break-up of the fjord ice.

7.1.5. Sampling method

For snow-cover monitoring in permanent plots, see sections 1.1, 1.4, 2.1 and 2.2. For ice-cover monitoring in Sommerfuglesø and Langemandssø, see section 6.1.2. Record occurrence of new snow e.g. in the upper part of the study area (give lower limit and duration of snow cover). Record the start of water flow in the Zackenberg river, and the date when the fjord ice in Young Sund off Zackenbergdalen breaks up (ice floes moving away from their original position and possible to go by boat to Clavering) and the disappearance of the ice from the outer fjord (possible to go by boat out of the fjord).

7.1.6. Laboratory work

None.

7.1.7. Input of data into data base

Data on snow-cover in permanent plots and lake ice cover are entered in PDA or flat tables. Remaining records are recorded in the diary.

8. Disturbance

8.1. Human activities in the study area

8.1.1. Parameters to be monitored

- 'Person-days' spent in the different research zones
- ATV trips in the different research zones
- Aircraft activities to and from and over Zackenbergdalen
- Discharges
- Manipulative research projects
- Take of organisms

8.1.2. Frequency of sampling

Continuously during entire field season.

8.1.3. Equipment to be used

PDA with GPS.

8.1.4. Marking of sampling plots

None.

8.1.5. Sampling method

Record arrival and departure of field active personnel together with visits in research zones other than 1A (see fig. 8.1.5). Record all trips with the ATV separated on driving on snow and bare ground, on or off the 'road' etc. Record all visits by fixed wing aircrafts and helicopters together with aircrafts passing over Zackenbergdalen at low altitude. Record discharges other than ordinary waste water from kitchen and bathrooms. Record research project manipulations of any kind, including UTM coordinates, along with other interferences to the local environment. Record all collection of individuals of plants and animals.

After the season, coordinate with Logistic manager to ensure that BioBasis and Logistics data are identical.

