GUIDELINE FOR COLLECTION OF ENVIRONMENTAL SAMPLES TO THE GREENLAND MINERAL RESOURCES ENVIRONMENTAL SAMPLE BANK

Technical Report from DCE – Danish Centre for Environment and Energy No. 374 2026

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Data sheet

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Abstract: This technical report includes guidelines and instructions on sampling pro-cedures for collection of environmental samples in relation to mineral re-source projects in Greenland. The aim of this report is to ensure that sampling is reproducible, comparable, and done according to international standards. The report also describes the registration and reporting of the samples.

This technical report replaces the instructions provided in the 'Guideline for collection of environmental samples to the Greenland mineral resources environmental sample bank; TR239.

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Contents

Preface

This technical guideline includes instructions on sampling procedures for collection of environmental samples in relation to mineral resource projects in Greenland. The report also describes the registration of the samples and the procedure for handing over samples to Danish Centre for Environment and Energy (DCE).

The aim of this technical guideline is to ensure that sampling is reproducible, comparable, and done according to international standards (e.g., Helcom 2017, OSPAR 2019). It replaces the Technical Report 239 with more sampling details and a more practically oriented set-up.

Results from environmental monitoring of mining projects in Greenland are registered in the Greenland mineral resources environmental sample bank hosted by DCE for the Greenland Authorities, The Environmental Agency for Mineral Resource Activities (EAMRA).

Sammenfatning

Denne tekniske rapport indeholder retningslinjer og instruktioner for udtagningsprocedurer ved indsamling af miljøprøver i forbindelse med mineralressourceprojekter i Grønland. Formålet med rapporten er at sikre, at prøvetagningen er reproducerbar, sammenlignelig og udført i overensstemmelse med internationale standarder. Rapporten beskriver også registrering og rapportering af prøverne.

Rapport erstatter de instruktioner, der er givet i 'Guideline for collection of environmental samples to the Greenland mineral resources environmental sample bank'; TR239.

Summary

This technical report includes guidelines and instructions on sampling procedures for collection of environmental samples in relation to mineral resource projects in Greenland. The aim of this report is to ensure that sampling is reproducible, comparable, and done according to international standards. The report also describes the registration and reporting of the samples.

This technical report replaces the instructions provided in the 'Guideline for collection of environmental samples to the Greenland mineral resources environmental sample bank'; TR239.

1 Introduction

The purpose of environmental monitoring of a mining project is to identify and quantify the environmental impact of the project. This technical guideline is prepared to ensure that Greenland samples are collected and sampled reproducible, comparable, and according to international standards.

Danish Centre for Environment and Energy (DCE) at Aarhus University (AU) hosts the Greenland mineral resources environmental sample bank in Roskilde. This is done under a data agreement between the Greenland Authorities, The Environmental Agency for Mineral Resource Activities (EAMRA) and DCE. Samples have been collected for decades and are either stored frozen or as freeze-dried pulverized samples. The samples have been collected with the aim of elemental analyses for baseline sampling when preparing environmental impact assessment (EIA) or monitoring in relation to mineral resource exploration and extraction. A database, Mineral Resources Environmental Database (MRED), hosted at the DCE contains all sample information and in case the samples are analyzed at AU, the results of the analyses are also stored in the MRED database. EAMRA can be contacted for availability of specific data.

To ensure a high-quality monitoring program, the sampling should follow described standard procedures. Standardization is of high importance to ensure comparability between sample results spatially and temporally. This report includes technical instructions on sampling procedures for collection, pre-treatment, shipment, and storage of Arctic environmental samples. The report also describes the registration of the samples.

For monitoring purposes, terrestrial and aquatic organisms, water, soil, sediments, and dust are often sampled. This guideline includes procedures for sampling of:

- Lichens: Crinkled snow lichen (*Flavocetraria nivalis*)
- Mussels: Blue mussels (*Mytilus edulis/Mytilus trossulus*)
- Seaweed: Bladderwrack (*Fucus vesiculosus, Fucus disticus, Ascophyllum nodosum*)
- Fish: Arctic char (*Salvenius alpinus*) and sculpins (*Myoxocephalus scorpius, Myoxocephalus quadricornis*)
- Water: Marine and freshwater
- Sediments: Marine and freshwater sediments.

For other types of samples, e.g., soil, prawns, crabs, marine and terrestrial mammals, birds, eggs, etc., DCE can be contacted for instructions at DCE-mining@dce.au.dk.

Contamination of samples must be avoided during collection, sample handling, and pre-treatment. As environmental samples are often collected along gradients around a contamination source, it is recommended (whenever possible) that samples are collected, pretreated etc. starting with the unpolluted samples first and following the gradient towards the pollution source to limit the risk of sample contamination.

It is important to note that if the samples are collected as part of a baseline study for a mining project in Greenland, duplicate samples must be collected, and one set of samples must be sent to DCE to be stored in the sample bank for future reference.

To develop an environmental monitoring program please see 'Environmental monitoring at mine sites in Greenland, - A review of research and monitoring practices and their role in minimizing environmental impact' (Søndergaard et al., 2020).

2 Registration of samples

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Figure 1. Orange ID number book and zip-lock plastic bags. The larger bag is used for the sample, while the smaller bag holds the sample ID number page. Photo by L. Bach.

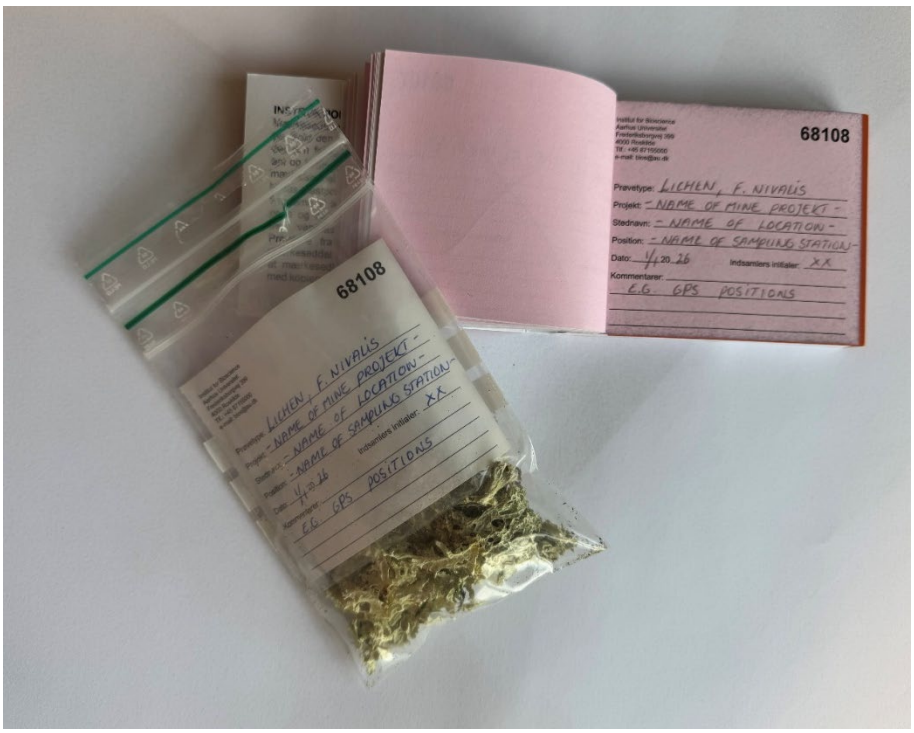

Figure 2. An example of a correctly packed sample, in this case a lichen sample. The smaller bag with the ID number page is placed inside the larger bag holding the sample to ensure the sample ID follows the sample. Remember also to write the ID number of the sample on the outside of the plastic bag with a permanent marker. Photo by L. Bach.

3 Required permissions

Permission to collect biological samples and/or samples containing soil, sediments and rocks in Greenland may be required prior to fieldwork in Greenland. The Greenland Authorities can provide details on the regulations in force.

4 Lichen sampling methodology

Lichens bioaccumulate atmospheric contaminants, such as metals, and are abundant in the Arctic (Søndergaard 2013; Søndergaard 2020). Their lack of roots, large surface area and long-life span enable lichens to effectively accumulate air contaminants and are therefore applied as a monitoring organism for air and dust pollution. The crinkled snow lichen, *Flavocetraria nivalis* is the preferred lichen species for monitoring purposes in Greenland (see Figure 3).

