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PATTLE DELAMORE PARTNERS LTD

ALIL Environmental Monitoring Plan

Ashburton Lyndhurst Irrigation Limited

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Prepared for

Ashburton Lyndhurst Irrigation Limited

December 2021

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Limitations:

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Table of Contents

Table of Figures

Table of Tables

Appendices

1.0 Introduction

Ashburton Lyndhurst Irrigation Ltd (ALIL) were granted consent CRC185469 on 28 June 2021 to discharge nutrients to land from farming between the Rakaia and Ashburton Rivers.

Consent CRC185469 requires ALIL to hold and adhere to an Environmental Monitoring Plan, that outlines how groundwater and surface water quality will be monitored for the duration of the consent.

This Environmental Monitoring Plan was prepared by Pattle Delamore Partners (PDP) on behalf of ALIL.

2.0 Consent Conditions

The requirements for the Environmental Monitoring Plan are listed in conditions 21 to 28 of CRC185469 and are included below:

21. Within six months of the Commencement Date, the Consent Holder shall submit to the Regional Leader - Monitoring and Compliance, Canterbury Regional Council, an Environmental Monitoring Plan that satisfies Conditions 22 and 23 and which has been prepared by suitably qualified and experienced person(s).
22. The objectives of the Environmental Monitoring Plan shall be to:
    a. obtain water quality information that may assist in better understanding the effects of nutrient discharges from properties within Schedule CRC185469A:
        i. on groundwater nitrate-nitrogen concentrations over-time; and
        ii. surface water quality over time.
    b. require the reporting of any water quality information gathered to the Canterbury Regional Council for the purpose of better informing future water resource management in the Command Area;
    c. require the Consent Holder to investigate and respond to changes in water quality attribute state(s) or band(s) (as might apply) for certain contaminants as to be identified based on five years of data in accordance with Condition 23 (a *Deterioration*) as specified in Table CRC185469-2; and
    d. to require the Consent Holder to prepare a Remediation and Response Plan in consultation with Te Runanga o Arowhenua following any identified Deterioration that includes:

i. requiring the Consent Holder to manage nutrient losses that are determined to be contributing to any identified Deterioration in a manner that is consistent with improving water quality over time; and

ii. reviewing individual Property Farm Environment Plans or Certified Freshwater Farm Plans (as might apply) through the EMS programme where it is determined those farming activities are contributing to any identified Deterioration.

23.

Catchment groundwater monitoring

a. Subject to Condition 26, the Consent Holder shall undertake (either directly or through a catchment group) water quality sampling on a minimum of 10 bores at the locations generally shown on attached Plan CRC185469X, with all bores being sampled quarterly for nitrate-nitrogen in accordance with the requirements of the National Environmental Monitoring Standards Water Quality – Part 1 Groundwater dated March 2019.

Localised surface water monitoring

b. Subject to Condition 26 the Consent Holder shall undertake (either directly or through a catchment group) surface water quality monitoring in the following waterbodies with the final monitoring site in each waterbody to be determined in consultation with Te Runanga o Arowhenua and the Regional Leader – Monitoring and Compliance, Canterbury Regional Council at the locations generally shown on attached Plan CRC185469Y:

i. Wakanui Stream;

ii. Mt Harding Creek;

iii. Ashburton River South Branch at Digbys Bridge; and

iv. any further or alternative location(s) that may be determined through the review of the Environmental Monitoring Programme that is to be undertaken in accordance with Condition26, and such surface water monitoring shall include monitoring of the contaminants listed in Table CRC185469-2, with:

A. monitoring to occur at the frequencies included in column 2 of Table CRC185469-2; and

B. where specified in column 3 of Table CRC185469-2, monitoring being undertaken for contaminants to

determine a Base Attribute State, calculated for each monitoring site at the commencement of the consent for monitoring sites where adequate water quality data already exists, or after five years of monitoring where no current water quality data exists, provided that in the case of:

i. Mt Harding Creek and the Ashburton River South Branch, monitoring shall include all contaminants listed in Table CRC185469-2; and

ii. the Wakanui Stream, monitoring shall be limited to Nitrate, Periphyton (where suitable hard bottom substrate is present), and Macrophytes.