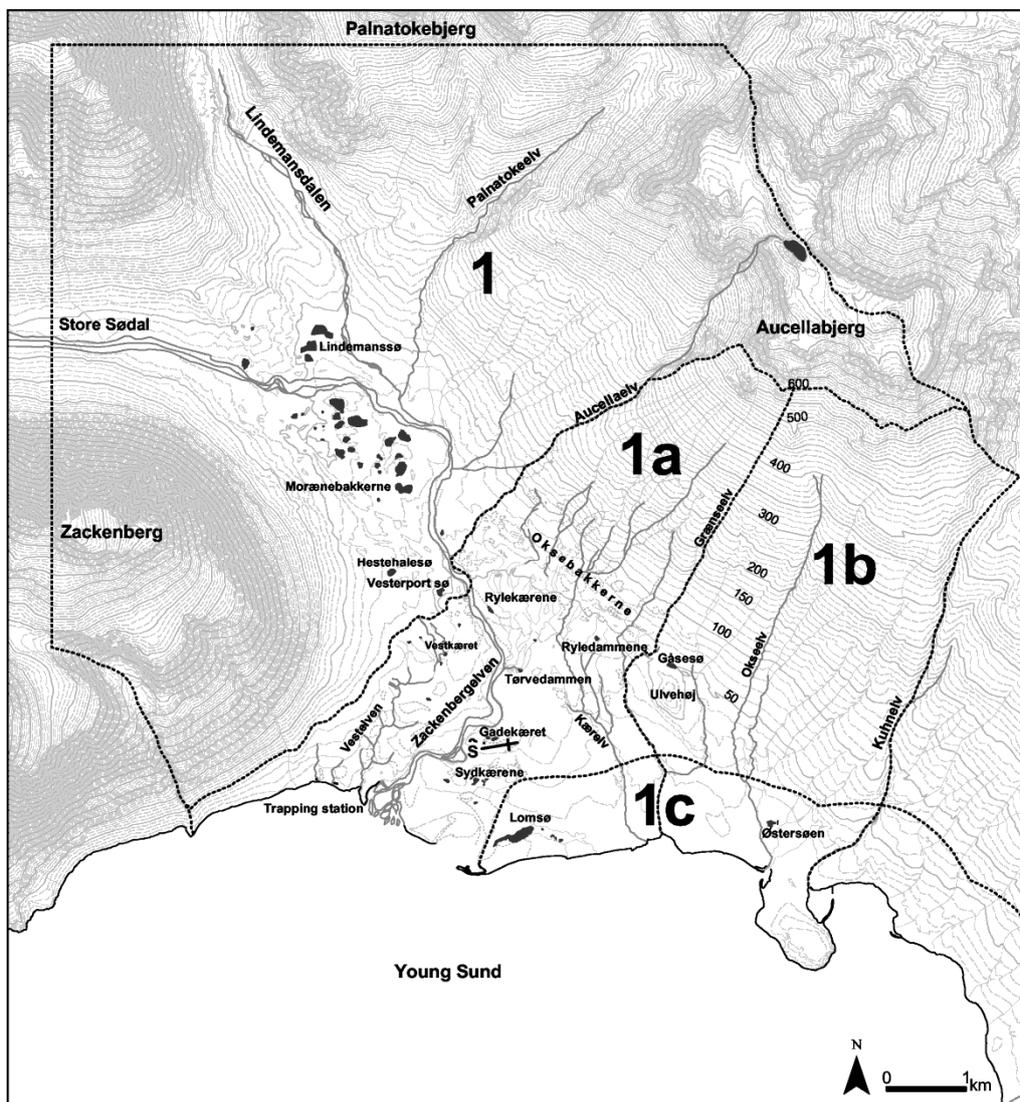


Fig 8.1.5. The study area in Zackenbergdalen with sub-zones within research zone 1. Research zone 2 is Store Sødal west of Zackenbergdalen.

8.1.6. Laboratory work

None.

8.1.7. Input of data into database

Keep an updated file of disturbance (Pax_&_disturbance). The records are summarised for each season, kept at the Dept. for Bioscience.

9. Field work schedule

See the calendar on page 91 for recordings that must take place on fixed dates.

Daily or near-daily during all or part of the summer field season

- Map breeding birds in the census area in Zackenbergdalen during June - July (no. 3.1).
- Keep record of bird nests and broods (i.e. breeding phenology and hatching success) in the census area in Zackenbergdalen during early June - mid August (no. 3.2).
- Monitor barnacle goose broods that appear in Zackenbergdalen during late June - August (no. 3.3).
- Count all Arctic hares on the east facing slopes of Zackenberg mountain every third day during July and August (no. 4.4).
- Record all 'random' observations of birds and mammals during the entire season (nos. 3.5 and 4.6).
- Keep record of snow and ice melt in the permanent plots, the entire study area and on the fjord during May, June and July (no. 7.1).
- Keep record of human activities in the different sectors of the study area including all aircraft operations in and around Zackenbergdalen (no. 8).

Weekly during the summer field season

- Record snow cover, NDVI, flowering phenology and herbivore insect occurrence in each of the 44 botanical plots together with snow cover in the four cotton grass plots and the plant community plot (section 1.1, 1.2, 1.4, 1.9, 2.1, 2.2 and 7.1).
- Empty the 20 pitfall traps for land arthropods (section 2.1).
- Empty the two 'window' traps for freshwater arthropods at the pond in Gadekæret (no. 2.2).
- Perform a complete census of musk oxen within the 47 km² census area in Zackenbergdalen, including sex and age classification (section 4.2).
- Check fox dens for occupation and cubs every week during the field season (section 4.3).

Every tenth to fourteenth day during July and early August

- Count barnacle goose families and brood sizes together with moulting immatures at Lomsø and elsewhere within the valley as often as possible.