As the lifespan of *Flavocetraria nivalis* is several decades and due to a limited ability of lichens to excrete the bioaccumulated contaminants again, transplanted lichens have often been used as a supplement to, or instead of, resident lichens to assess the year-to-year variation in dust deposition (Søndergaard et al., 2013). Transplantation also makes it possible to monitor sites where no natural lichen occurs. Lichens to be transplanted are collected from uncontaminated reference sites as close to the monitoring site as possible. They are typically placed at the monitoring sites for one year.

Figure 3. To assess dust deposition of contaminants from Greenland sites the lichen Flavocetraria nivalis is frequently used. Photo by L. Bach.

4.1 Sampling equipment

- Tweezers (plastic or stainless steel; may be needed if the lichens are very dry).
- ID number book (provided by DCE) and pencil.
- Polyethylene zip lock plastic bags (10 x 15 cm), one per sample.
- Polyethylene zip lock plastic bags (10 x 10 cm) for ID number sheet, one per sample.

4.2 Sampling

- Each sampling station covers an area of approximately 15 x 15 meters with several lichen colonies.

- At each sampling station, collect from several lichen colonies (at least 4). The parts of the lichen embedded in the substrate should not be included. Soil, debris and other organic parts than the leaf-like branched lichen should be discarded.
- A sample size of approximately half a bag (10 x 15 cm) of fine sorted material is sufficient. No more than that should be collected to avoid unnecessary impact on the environment, to secure lichens for future environmental sampling and avoid unnecessary use of storage capacity.
- Use tweezers if relevant e.g., if the lichen is very dry and thus brittle.
- Store sample in a zip-lock bag (10 x 15 cm).
- Fill out the ID number page. Place the ID number page in a separate zip-lock bag (polyethylene 10 x 10 cm) to keep the ID number page dry.
- Place the sample in a 10 x 15 cm bag together with the ID bumber bag (see Figure 2), so that the ID number don't get wet and securely follows the sample. Write the ID number on the bag with the sample. The bag with the ID number sheet should be inserted in the sample bag so the ID number and other information are readable without opening the sample bag.
- Store the sample cold ca 5 °C. If lichens were sampled wet or damp then freeze at -18°C as soon as possible to keep the lichens from going mouldy.
- If the sample is a part of a baseline sampling, this is the level where DCE should receive it.

4.3 Sample registration and reporting

- Enter the data on lichen samples in the sheet named 'Basic' (see appendix A) in the Microsoft Excel workbook DCE_SampleReporting (TR374_Bilag.xlsx) provided by DCE. Besides filling out the standard fields in this sheet, be sure to make a note in the column "Comments" in case the lichens were transplanted.

4.4 Preparation for chemical analysis in laboratory

- Dry the samples at max 50°C or by freeze drying.
- Crush the sample to smaller pieces and homogenize.
- A representative subsample should be used for chemical analysis.

5 Mussel sampling methodology

Blue mussels are suspension feeders that filter large volumes of water through their gills (typically ca. 3 liters per hour for an adult mussel) and feed mainly on phytoplankton (Famme et al., 1986). The contaminant accumulation in mussels is considered as an integrated measure of the concentration and bioavailability of both contaminants bound to particles and contaminants dissolved in the seawater (Beyer et al., 2017; Rainbow 1995).

In Greenland, the blue mussel (*Mytilus* sp.) occurs in shallow waters along most of the coasts of west Greenland, and as far north as ~70°N on the east coast (see Figure 4). The species is therefore suitable for monitoring in nearshore waters. Blue mussels in Greenland consist of the two species *Mytilus edulis* and *Mytilus trossulus*. In general, *M. edulis* is distributed from the Disko Bay area and south as well as on the east-coast of Greenland, while *M. trossulus* is distributed from Disko Bay and northwards (Bach et al., 2019; Wenne et al., 2020). As the two species are indistinguishable by the naked eye, and the two species co-occur in some areas, the sampling should be done with no distinction between the two species.

If mussels are not natively present at the desired station for monitoring, they may be transplanted from an unpolluted area and then left at the desired station (Benedicto et al., 2011; Søndergaard et al., 2011) for e.g., one year before collection and analyses are conducted. The results will reflect last year's contamination impact in contrast to resident mussels that will reflect several years of contamination. Transplantation of mussels may be a solution, but be aware that exposed coasts (e.g., wind and waves) and coastlines with heavy influence by ice during winter may be difficult to monitor using this setup.

Figure 4. Blue mussel, *Mytilus* sp., bed at a rocky surface. Photo by L. Bach

Mussels for monitoring should in general be depurated prior to dissection. This is to facilitate the discharge of unassimilated particles in the mantle cavity or the gut that might significantly influence the metal concentration in the

sample. This is especially important for mussels collected in water with high turbidity or on silt/clay bottoms

An important note for projects where baseline samples have consisted of un-depurated mussels: For these projects, both un-depurated samples and depurated samples should be collected to enable comparison with previously collected samples. This 'duplicate' sampling is expected to cover a transition period of 3 years, where after only depurated mussel samples need to be included in the sampling program.

5.1 Sampling equipment

Sampling

- Survival suit or similar (waders etc.).
- Neoprene gloves or similar.
- Bucket or mesh bag for the collected mussels.
- Caliper or ruler.
- Buckets with lid (min 10 l) for collection of seawater for depuration of mussels (a bucket per sampling station).

Depuration

- Mesh bag (polyethylene, mesh size: ca 1 x 1 cm)
- Buckets with lids (min 10 l) for depuration of mussels. Buckets should be made of polypropylene and approved for food storage
- Air supply for depuration of mussels (pump, hose, air stone)

Dissection

- Scalpel (stainless steel) or small knife (preferable ceramic material)
- Tissue paper, dish drainer tray, or similar for drainage of mussels.
- Cup (holder for zip lock bags (10 x 15 cm)).
- Laboratory gloves (optional).
- ID number book (provided by DCE) and pencil.
- Polyethylene zip lock plastic bags (10 x 15 cm), one per sample.
- Polyethylene zip lock plastic bags (10 x 10 cm) for ID number sheet, one per sample.

In case the samples are to be analysed for PAHs, other oils, or organic contaminants, DCE should be contacted for proper sampling bags and methods.

5.2 Sampling

- Collect mussels at the same time every year. To avoid spawning season and to get a full growing season the preferred time for sampling is in August or early September.
- Sample at low tide. In Greenland, sampling can most often be done from the coast during low tide (check the local tide schedule at DMI (https://www.dmi.dk/hav-og-is/temaforside-tidevand/tidevandstabeller). Be aware that the timetables are given in Greenlandic standard time

(UTC-2 hours). When daylight saving time applies (last Sunday in March through last Sunday in October), the times stated must be increased by 1 hour.

- The samples at the different stations should be collected as close to the same depth and exposure (i.e., in terms of light and wave action) as possible to reduce variability in contaminant uptake.
- Each sampling station covers an area of approximately 20 meters along the coastline with several mussel colonies.
- Collection:
    - Baseline samples: Collect a mussel sample consisting of 20 individuals.
    - Existing mine sites, where baseline samples consisted of un-depurated mussels: Collect two mussel samples each consisting of 20 individuals. One sample for depuration and one sample for direct dissection, without depuration.
- Preferred mussel size is 4 - 5 cm. or alternatively 3-4 cm.
- The sampled mussels should be cleaned from sediment, gravel, etc. attached to the exterior of the shell.
- Store mussels moist and cool until depuration.
- Avoid collecting more mussels than necessary to reduce the impact on the environment, and to secure mussels for future environmental sampling.
- Collect seawater for depuration of the mussels. Collect the water at subsurface from the same site as the samples. If the water is visually impacted by mine activities or is silty, then collect sea water from a less impacted site. Store the sampled seawater at ambient temperature (preferably not in direct sunlight).
- Leave the collected seawater for 6 hours for particles to settle in a 10 L bucket with a loose lid at ambient temperature (preferably not in direct sunlight). Here after, the water is decanted to another bucket and the settled particles discarded.

An important note if mussels are to be transported over long distances, the live mussels can be placed in buckets, bags or coolers and should be kept moist – but not immersed in water. Moist conditions can be met by placing a wet cloth or seaweed from the collection site on top of the mussels. If mussels have opened during the transport and/or smell rotten, theses mussels must be discarded.

5.3 Depuration

- Depurate the mussels no later than 48 hours after collection.
- Depurate the mussel sample. One sample consisting of 20 individuals per bucket with ca. 10 L seawater.
- Tie a knot in the polyethylene mesh to create a net. Place the mussel sample in the net and close it with a knot at the other end (illustrated in Figure 5).
- Attach the net to the bucket in a way so the net hangs freely in the water above the bottom of the bucket.
- Aerate the water and place the bucket at ambient temperature (preferably not in direct sunlight).
- Depurate the mussels for 20-24 hours.