Once (or a few times) during the summer field season

- Count flowers etc. in the 44 botanical plots once between early/mid June and early August (depending on species and plot) (section 1.2).
- Measure NDVI along the three transects covering the slopes of Aucella and the valley lowland four times during the season (section 1.10).

-
- If possible, conduct vegetation composition analysis in ITEX and UV plots in late July (section 1.13).
 - Record and map winter nests of lemmings in the census area in Zackenbergdalen after snow melt (section 4.1).
 - If appropriate, record numbers of breeding eiders, Sabine's gulls and Arctic terns together with walruses on Sandøen and at Daneborg in mid July (section 3.4 and 4.5).
 - Sample Langemandssø and Sommerfuglesø in Morænebakkerne for physico-chemical characteristics, phytoplankton and zooplankton four times during the season (section 6.1).
 - Record and sample all fresh carcasses found during the season (section 5.1).
 - Collect stoat and Arctic fox faeces together with snowy owl and skua casts at 29 selected stones at the end of each season (section 5.2).
 - Collect information on waste water and other discharge into Zackenbergelven, together with manipulative research projects and collection of organisms (section 8).

With intervals of five years

- Record changes in vegetation composition and coverage along the ZERO transect line (section 1.3) (by specialist).
- Check the 41 study plot for changes in the cryptogamic vegetation (section 1.5 and 1.6) (by specialist).
- Check the nine ITEX point frame plots for changes in species composition and cover (section 1.7).
- Check northern range species (section 0).
- Check the three GLORIA summits (section 1.14)

10. Calendar

Date	May	June	July	August	September
1		Flowering phenology and arthropod traps	Musk ox total count		NDVI transects
2					Musk ox total count
3				Flowering phenology and arthropod traps	
4	Flowering phenology and arthropod traps				
5				Musk ox total count	
6			Flowering phenology and arthropod traps		
7					Flowering phenology and arthropod traps
8		Flowering phenology and arthropod traps	Musk ox total count		
9		NDVI transects			Musk ox total count
10				Flowering phenology and arthropod traps	
11	Flowering phenology and arthropod traps			NDVI transects	
12				Musk ox total count	
13			Flowering phenology and arthropod traps		
14					Flowering phenology and arthropod traps
15		Flowering phenology and arthropod traps	Musk ox total count		
16					Musk ox total count
17				Flowering phenology and arthropod traps	
18	Flowering phenology and arthropod traps				
19				Musk ox total count	
20			Flowering phenology and arthropod traps		
21			NDVI transects		Flowering phenology and arthropod traps
22		Flowering phenology and arthropod traps	Musk ox total count		NDVI transects
23					Musk ox total count
24				Flowering phenology and arthropod traps	
25	Flowering phenology and arthropod traps				
26				Musk ox total count	
27			Flowering phenology and arthropod traps		
28					Flowering phenology and arthropod traps
29		Flowering phenology and arthropod traps	Musk ox total count		
30		NDVI transects			Musk ox total count
31				Flowering phenology and arthropod traps	

11. General duties

BioBasis personnel must, in collaboration with station personnel, encourage other station guests and visitors to respect the access regulations in the study areas (see separate ZERO folder), because it is primarily due to BioBasis interests that these regulations have been established. This concerns the obligation that all transit traffic to the east and north in the valley follows the runway to the east and then the road towards the climate station – and not along the river, over Gadekæret or on the dry ridge going north from the east end of the runway. The latter ridge holds some of the best lichen habitat in the entire valley, and we have several botanical plots and the ZERO-line on the ridge. Since this ridge is snow free much earlier than the road, people tend to use it as transit road early in the season, which is unfortunate. Note, however, that after 15 September and/or completely frozen soil, the route along the river can be utilized. Also the ban on non-research access to the “goose reserve” 1 km landwards and seawards of the coast from the old delta to “Pashytten” 2 km south-east of Halvøen during the goose moulting and gosling rearing period 20 June – 10 August is of prime importance. Finally, traffic in the “low impact study area” between Grænseelv and Kuhnelv should be kept at a minimum and monitored carefully (see section 8).

After each season, all BioBasis equipment and materials must be checked and organised tidily in the cabinets. BioBasis personnel must make a complete stock-taking list that is brought back to Aarhus University at the end of the field season. Check that there is enough of everything for one full season. Regarding alcohol for arthropod preservation, there should be enough for at least two seasons. All BioBasis equipment should be taken good care of and **should be kept in house 4, house 11 or the BioBasis shelter**. It should only exceptionally be lend to other guests at the research station and only with the obligation to deliver it back immediately after use. Naturally, damaged or defect equipment should be brought back to Aarhus University for repair or replacement equipment ordered.

Under house 4, we have a large number of ranging poles (landmålerstokke) and plastic road markers for use in the lemming monitoring. Furthermore, we have hundreds of white 50 cm long nylon sticks for use as bird nest markers and aluminium tubes for use on the ZERO-line.

In the Zarges box situated at the top of the hill between Sommerfuglesø and Langemandssø we have a rubber dinghy with oars and other lake sampling equipment (see section 6).

Concerning safety, station regulations must be carefully observed by BioBasis personnel. The safety instructions given by station personnel and Aarhus University must be followed.

The most dangerous period is during snow melt, when river beds may be filled with water soaked snow, which will ‘soak you up’, if you try to cross. Never try to cross water soaked snow! Find a place, where you can see the bottom, before you cross. This is particularly dangerous in the lowland, while on the slopes, the water normally drains out of the snow.

12. References

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Appendices

Musk ox Field Guide to sex and age classification

Calf (1)

No horns are visible. Through the winter the white hairs on the forehead become more woolly and appear to cover the horns. The neonate coat colour is dark brown with light brown underwool. There is no light coloured area (saddle) on the back as in yearlings and older animals. Guard hairs are not developed and the lower part of the legs is covered with short caramel-coloured hairs. Through the first summer the dark coat colour achieves a lighter shade.



Calf. Photo: Henning Thing.

Yearling (2)

The short horns project straight out with a very slight forward curve. The length of the visible part of the horns depends on the development of the white curly hairs on the forehead. At the end of the second winter the yearling horns have reached a length of 10–15 cm. The body size is larger than of the calf. Blackish guard hairs reach the ventral part of the abdomen but do not cover the legs. The horns of a male yearling are a little thicker, longer, and less curved than the horns of a female of the same age.



Yearling. Photo: Henning Thing.

Two-year-old female (3)

Horns are slender and appear in frontal view to project laterally straight out. In profile they are bent straight forward with white woolly hair and no horn bases are

visible. Coat colour varies somewhat with the season, as in other age-classes, being lightest in fall and darkest in late winter. Guard hairs reach down just distal of the joint to the lower legs. This cohort loses the underwool (qiviut) earlier than older animals and appears to have a smooth looking coat throughout most of the summer, when older individuals are still shedding the qiviut. Body size is significantly larger compared with yearlings and distinctively smaller than in 3-year-old females.



Two-year-old female. Photo: Henning Thing.

Two-year-old male (4)

The horns of a 2-year-old male are thicker and more conical than in a female of similar age. The proximal two-thirds projects laterally from the head at a 45° angle and the distal part is curved sharply forward. Horn ends appear very pointed. These characteristics are best seen in the profile view. Between the horns, white woolly hair covers the forehead and no horn bases are present. Guard hairs of the coat reach down proximal to the joint to the lower legs. Body size is usually slightly less than that of an average-sized adult female.



Two-year-old male. Photo: Henning Thing.

Three-year-old female (5)

In this age-class, horns reach their full length, but are still slender and have black pointed ends. Both in frontal view and in profile the curved appearance of the horns is apparent. The forehead still has cream-coloured woolly hair covering the developing horn bases. Body size is near its maximum and guard hairs of the coat may be long enough to cover half of the lower legs. Females often reach sexual maturity at this stage.