An important note: As mussels will not depurate in water low of oxygen, the water needs to be aerated. Use a plug-in pump; or alternatively a battery driven air pump.

Figure 5. An example of a setup for depuration of mussels. A sample of 20 mussels is placed in a net in the water column of the bucket for 20-24 hours. Air is supplied using a battery powered air pump. Photo by L. Bach.

5.4 Dissection

- Measure the shell length of each mussel in a sample consisting of 20 individuals with a caliper for each individual and note the lengths in the excel sheet 'Shellfish'.
- Cut open the mussel by cutting the adducter muscle of the mussel, and twist the scalpel. That will open the mussel. Let it drain on tissue paper, dish drainer tray, or similar for 5 minutes (see Figure 6). The mussels should be placed inverted so excess water can drain.
- Using a stainless steel scalpel or a knife, cut off the byssus threads that must be discarded from the sample.
- Using a stainless steel scalpel or a knife, sample all soft parts.
- Fill out the ID number page. Place the ID number page in a separate zip-lock bag (polyethylene 10 x 10 cm) to keep the ID number page dry.

- Place all soft mussel parts from the 20 sampled mussels in a zip-lock bag (10 x 15 cm). Place the ID number bag inside the 10 x 15 cm bag together with the sample, so that the ID number securely follows the sample. Write the ID number on the sample bag (10 x 15 cm). The bag with the ID number sheet should be inserted in the sample bag so the ID number and other information is readable without opening the sample bag.
- Store the sample cold ca 5 °C until it can be frozen at -18°C.
- If the sample is a part of a baseline sampling, this is the level where DCE should receive it.

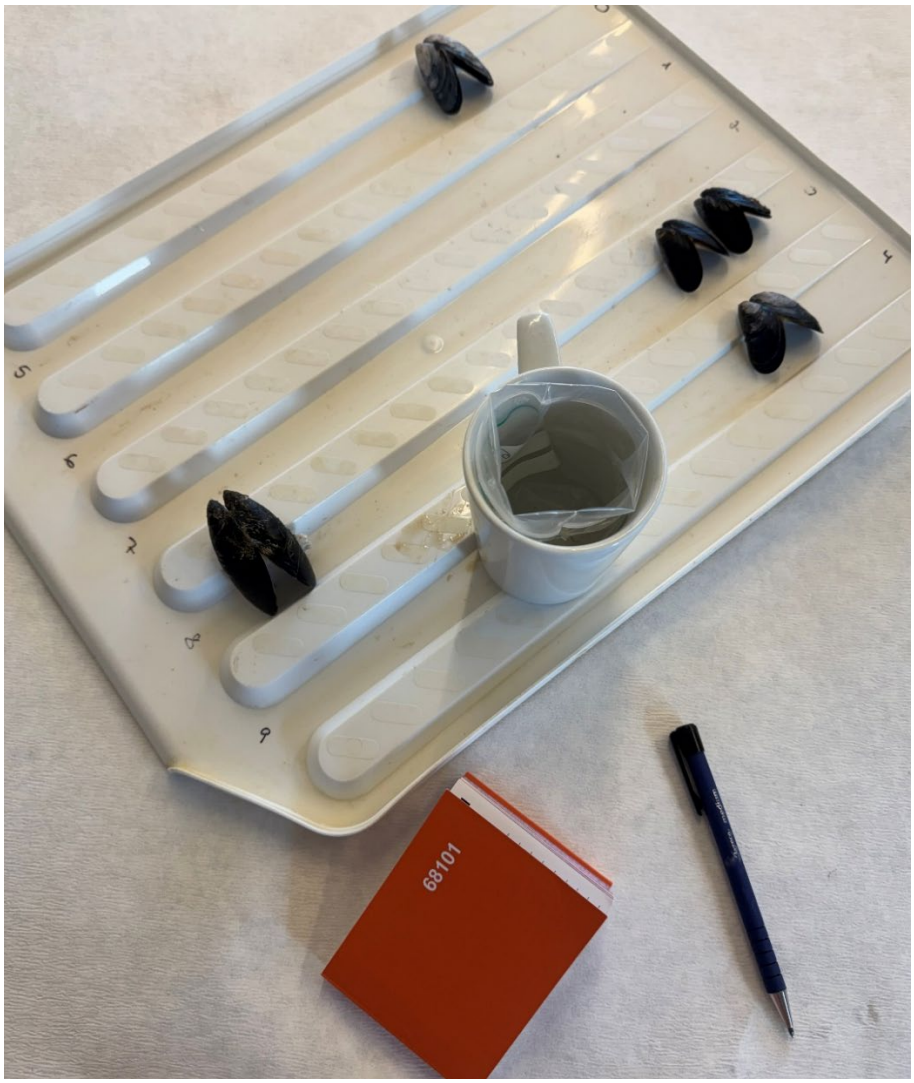

Figure 6. Blue mussels left to drain on a dish drainer tray before dissection of the soft meat. Photo by L. Bach

5.5 Sample registration and reporting

- Enter the data on mussel samples in the sheets 'Basic' (see appendix A) and 'Shellfish' (see appendix B) in the Microsoft Excel workbook DCE_SampleReporting (TR374_Bilag.xlsx) provided by DCE.
- Besides filling out the standard fields in these sheets, be sure to make a note in the column 'Comments' in 'Basic' in case the mussels were transplanted. The habitat of the sampling site should be noted under 'Comments' in 'Basic', e.g., mussels collected on rocky shore, sandy beach, mud, etc.

- In DCE_SampleReporting.xlsx, there is a field recording sheet in Danish called 'LegacyFieldSheet_BlueMussels_DK', which can be printed and used, when processing the mussel. However, for the data reporting to DCE, all information needs to be transferred to the sheets 'Basic' and 'Shellfish'.

5.6 Preparation for chemical analysis in laboratory

- Freeze dry the sample.
- Pulverize and homogenize the sample.
- A representative subsample should be used for chemical analysis.

6 Seaweed sampling methodology

In Greenland, three brown seaweed species dominate i.e., *Fucus vesiculosus* (Bladder wrack), *Fucus disticus* (Two-headed wrack or Common rock weed) and *Ascophyllum nodosum* (Knotted wrack). Sampling of *F. vesiculosus* should be preferred over *F. distichus*, which is preferred over *A. nodosum*. For identification of *F. vesiculosus* see Figure 7, where the typical characters of *F. vesiculosus* with two parallel bladders are shown.

Figure 7. Photos of the three species, A) *F. vesiculosus*, B) *F. disticus* and C) *A. nodosum* for identification. Photos by O. Geertz-Hansen.

Growth tips of seaweed are used for monitoring purposes, as accumulation of metals in seaweed is regarded as a relative measure of the contaminant concentrations dissolved in the seawater (Rainbow, 1995). The growth tip contaminant concentrations reflect the accumulation in the present growing season (spring-autumn).

Seaweed should be collected during August or September to be able to retrieve the new growth tips (se Figure 8). The new growth tips are usually more green and is the part seperated by the most outer branch. Sampling of *F. vesiculosus, F. disticus* and *A. nodosum* is most often possible from land during low tide.

6.1 Sampling equipment

- A pair of scissors (stainless steel)
- Food strainer (plastic or stainless steel)
- Demineralized water (or alternatively tap/fresh water)
- Survival suit or similar (waders etc.)
- ID number book (provided by DCE) and pencil.
- Polyethylene zip lock plastic bags (10 x 15 cm), one per sample.
- Polyethylene zip lock plastic bags (10 x 10 cm) for ID number sheet, one per sample.

In case the samples are to be analysed for PAHs, other oils, or organic contaminants, DCE should be contacted for proper sampling bags and methods.

6.2 Sampling

- Collect seaweed at the same time every year.
- Sample at low tide. In Greenland, sampling can most often be done from the coast during low tide (check the local tide schedule at DMI (https://www.dmi.dk/hav-og-is/temaforside-

tidevand/tidevandstabeller). Be aware that the timetables are given in Greenlandic standard time (UTC-2 hours). When daylight saving time applies (last Sunday in March through last Sunday in October), the times stated must be increased by 1 hour.