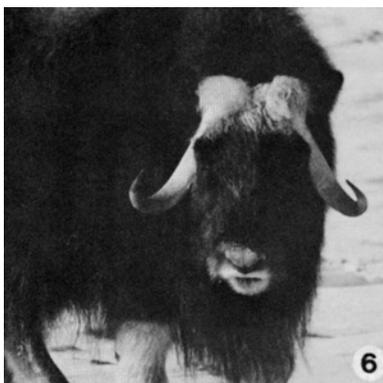


Three-year-old female. Photo: Carsten Riis Olesen

Three-year-old male (6)

Horn development in this age-class is characterised by a pronounced downward orientation of the proximal two-thirds of the horn and a distinct upcurving of the distal part. Except for the black ends the horns have a characteristic yellowish colour easily recognisable in the field.

The horn bases extending dorsolaterally from the forehead with a decreasing amount of white woolly hair, appearing as two fluffy structures at the proximal end of the horns. Horn diameter at the orbital level is greater than that of adult females, but horn spread is similar. Guard hairs of the coat may cover half of the lower legs. Body size is a little bigger than for an average adult cow.

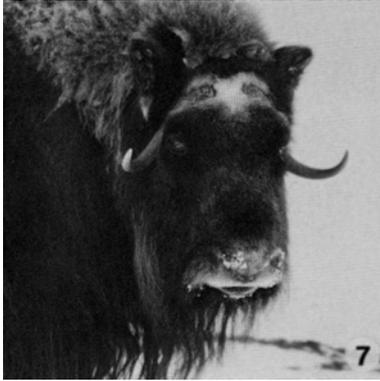


Three-year-old male. Photo: Carsten Riis Olesen.

Four-year-old and older female (7)

Beginning at the age of 4 years, and continuing to the age of 7-10 years, the whitish woolly hair on the forehead has partially disappeared exposing the horn bases, thus

allowing the observer to the best characteristic of a mature older female. Horn bases may increase to 6-7 cm in width, and therefore are much smaller than those of mature bulls. The coats of older females are often characterised by a fleece of shed under wool loosely attached to the guard hairs throughout the summer. In very old females guard hairs may be worn so short that the cream-coloured legs again are mostly exposed.



Four-year-old and older female. Photo: Carsten Riis Olesen.