- Collect from at least 4 plants covering an area of approximately 20 meters along the coast.
- Avoid collecting more seaweed than necessary to reduce the impact on the environment, and to secure seaweed for future environmental sampling.
- Cut the annual fresh growth tips of the seaweed with a pair of scissors (stainless steel) into a food strainer (plastic or stainless steel).
- An amount of approximately 100 g of growth tips is needed per sample (about two handfulls).
- Rinse the growth tips of the seaweed 3 times in demineralised/fresh water (or alternatively tap water) to discard gravel and small organisms.
- Fill out the ID number page. Place the ID number page in a separate zip-lock bag (polyethylene 10 x 10 cm).
- Store the sample in a zip-lock bag (10 x 15 cm) together with the ID number bag, so that the ID number securely follows the sample. Write the ID number on the bag with the sample. The bag with the ID number sheet should be inserted in the sample bag so the ID number and other information are readable without opening the sample bag.
- Store the sample cold ca 5 °C until it can be frozen at -18°C.
- If the sample is a part of a baseline sampling, this is the level where DCE should receive it.

Figure 8. The annual fresh growth tips of the seaweed are used as a proxy for the year-to-year variation in dissolved contaminants. Photo by O. Geertz-Hansen.

6.3 Sample registration and reporting

- Enter the data on seaweed samples in the sheet named 'Basic' (see appendix A) in the Microsoft Excel workbook DCE_SampleReporting (TR374_Bilag.xlsx) provided by DCE.

6.4 Preparation for chemical analysis in laboratory

- Freeze dry the sample.
- Pulverize and homogenize the sample.
- A representative sample should be used for chemical analysis.

7 Fish sampling methodology

Fish species such as sculpin and Arctic char are often used to monitor the marine and freshwater environment, respectively.

Sculpins (*Myoxocephalus* spp.) have been the preferred marine fish species used in monitoring at Greenland mine sites because they are the most common of the sedentary fish species and abundant in both west and east Greenland. The shorthorn sculpin (*Myoxocephalus scorpius*) and fourhorn sculpin (*Myoxocephalus quadricornis*) are the most abundant sculpin species in Greenland.

Figure 9. Shorthorn sculpins, *Myoxocephalus scorpius*, are easily caught in most fjords in Greenland. As it is relatively sedentary, it is considered an indicator species for higher trophic level exposure. Photo by L. Bach.

Arctic char (*Salvelinus alpinus*) is a widespread fish in the Arctic and populates almost every lake and river in Greenland as either anadromous or landlocked populations. While landlocked fish stay in the freshwater system, the migrating fish will seek towards marine waters in May/June to feed, whereafter they will return to the freshwater systems in July/September. Here they will stay, usually within approximately 20 km of the system, where they overwinter (Muus et al., 1990). It can be difficult to identify whether a caught fish is a landlocked or migrating fish based on morphology. While landlocked fish are most often found in lakes, migrating fish are dominant in rivers. In general, the two forms can be distinguished by morphological divergences related to swimming performance and maneuverability. The landlocked Arctic chars are generally smaller, have a relatively larger head, smaller body, and longer caudal fin compared to migrating char (Damsgård 1991).

Figure 10. Arctic chars are caught in rivers, often in river pools. Photo by L. Bach.

To apply fish as environmental monitoring organisms, analyses of the liver are often recommended, as it has an important role in contaminant storage, redistribution, detoxification, or transformation, and it is an important site of pathological effects induced by contaminants (Ewans et al., 1993). Muscle tissue is also often collected and is considered a proxy for a more recent uptake/accumulation of pollutants than the liver (Hansson et al., 2020). Muscle tissue is also preferred for studying transfer of pollutants in the food chain and thus the health aspects of human consumption. As the concentration of pollutants may be different between males and females due to the transfer of some substances to roe during spawning, it is important that the sex is determined and noted. Both Arctic char and sculpin, males are usually smaller. Thus females are preferred to achieve that largest liver.

7.1 Sampling equipment

- Fishing gear relevant for the species in question.
- Scalpel (stainless steel) or small knife (preferable ceramic material) or a pair of stainless steel scissors.
- Poultry shears to cut up the fish head for collection of otolites.
- Cutting board.
- Kitchen scale sensitive to at least one decimal (used for total fish and liver weights).
- Ruler.
- Demineralized water (or alternatively tap water).
- Laboratory gloves (optional).
- ID number book (provided by DCE) and pencil.
- Polyethylene zip lock plastic bags (10 x 15 cm), one per sample.
- Polyethylene zip lock plastic bags (10 x 10 cm) for ID number sheet, one per sample.
- Polyethylene zip lock plastic bags (7 x 10 cm or smaller) or a small paperbag, one per sample.

- In case the samples are to be analysed for PAHs, other oils, or organic contaminants, DCE should be contacted for proper sampling bags and methods.

7.2 Sampling

- Collect 4 fish – preferably female to obtain the largest livers.
    - Fish, including Arctic char and sculpin, can most often be collected by angling, but may also be caught by fine meshed nets at low water depths.
    - Sculpins are most easily taken with rising waters in the tide scheme (check the local tide schedule at DMI (https://www.dmi.dk/hav-og-is/temaforside-tidevand/tidevandstabeller). Be aware that the timetables are given in Greenlandic standard time (UTC-2 hours). When daylight saving time applies (last Sunday in March through last Sunday in October), the times stated must be increased by 1 hour.
- Collect the fish at the same time every year outside of spawning season.
- For ethical reasons, the fish must be killed with a blunt instrument or a knife through the head immediately after capture.
- Avoid collecting more fish than necessary to reduce the impact on the environment, and to secure fish for future environmental sampling.

7.3 Dissection

- Process the fish right after collection. Alternatively, the fish should be stored cold until processing and for maximum of 12 hours.
- Identify Arctic char form or sculpin species.
- Note the total weight (to nearest gram).
- Note the total length (fork length to nearest cm or 5 mm level).
- Cut open the fish from the vent towards the head using a scalpel or a pair of stainless steel scissors.
- Note stomach content.
- Note the sex of the fish.
- Cut out the liver (see example for sculpin in Figure 11).
- Rinse the liver in demineralized water (or alternatively tap water).
- Note the total liver weight (to nearest 0.1 g).
- Place the liver into a zip-lock bag (7 x 10 cm).
- Cut out a piece of a muscle from the side of the fish behind the dorsal fin (ca. 4 x 2 cm) using a scalpel.
- Rinse the muscle in demineralized water (or alternatively tap water).
- Place the muscle into a zip-lock bag (7 x 10 cm).
- Open the head from the dorsal side behind the eyes and collect the otoliths (see example for sculpin in Figure 12)
- Place the otoliths into a zip-lock bag (polyethylene 7 x 10 cm or smaller) or a small paper bag.
- Fill out the ID number page. Place the ID number page in a separate zip-lock bag (polyethylene 10 x 10 cm) to keep the ID number page dry.

- Place all bagged samples (liver, muscle and otoliths) in a zip-lock bag (10 x 15 cm together with the ID number bag, so that the ID number securely follows the sample. Write the ID number on the bag with the sample. The bag with the ID number sheet should be inserted in the sample bag so the ID number and other information are readable without opening the sample bag.
- Store the sample cold at 5°C until it can be frozen at -18°C.
- If the sample is a part of a baseline sampling, this is the level where DCE should receive it.

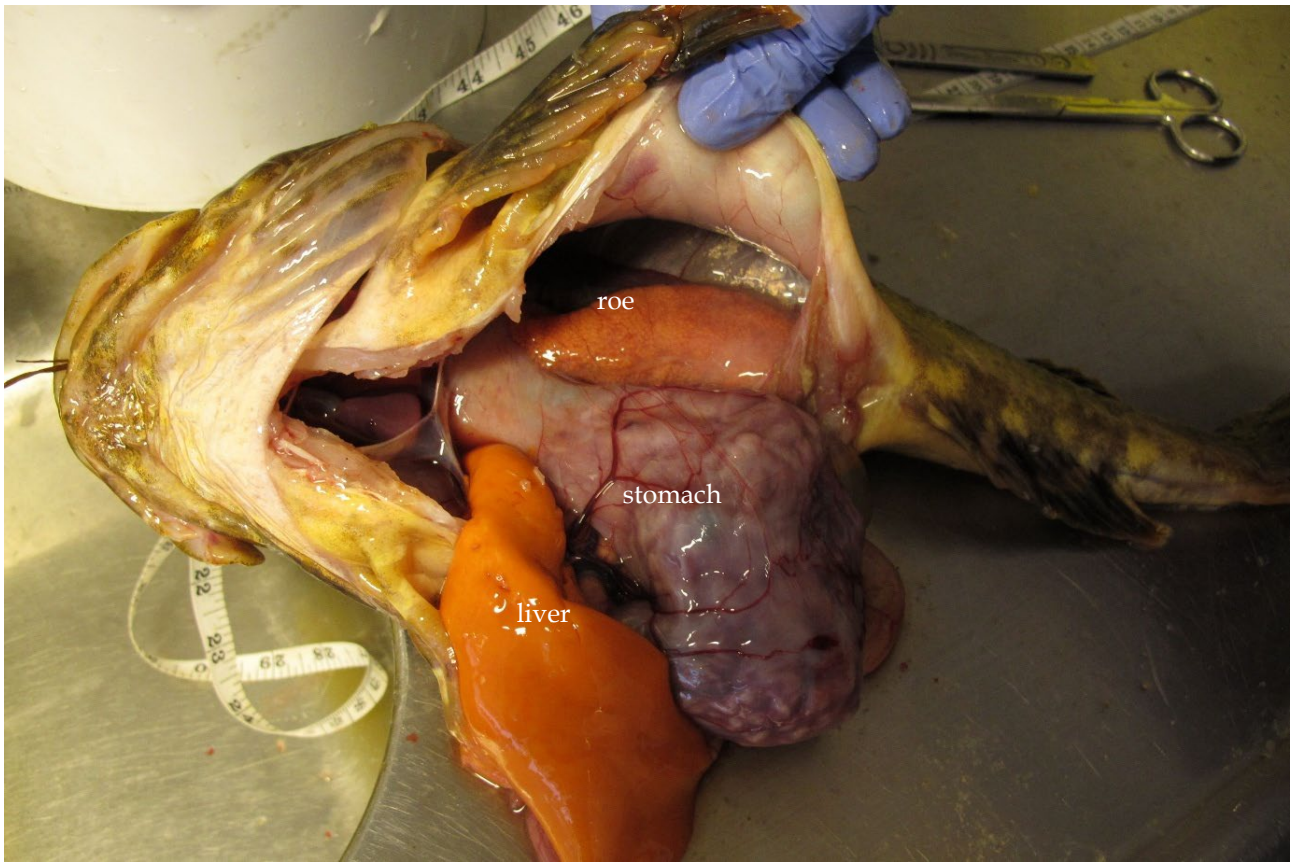

Figure 11. Dissection of a sculpin with indication of the liver. Here the liver appears orange, but it may also be more pale pink. Photo by L. Bach.

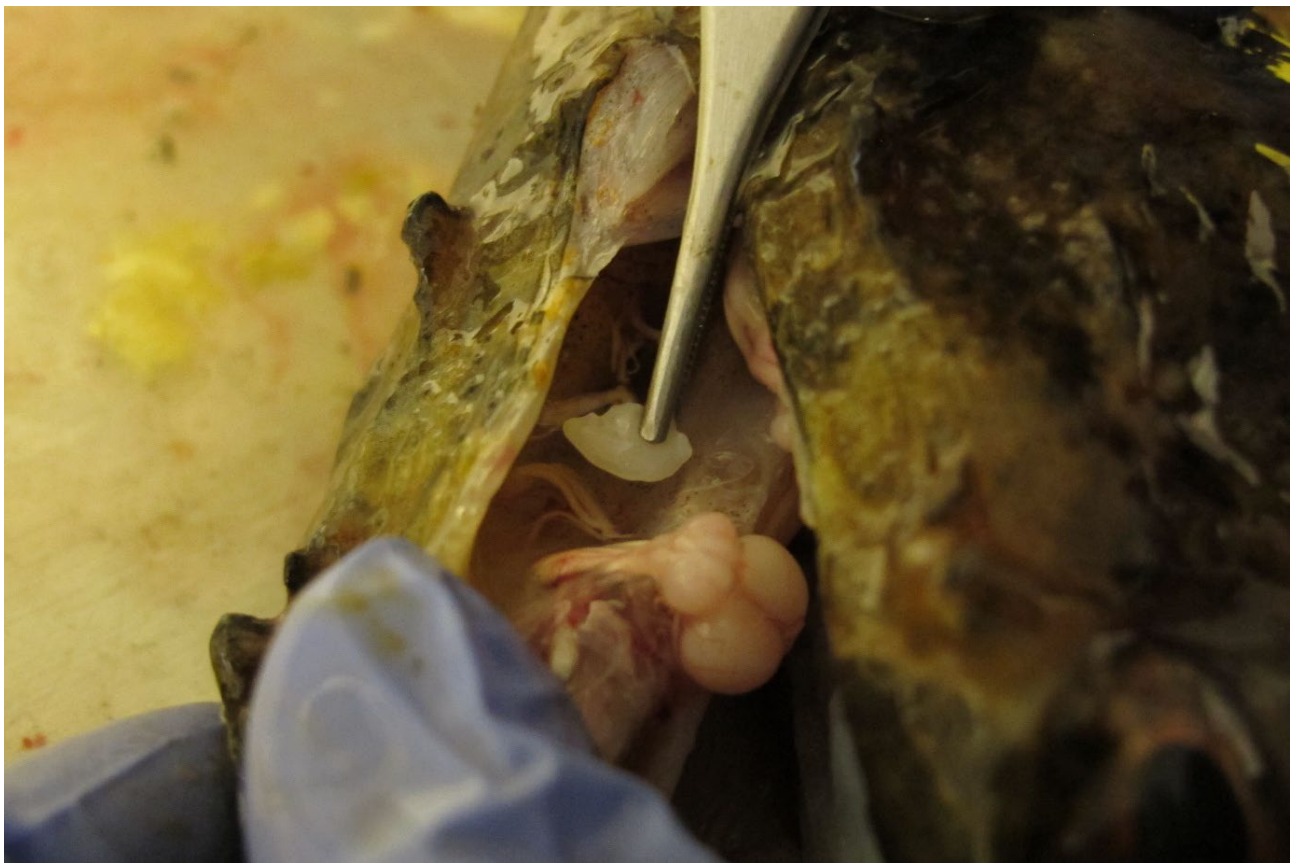

Figure 12. Dissection of a sculpin to collect the otolites. A cut is made along the dorsal direction with a poultry shears And the otolites can be found in two smaller holes on each side. Photo by L. Bach.

7.4 Sample registration and reporting

- Enter the data on fish samples in the sheets 'Basic' (see appendix A) and 'Fish' (see appendix C) in the Microsoft Excel workbook DCE_SampleReporting (TR374_Bilag.xlsx) provided by DCE.
- In DCE_SampleReporting.xlsx, there is a field recording sheet in Danish called 'LegacyFieldSheet_Fish_DK', which can be printed and used, when processing the fish. However, for the data reporting to DCE, all information needs to be transferred to the sheets 'Basic' and 'Fish'.

7.5 Preparation for chemical analysis in laboratory

- Freeze dry the sample (liver and muscle).
- Pulverize and homogenize the sample.
- A representative subsample should be used for chemical analysis.

8 Water sampling methodology

Water samples are often collected as a part of monitoring programs, both from marine as well as freshwater environments such as streams, rivers, and lakes. Generally, both unfiltered and filtered (0.45 µm) samples are collected to provide information on total and dissolved concentrations of contaminants, respectively. In addition to water sampling for elemental analyses, measurement of pH, conductivity and total suspended solids are often conducted. Also, the concentrations of suspended particulate-bound pollutants in the water are often quantified and elements determined by analysing the solid residue from a filtration process (Loring and Asmund, 1989; Søndergaard et al., 2011).

Passive chemical samplers, i.e., Diffusive Gradients in Thin films, (DGT) have been used in addition to conventional techniques for measuring dissolved metals in both freshwater and seawater (Søndergaard et al., 2014). DGT samplers have the advantage that they provide a measure of the time-integrated and 'labile' metal concentrations during the deployment period as opposed to conventional water sampling that only provides a snapshot of the water chemistry. DGT samplers, however, only work for some metals depending on the type of device. However, water quality criteria guidelines are typically only established for total metal concentrations in sea- and freshwater (MRA, 2015). Thus, when comparing data to water quality criteria guidelines, conventional water sampling is required. Consequently, DGT samplers should be regarded as a "supplement to" rather than a substitute for the conventional water sampling.

Figure 13. Fresh water sampling. First a clean 1 L bottle is used to take a depth-integrated sample in the stream. Then a syringe is used to take subsamples of unfiltered and filtered water into sample vials, the latter using disposable 0.45 µm syringe filters. Photo by O. Geertz-Hansen.

The exact protocol for sampling of seawater and freshwater is very laboratory dependent as it depends on the specific analytical methods etc. Please consult the laboratory that will be analysing the samples for instructions. For reference, the DCE water sampling protocol is given below.