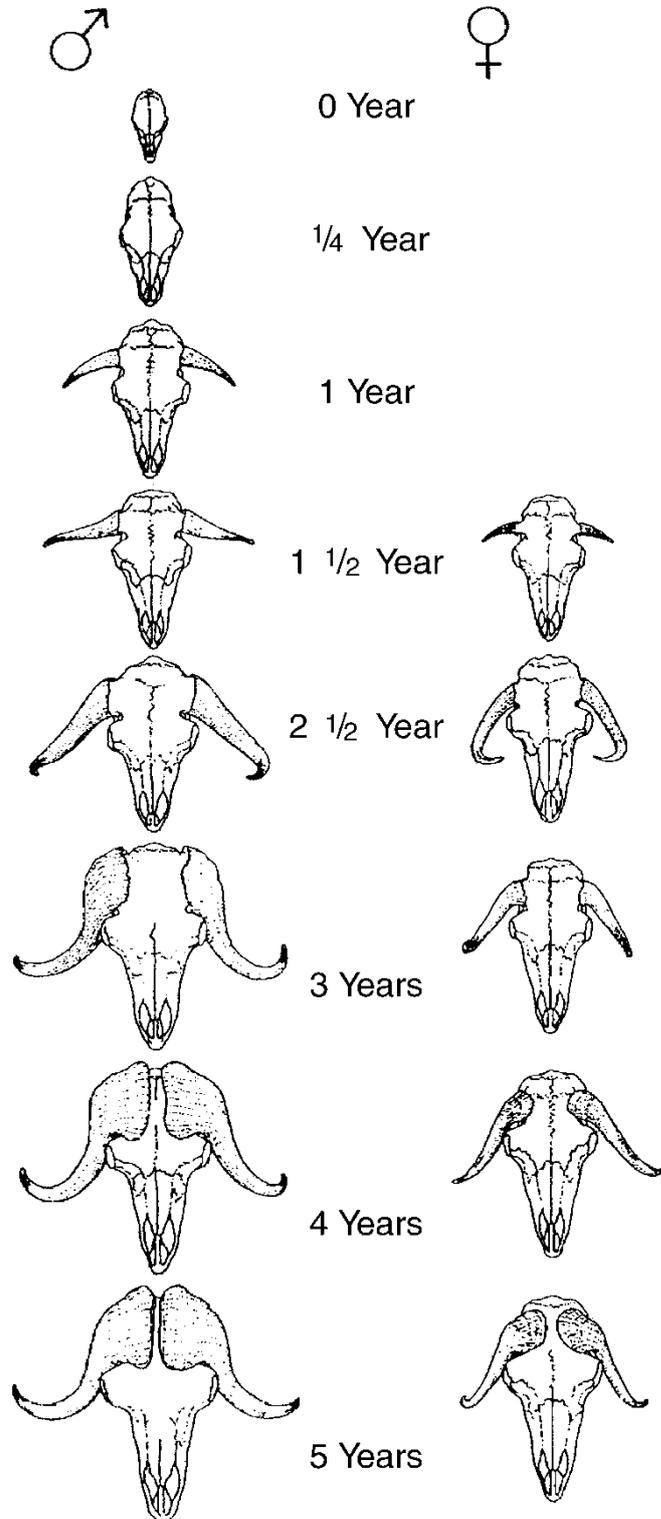
Four-year-old and older male (8)

During the fifth summer and fall the forehead patch of white woolly hair is reduced considerably and the massive developing horn boss becomes evident. When the animal turns 5 years old (Fig. 8) the horn boss has increased to a near-maximal size, leaving only a narrow (the width of a finger) hair-filled space in the midline. At this age the horns are longest with the greatest forward curve and with black-coloured pointed ends. The horn base is a massive structure with a somewhat cracked surface. It may increase to a width of 20 cm (measured as a straight line parallel to the midline between horns). Especially during winter, the massive boss may appear very light coloured when observed at a distance. This could lead the observer to misinterpret the boss as the whitish forehead hair of a cow or a subadult bull and consequently to misclassify the adult male, particularly when no cow or subadult bull is nearby for comparison. As the musk ox bull grows older than 8-9 years the horns are worn down or broken so that the curved ends and black tips disappear.



Four-year-old and older male. Photo: Carsten Riis Olesen.

Musk ox skulls can be sexed and aged with the same accuracy as with living individuals as shown in the figure below.