8.1 Sampling equipment

- For sea or lake water, a water sampler (like the Ruttner-type sampler or Niskin) along with cord and messenger load. The sampler should be acid-washed prior to sampling. Stream or river samples are most often collected directly by hand in the sampling bottles/test tubes.
- Laboratory gloves.
- A 1 or 2 L clean (acid-washed) polyethylene sampling bottle.
- Disposable polypropylene/polyethylene syringes (without rubber O-ring, which is prone to zinc carry-over).
- Syringe filters (nylon or PES, 0.45 µm pore size).
    - Sample vials/bottles:
    - For trace metal analyses in freshwater: typically, 15 ml polypropylene ICP-MS vials.
- For trace metal analyses in seawater: typically, 100 ml or larger polyethylene bottles (acid-washed).
- ID number book (provided by DCE).
- Water resistant pen.
- Polyethylene zip lock plastic bags, one per sample.
- Polyethylene zip lock plastic bags (10 x 10 cm) for ID number sheet, one per sample.

8.2 Sampling

- Use laboratory gloves
- Rinse the large sampling bottle 3 times with water similar to the water that will be sampled.
- Collect the water sample in the large sampling bottle:
    - In the sea and in lakes, depth-specific sampling is usually conducted using a water sampler equipped with a messenger load released when the sampler reaches the desired depth. Once the sampler is retrieved to the surface, transfer the water to the large sampling bottle.
    - In a freshwater stream it is usually preferred to obtain a depth-integrated sample by moving the sampling bottle up and down through the water column until it is filled. Avoid surface water which can contain debris like insects, leaves etc. Also avoid kicking up and suspending sediment from the bottom of the stream, which can contribute to an elevated particle concentration in the sample (see Figure 13 for an example of collection in a river).
- Unpack a syringe and rinse it 3 times with the water from the sampling bottle (i.e., fill and empty the syringe 3 times and discard the water).
- Fill the syringe and take an unfiltered sample by transferring 15 ml (freshwater) or 100 ml (seawater) to a vial/bottle.
- Unpack a filter, fill the syringe, put the filter on and rinse the filter by pressing the volume of water in the syringe though the filter once and discard the water.
- Fill the syringe, put the filter on again and press the water slowly into the sample vial/bottle to collect a filtered sample. It is often an advantage to press the filter against the edge of the vial/bottle when the water is pressed through it to avoid the filter detaching from the syringe under pressure. If

the water contains high concentrations of particles, it may be necessary to use more than one filter per sample. In that case, remember to also rinse new filters before use. Fill the sample bottle entirely.

- It is usually preferred to take duplicate samples such as two unfiltered and two filtered samples (using two separate filters) at each location.
- Fill out the ID number page. Place the ID number page in a separate zip-lock bag (polyethylene 10 x 10 cm) to keep the ID number page dry.
- Place the water samples in a zip-lock bag (10 x 15 cm together with the ID number bag, so that the ID number securely follows the sample. Write the ID number on the bag with the sample. The bag with the ID number sheet should be inserted in the sample bag so the ID number and other information are readable without opening the sample bag.
- Store the samples cold at 5°C if possible until they reach the lab. **Do not freeze the samples** as the lid may break and metals may precipitate during the freezing process.
- It is necessary to take into account that water samples need to be analysed within 2 years after collection to be valid.
- If the sample is a part of a baseline sampling, this is the level where DCE should receive it.

8.3 Sample registration and reporting

- Enter the data on water in the sheet named 'Basic' (see appendix A) in the Microsoft Excel workbook DCE_SampleReporting (TR374_Bilag.xlsx) provided by DCE.

8.4 Preparation for chemical analysis in laboratory

- Once the samples are received in the lab, they will be preserved with clean acid prior to analysis.
- Store the samples cold at 5°C.
- Note that water samples needs to be analysed within 2 years after collection to be valid. It is not adequate to store environmental water samples for a longer period (Grasshoff et al., 1999).
- When received, DCE preserves the samples using nitric acid.

9 Sediment sampling methodology

Many environmentally harmful substances have a high affinity for particles to which they can adsorb/absorb and thereby settle out of the water phase. The sediment can re-suspend and gradually be transported by currents to a final sedimentation area. The sediment acts as a reservoir for a large group of environmentally harmful substances discharged into the marine or freshwater environment and is therefore a good proxy for monitoring pollution (temporal and/or geographical). In accumulation areas, both the current and previous pollution problems can be investigated by sampling and analysing segments of a sediment core. A sediment sample always represents a time period whose length depends on natural conditions such as sedimentation rate and bioturbation e.g., animals digging in the sediment in the area.

Sediment samples are collected at fixed locations (stations) in the selected areas with a sampler capable of extracting a core of sediment. Parameters that are important for the assessment of the result (e.g., information about the sediment such as color, layers, indication of bioturbations, and appearance of the column) are noted. It is advised to use sediment core samplers. Using sediment core samplers, it can be ensured that the sampling method used does not mix the surface sediment (most recent deposited) with the underlying sediment (older depositions), as can occur by using different types of grabs (Xu et al., 2011). Sediment core samplers can be a HAPS-core sampler, a Kayak core sampler or others. Alternatively, surface samplers like the Ekman sampler for soft sediments, or a van Veen grab sampler for soft or medium-hard bottoms like sand, gravel, consolidated marl or clay, can be used if only the upper few centimeters of sediment are to be sampled (as a proxy of the recent deposition).

Figure 14. Sediment sampling of bottom surface sediment using an Ekman grab sampler. A slice of bottom surface sediment of 1-3 cm thickness can be obtained. Photos by J. Søndergaard.

Marine and freshwater sediment samples should as far as possible be collected in sedimentation areas. In marine areas, the sampling station should be > 50 m from point sources and the coast to avoid direct impact from land.

In freshwater streams, rivers and lakes, surface sediment samples should be taken in an area with low or no current, where transported particulate matter typically accumulates (sedimentation area). It is recommended to search the watercourse around the defined sampling station for the most suitable places to collect a sufficient amount of sediment.

To acquire knowledge on deposition in time intervals, a sediment core can be collected. Often the core is divided into slices of 1-2 cm but with shorter intervals at the top i.e., the uppermost 5 cm is often divided into 0.5-1 cm slices.

9.1 Sampling equipment

- Sediment sampler (spoon, grab, corer, haps etc.).
- Spoon (stainless steel).
- ID number book (provided by DCE) and pencil.
- Polyethylene zip lock plastic bags (10 x 15 cm), one per sample.
- Polyethylene zip lock plastic bags (10 x 10 cm) for ID number sheet, one per sample.

9.2 Sampling

- Collect the sample upstream from the sampler to prevent contamination by suspended sediment disturbed by the person.
- To assess the most recent contributions of contaminants, the upper section (or part of) of the sample (1-2 cm) should be carefully collected. It is important that only the fine sediment deposited is collected. This will be light and with a loose structure.
- To acquire knowledge on deposition in time intervals, a sediment core can be collected. Often the core is divided into slices of 1-2 cm but with shorter intervals at the top i.e., the uppermost 5 cm is often divided into 0.5-1 cm slices.
- Collect three subsamples of sediments to be pooled into one sample to minimize the variability and impact of heterogeneous sediments:
- Sampling of:

  River or stream sediments with hard bedrock:

  - Use a spoon to scrape off the uppermost 2 cm. In freshwater systems, avoid getting coarse-grained gravel included in the samples.

  Marine or lake sediments:

  - Use a Van Veen grab, Eckmann grab or a similar sampler.
  - When the sediment sampler is retrieved, use a spoon to scrape of the uppermost 2 cm directly from the sampler.

  Sediment core:

  - Use a Kayak pipe sampler or other core sampler.
  - After sampling, let the surface sediment settle within the pipe.
  - Decant the overlying water. This can preferably be done by using a tube as this method allows for a more sensitive separation of water and sediment.
  - Slice the sediment core carefully into preferred sizes. For a 'recent' baseline sample it is recommended to collect the upper 2 cm. If the

sediment is very loose, the upper 1-2 cm can alternatively be transferred from the pipe with a tube or pipette.

- Fill out the ID number page. Place the ID number page in a separate zip-lock bag (polyethylene 10 x 10 cm) to keep the ID number page dry.
- Place the sediment sample (a pool of three subsamples per sediment layer) in a zip-lock bag (10 x 15 cm). Assign the sample with a unique ID number and store it in a separate zip-lock bag (10 x 10 cm). Place the ID number bag inside the 10 x 15 cm bag together with the samples, so that the ID number securely follows the sample. Write the ID number on the 10 x 15 cm sample bag. The bag with the ID number sheet should be inserted in the sample bag so the ID number and other information are readable without opening the sample bag.
- For sediment core samples, each sediment core is assigned a unique ID number, and thus all slices from the same core have the same ID number. Each section of sediment (i.e., 0-1 cm, 1-2 cm, 2-4 cm, 4-6 cm etc.) is stored in separate zip-lock bags (polyethylene) marked with the ID number and the depth of the slice. All slices are stored in one larger bag together with the ID number pad. Write the ID number on the outside of the larger bag. The bag with the ID number sheet should be inserted in the sample bag so the ID number and other information are readable without opening the sample bag.
- The sediment sample collected must be corresponding to a minimum of 75 g dry matter (approximately 200 g wet weight).
- The samples should be stored cold until they can be frozen at -18°C.
- If the sample is a part of a baseline sampling, this is the level where DCE should receive it.

9.3 Sample registration and reporting

- Enter the data on sediment samples in the sheets 'Basic' (see appendix A) and 'Sediment' (see appendix D) in the Microsoft Excel workbook DCE_SampleReporting (TR374_Bilag.xlsx) provided by DCE.
- Variables such as water depth, distance from land, colour, bioturbation, smell, grain size, etc. that can have impact on the data assessment should be documented in the field 'Comments' in the sheet 'Sediment'. Also be sure to note in this field, if there are signs of disrupted stratigraphy when the core is visually assessed (e.g., signs of bioturbation).
- A photo of the core should be taken to document the layering and its orientation relative to the slices prepared. These photos should be delivered to DCE along with the Excel file (to be downloaded via the Data Sheet).

9.4 Preparation for chemical analysis in laboratory

- Dry/freeze dry the sample.
- Homogenize the sample.
- A representative subsample should be used for chemical analysis.

10 Procedure for handing over samples and data to DCE

DCE stores environmental samples and data from mineral projects on behalf of the Environmental Agency for Mineral Resources Activities (EAMRA). The purpose of this chapter is to provide mining companies and their environmental consultants with a clear, transparent description of DCE's procedure for reception, analysis and storage of environmental samples and data in DCE's sample archive and in the Mineral Resources Environmental Database (MRED).

Procedures are given for three categories of samples and data:

- Baseline samples collected by mining companies and their consultants as part of their baseline studies and Environmental Impact Assessment (EIA) work.
- Environmental samples collected by the mining companies and their consultants during the mining phase as part of their compliance control program.
- Environmental samples collected during the mining phase and after mine closure as part of the Environmental Authorities' environmental monitoring program.

10.1 Procedure for baseline samples and data

The procedure for reception, analyses and storage of baseline samples and data collected by mining companies and their consultants as part of their baseline studies and/or Environmental Impact Assessment (EIA) work is as follows:

- Before any baseline samples are collected, the baseline sampling program needs to be approved by EAMRA. Otherwise the samples are not valid as baseline samples.
- Prior to sampling, the mining company contacts DCE at DCE-mining@dce.au.dk to receive sample ID number books.
- After sampling, the mining company delivers one set of samples to DCE for storage and keeps another duplicate set of samples for their own storage and subsequent analyses. Duplicate samples can e.g., be fish liver samples split into two equally sized parts or replicate samples of seaweed, mussels and lichens from the same date and station. If duplicate samples are not available and only one sample set exists, DCE stores the samples. Both samples stored by DCE, and duplicate samples stored by the mining company, must be numbered using the DCE ID number books.
- Prior to the delivery, the mining company sends the completed Microsoft Excel workbook DCE_SampleReporting (TR374_Bilag.xlsx) to DCE with information on the samples. It needs to include information both on samples to be stored at DCE and duplicate samples to be stored by the mining company (recorded in the field 'Storage' in the sheet 'Basic'). DCE cannot receive any samples before DCE_SampleReporting.xlsx has sent to DCE-mining@dce.au.dk, where the delivery of the physical is also arranged.

- During the delivery of the physical samples, used up ID number books are also handed over to DCE.
- DCE archives the ID number books, performs quality control of the sample information in DCE_SampleReporting.xlsx, and formats and adds the data to the MRED database. Before storage in freezer or dry store, DCE also performs a check that the received samples match what is listed as 'Storage=DCE' in the Excel file. Upon completion of these tasks, DCE reports back to the company via EAMRA on reception of data and samples, shortly listing what has been received and highlighting insufficiencies if such were encountered.
- *For water samples only*: DCE notifies EAMRA that the samples have been received and are stored in the sample bank (refrigerated). Prior to storage, DCE preserve the samples with nitric acid. Due to the limited chemical stability of water samples, analyses should be performed as soon as possible and not later than 2 years after collection data, given that the samples have been properly acid preserved (Grasshoff et al., 1999). After 2 years of storage, and if nothing else is agreed, DCE will contact EAMRA to discuss whether the samples can be discarded.
- *For all other samples than water samples*: DCE stores the samples in the sample bank (freezers or dry storage) until further instructions from EAMRA. In case EAMRA finds it relevant to analyze the samples, or just a selection of the samples, for control, this can be done at any time.
- The company decides if and when chemical analyses shall be made, often as a step in the preparation of an EIA report. It can also be upon request from EAMRA. If the company moves on with chemical analyses of the samples, the laboratory performing the analyses should be accredited to do the chemical analyses and the lab's detection limits for environmentally important elements should comply with the Danish Environmental Protection Agency's current requirements for environmental measurements (in Danish: "Bekendtgørelse om kvalitetskrav til miljømålinger", available at www.retsinformation.dk). Analyses can be made on the duplicate sample set stored by the company. If duplicate samples are not available, DCE can send a subsample to the mining company for a fee covering DCE's expenses for sample preparation (handled through EAMRA). For seaweed and mussel samples, the sample preparation will involve freeze-drying and homogenizing of the samples to provide a representative subsample.
- The company sends analysis results from baseline samples to DCE for inclusion in the MRED database via DCE_SampleReporting.xlsx. Currently, DCE_SampleReporting.xlsx contains a sheet called 'Multielementdata', intended for reporting Inductively Coupled Plasma Mass Spectrometry (ICP-MS) data (see Appendix E for example data and instructions). If other types of analyses are conducted, DCE should be contacted to receive an appropriate template for data reporting. Reports from the laboratory performing the analysis should also be sent to DCE.
- DCE adds the analysis results to the MRED database. DCE also performs a screening of the results to evaluate the quality of the data. In case concerns arise about the analytic results or lacking information, DCE notifies EAMRA. If EAMRA wants to check the quality of results from the other laboratory, DCE can analyze samples for control.
- Through EAMRA, all mineral license holders receive an annual status on the samples and analysis results in the MRED database on their license. On request through EAMRA, data can also be extracted from the MRED database but may be subject to a fee covering DCE labor costs.

10.2 Procedure for company compliance control samples and data

Unless specifically requested by EAMRA, samples from the mining phase collected as part of the Mining Companies' Compliance Control Program are not included in the DCE's sample archive and shall not be sent to DCE. DCE ID books should not be used to attribute ID numbers to these samples. Analytical results from the samples from the Mining Companies' Compliance Control Program shall be sent to EAMRA, which may be forwarded them to DCE for evaluation and/or filing, but the data will not be included in the MRED database.

10.3 Procedure for the authorities environmental monitoring samples and data

During the mining phase and after mine closure samples are collected at regular intervals by DCE/GINR on behalf of EAMRA as part of the Authorities' Environmental Monitoring Program. The procedure for reception, analyses and storage of samples and data from the Authorities' Environmental Monitoring Program (in DCE's sample archive and MRED database) are exactly the same as outlined Section 10.1, but with the following modifications:

- DCE/GINR are the collectors of data and samples, and DCE is the receiver of data and samples, both roles on behalf of EAMRA.
- *For water samples only*: Prior to storage, DCE preserve the samples with nitric acid.
- EAMRA decides if and when the chemical analyses shall be made.
- The DCE lab performs the chemical analyses.
- DCE adds the chemical analysis results to the MRED database.
- DCE prepares an analyses report to EAMRA, including an interpretation of the results as per agreement with EAMRA.
- Samples and analysis results from the Authorities' Environmental Monitoring Program are included in EAMRA's annual status to the mineral license holders on the samples and analysis results on their license. On request through EAMRA, data can also be extracted from the MRED database but may be subject to a fee covering DCE labor costs.

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Appendix A – Basic registration for all sample types

For all samples collected, regardless of type, information from the ID number book must be entered in the sheet named 'Basic' in the Microsoft Excel workbook DCE_SampleReporting (TR374_Bilag.xlsx) provided by DCE. The sheet contains help text explaining how to fill out the different fields, and in some cases field values are constrained to certain predefined codes. All fields need to be filled out. In case of uncertainty regarding the registration, please contact DCE.