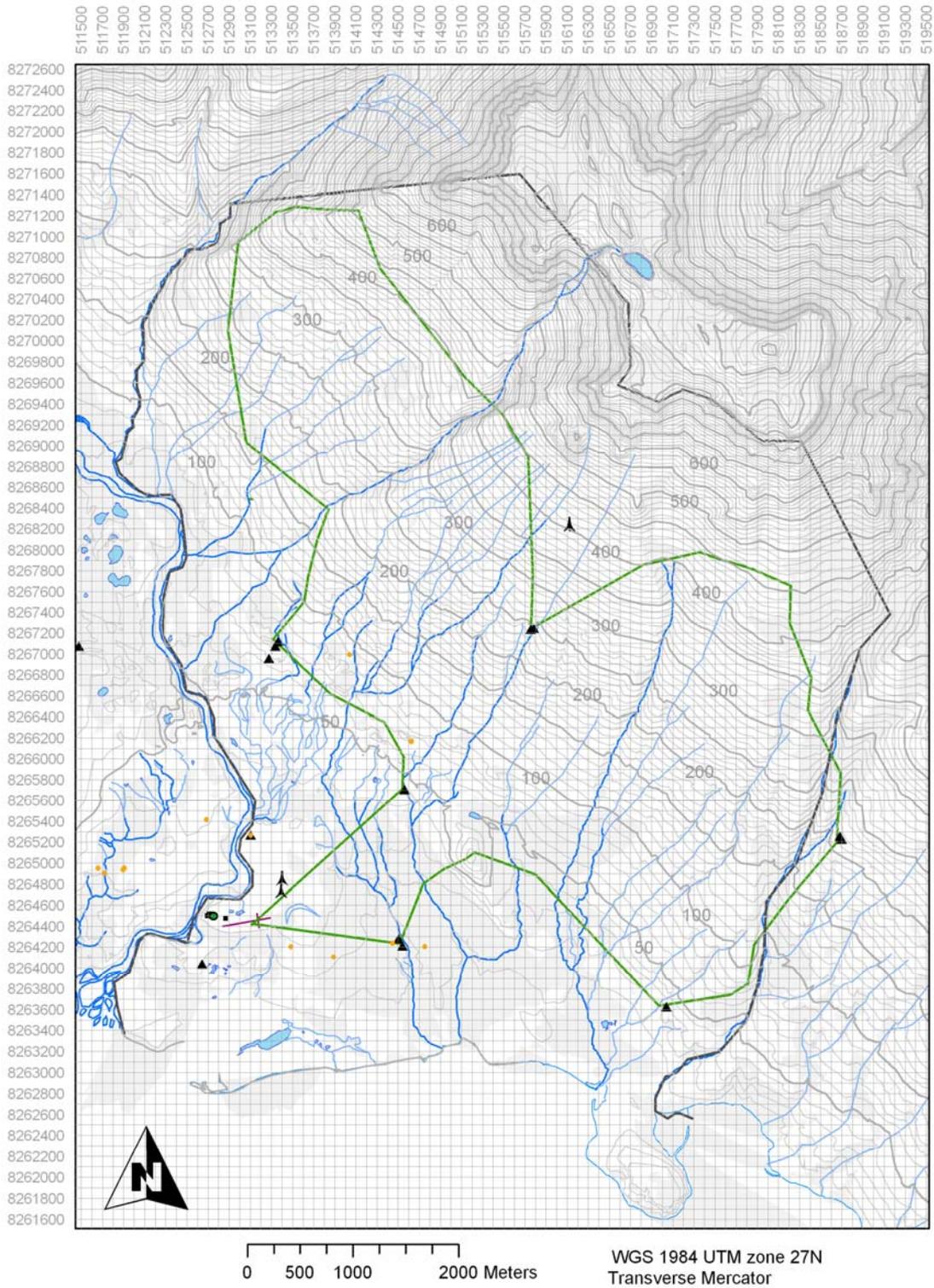


This guide is adopted from Olesen & Thing (1989: Canadian Journal of Zoology 67: 1116-1119) and Henrichsen & Grue (1980: Danish Review of Game Biology 11: 1-18).