Hypothetical example of data recorded in the Basic table

ID no	License no	Mining company	Sampling company	Project	Project part	Sample type	Collection date	Station	LatDecDeg	LonDecDeg	Collector	Collection method	Comments	Storage
15126	MEL 1962-15	Black Angel Mining	Green water	Maarmorilik	1992 monitoring	Blue mussel	1992091	T12Ø	71.13766667	-51.24033333	John Bly	Hand	40-49 mm	DCE
15127	MEL 1962-15	Black Angel Mining	Green water	Maarmorilik	1992 monitoring	Crinkled snow lichen	1992091	T12Ø	71.13766667	-51.24033333	John Bly	Hand		DCE
15128	MEL 1962-15	Black Angel Mining	Green water	Maarmorilik	1992 monitoring	Bladder wrack	1992091	T12Ø	71.13766667	-51.24033333	John Bly	Hand		DCE
15129	MEL 1962-15	Black Angel Mining	Green water	Maarmorilik	1992 monitoring	Shorthorn sculpin	1992091	T12Ø	71.13766667	-51.24033333	John Bly	Angling		DCE
15129	MEL 1962-15	Black Angel Mining	Green water	Maarmorilik	1992 monitoring	Sediment	1992091	T5	71.26185	-51.326687	John Bly	Kayak corer		Company

For certain sample types (blue mussels, fish and sediments) additional Excel sheets also need to be filled out (see appendix B, C and D).

Appendix B – Additional registration for shellfish samples

For mussel samples, not only the sheet 'Basic' in DCE_SampleReporting.xlsx needs to be filled out, but also a sheet called 'Shellfish', which contains special information pertaining only to this sample type. The sheet contains help text explaining how to fill out the different fields, and in some cases field values are constrained to certain predefined codes. All fields need to be filled out. In case of uncertainty regarding the registration, please contact DCE.

Hypothetical example of data recorded in the Shellfish table

ID no	No of individu-als	Shell length min (mm)	Shell length max (mm)	Shell length average (mm)	sample wet weight (g)	Depurated	Comments
25317	20	40	49	43.5	82	Yes	
25318	20	50	59	54.2	93	Yes	
25319	20	40	49	44.1	88	No	
25320	20	50	59	55.2	97	No	

Appendix C – Additional registration for fish samples

For fish samples (e.g. sculpins and Arctic char), not only the sheet 'Basic' in DCE_SampleReporting.xlsx needs to be filled out, but also a sheet called 'Fish', which contains special information pertaining only to this sample type. The sheet contains help text explaining to how to fill out the different fields, and in some cases field values are constrained to certain predefined codes. All fields need to be filled out. In case of uncertainty regarding the registration, please contact DCE.

Hypothetical example of data recorded in the Fish table

ID no	Sex	Total length (cm)	Weight (g)	Liver sampled	Muscle sampled	Otoliths sampled	Liver weight (g)	Stomach content	Char pop	Comments
34073	Female	34.5	581	Yes	Yes	No	25.43	Crabs		From polluted area
34074	Female	23	204	Yes	Yes	Yes	13.15	Empty		
34075	Male	26	258	Yes	Yes	Yes	18.33			
34076	Female	52	2102	Yes	Yes	Yes	21.51	Capelin	Anadromous	
34077	Male	22	95.4	Yes	No	No	1.72	Empty	Landlocked	
ID no	Sex	Total length (cm)	Weight (g)	Liver sampled	Muscle sampled	Otoliths sampled	Liver weight (g)	Stomach content	Char pop	Comments

Appendix D – Additional registration for sediment samples

For sediment samples, not only the sheet 'Basic' in DCE_SampleReporting.xlsx needs to be filled out, but also a sheet called 'Sediment', which contains special information pertaining only to this sample type. The sheet contains help text explaining to how to fill out the different fields, and in some cases field values are constrained to certain predefined codes. All fields need to be filled out. In case of uncertainty regarding the registration, please contact DCE.

Hypothetical example of data recorded in the Sediment table

ID no	Water depth (m)	Slice size (cm)	Number of slices	Core height (cm)	Sieved	Mesh size (mm)	Comments
40011	10	5	2	10	Yes	1	Polluted area, photo is captured
40114	65	2	20	40	No		Shells, photo is captured
40115	70	2	10	20	No		Corer hit rock at bottom

Appendix E – Multielement (ICP-MS) data from analysis of baseline samples

DCE_SampleReporting.xlsx contains a sheet called 'Multielementdata' which is used for the mandatory reporting of data from ICP-MS analyses of baseline samples. The sheet contains help text on how to fill out the different fields. Each row in the table is an analysis result with concentrations of different elements in the columns (90 different element columns). There can be several analysis results for one sample ID number (duplicate measurements of same sample; different tissues of the same fish etc.). If an analysis is not conducted on a whole organism (like lichen), it is very important to specify what part is analyzed under 'Subsample' (e.g., muscle, liver, new growth tips, size group 2-3 cm, soft parts etc.). For each series of measurements, it is also important to report detection limit values for the different elements. Detection limits are reported in rows, where 'ID no' is coded as 0 and 'Subsample' is coded as 'Detection limit'. Both detection limits and element concentrations in samples are reported as positive decimal numbers (use blank fields for elements that were not measured). However, measurements under detection limit are reported as detection limit * -1. Thus, in the table with example data (below), the measurement of P in the filtered water sample 70147 was below detection limit, and the element concentration value is therefore reported as the detection limit for P for that measurement series (37.4097; see row 4, column P) times -1, which equals -37.4097. The measurement of Be in blue mussel samples 66602 and 66603 were also below detection limit and are reported as -0.0118 ([row 1, column Be] * -1). Remember to record the unit of measurement (mg/kg or ug/l) and the basis of measurement (d=dry; w=wet). If concentrations are given on wet basis, the dry matter percentage also needs to be given in the column 'dm%'. In case of uncertainty regarding the registration, please contact DCE.

Hypothetical example of ICP-MS analysis data reported via the multielement table

Sample_type	ID no	Subsample	Subsample ID	Lab	Lab no	Date	Methods	Notes	dm%	Basis	Unit	H	He	Li	Be	…	Al	Si	P	…	U
Detection limit	0	Detection limit		DCE		20230607	ICP-MS			d	mg/kg			0.1148	0.0118		36.9273		18.2080		0.0007
Blue mussel	66602	2-3 cm		DCE	4324	20230607	ICP-MS			d	mg/kg			0.5306	-0.0118		115.2416		11807.8680		0.2274
Blue mussel	66603	4-5 cm		DCE	4325	20230607	ICP-MS			d	mg/kg			0.4792	-0.0118		119.6675		11241.2010		0.2570
Detection limit	0	Detection limit		DCE		20241200	ICP-MS			w	ug/l			0.0405	0.0013		0.1878		37.4097		0.0001
Freshwater, unfiltered	70146			DCE		20241200	ICP-MS			w	ug/l			2.7327	0.0158		707.3020		42.5602		1.6297
Freshwater, filte-red_045my	70147			DCE		20241200	ICP-MS			w	ug/l			0.9642	-0.0013		31.5620		-37.4097		1.4257
Detection limit	0	Detection limit		DCE		20241200	ICP-MS			d	mg/kg			0.0158	0.0001		1.8168		2.4791		0.0001
Crinkled snow lichen	70035			DCE	5721	20241200	ICP-MS			d	mg/kg			0.0496	0.0024		93.4981		478.4570		0.0211
Crinkled snow lichen	70039			DCE	5722	20241200	ICP-MS			d	mg/kg			0.0912	0.0039		165.7945		536.7072		0.0295
Detection limit	0	Detection limit		DCE		20150905	ICP-MS			w	mg/kg			0.0391	0.0120		0.5793	0.0637	2.9936		0.0020
shorthorn sculpin	51124	Liver		DCE	3490	20150905	ICP-MS		32.9	w	mg/kg			0.0330	0.0018		1.6268	0.0500	3232.3000		0.0002
shorthorn sculpin	51124	Muscle		DCE	4495	20150906	ICP-MS		20.8	w	mg/kg			0.1236	0.0018		68.2490	0.4281	2010.8000		0.0023

GUIDELINE FOR COLLECTION OF ENVIRONMENTAL SAMPLES TO THE GREENLAND MINERAL RESOURCES ENVIRONMENTAL SAMPLE BANK

This technical report includes guidelines and instructions on sampling procedures for collection of environmental samples in relation to mineral resource projects in Greenland. The aim of this report is to ensure that sampling is reproducible, comparable, and done according to international standards. The report also describes the registration and reporting of the samples.

This technical report replaces the instructions provided in the 'Guideline for collection of environmental samples to the Greenland mineral resources environmental sample bank; TR239.

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