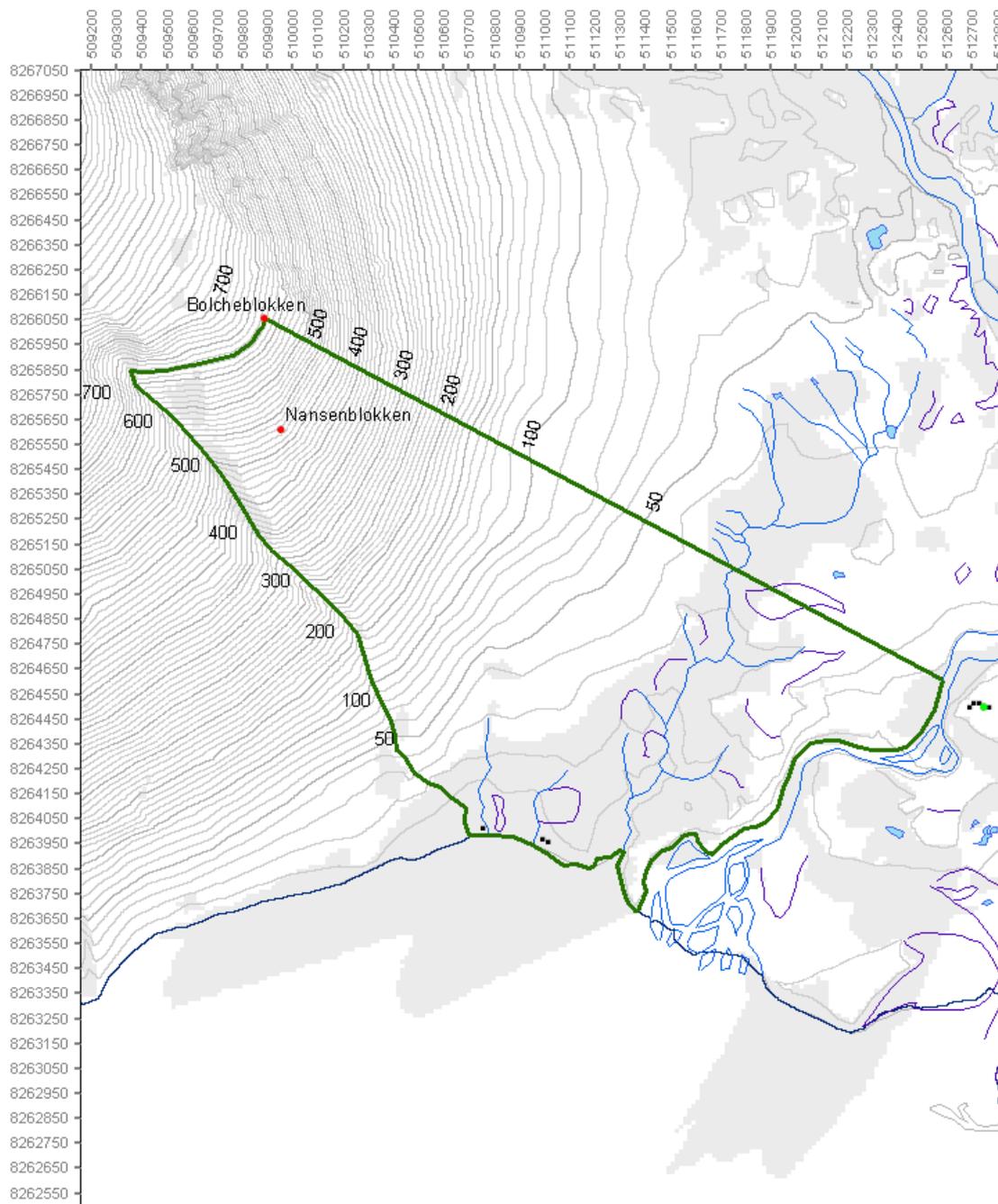
Date (YY/MM/DD): / /			Observer: _____				
No.	UTM East	UTM North	Category	Size	Placement	Breeding nest	Predation
			1: Fresh; 2: Old	1-5	1: In; 2: On	0: No; 1: Yes	0: No; 1: Stoat; 2: Fox
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S1: Lemming winter nest survey							

Date (YY/MM/DD): / /		Full hour: _____		Species: _____	
Cloud cover: /8		Visibility: -		Observer: _____	
No.	UTM East	UTM North	Altitude	Number	Remarks
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2					
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4					
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Other observations from the roof					
No. of seals on the ice in Young Sund					
No. of pink-footed geese					
No. of barnacle geese					
No. of divers					
Others					
S2: Roof counts					

Date (YYYY-MM-DD): - - Observer:



Field map M1



Field map M2