Table CRC185469-2

Contaminant	Frequency of sampling	Base Attribute State	Deterioration
Nitrate toxicity mg NO_3- N mg/L	Monthly	Median and 95th percentile of previous 5 years' data.	Where the annual (1 July to 30 June) median and/or 95th percentile NO_3- N mg/L are greater than the calculated base attribute state.
Dissolved reactive phosphorous DRP mg/L	Monthly	Median and 95th percentile of previous 5 years' data.	Where the annual (1 July to 30 June) median and/or 95th percentile DRP mg/L are greater than the calculated base attribute state.
Escherichia coli E. coli/100ml	Monthly	The attribute band as calculated in accordance with the Table 9 of the NPSFM 2020 (August 2020) and using 5 years of data	Where the attribute band (as per the NPSFM 2020) is worse than the calculated base attribute state (using 5-year rolling data).
Macroinvertebrates	Annually between December and March (inclusive)	The median attribute band as calculated in accordance with Table	Where the attribute band (as per the NPSFM 2020) is worse than the

Contaminant	Frequency of sampling	Base Attribute State	Deterioration
	(QMCI or MCI using NEMS 2020 methodology)	14 of the NPSFM 2020 (August 2020) and using 5 years of data.	calculated base attribute state (using annual data)
Deposited fine sediment (percentage cover)	Monthly	The attribute band as calculated in accordance with Table 14 of the NPSFM 2020 (August 2020) and using 5 years of data.	Where the attribute band (as per the NPSFM 2020) is worse than the calculated base attribute state (using 5-year rolling data).
Periphyton (percentage cover and chlorophyll-a)	Monthly	Not applicable for percentage cover For Chlorophyll-a: the attribute band as calculated using 5 years of data in accordance with Table 2 of the NPSFM 2020 (August 2020).	Not applicable for percentage cover For Chlorophyll-a: where the attribute band (as per the NPSFM 2020) is worse than the calculated base attribute state (using 5-year rolling data).
Macrophytes (percentage cover)	Monthly	Not applicable	Not applicable

Advisory note 1: The Base Attribute State(s) are based on the attribute bands and attribute states in Appendix 2A of the National Policy Statement for Freshwater Management 2020 (August 2020).

Advisory note 2: Where water quality sampling is undertaken as part of a catchment group, members of the group may seek to rely on the same groundwater monitoring bores or surface water monitoring sites as part of their respective consent requirements.

Advisory note 3: The 'Base Attribute State' (numeric) for nitrate will be calculated as the maximum of annual median and the maximum of annual 95 percentiles from the first five years of numeric attribute states calculated from monthly data.

24. The Consent Holder shall implement the Environmental Monitoring Programme within 12 months of the Commencement Date. Following its implementation, the Consent Holder shall provide a summary within the annual report by 1 December 2022 and then annually thereafter that sets

out the results of all sampling undertaken over the previous 12-month period, including a discussion on:

a. the extent to which there has been an identified Deterioration; and

b. for contaminants where a Deterioration is not defined (being Periphyton and Macrophytes percent covers), the nature of any changes over time, including any unexpected declines.

25. In the event that there is a Deterioration that is identified as a part of the water monitoring required under Condition 23(b), the Consent Holder (either directly or through a catchment group) shall, within one month of the Deterioration being identified, engage a suitably qualified and experienced person to prepare a Remediation and Response Plan. The Remediation and Response Plan shall:

    a. discuss the potential causes of the Deterioration, and the extent to which they might be attributable to the activities on farmland under the management of this resource consent;

    b. advise on any changes that might be made to a Farm Environment Plan or Management Plan for Farming Activities for the Properties included in Schedule CRC185469A, on the basis that any changes will be proportionate to the relative contributions of those Propert(ies) to the Deterioration;

    c. advise how nutrient discharges may be further managed to ensure improving water quality over time;

    d. advise on any further or amended monitoring that may be required to better understand the Deterioration (and the timeframes for that monitoring); and

    e. remain in place and be subject to regular reviews for the duration of any Deterioration, as might be identified through further monitoring.

    The Remediation and Response Plan shall be prepared in consultation with Te Runanga o Arowhenua and shall be completed within six months of the Deterioration being identified (or such other time as may be agreed to by the Regional Leader - Monitoring and Compliance, Canterbury Regional Council) and the Consent Holder shall implement any recommendations. A copy of the completed Remediation and Response Plan shall be provided to Te Runanga o Arowhenua and to the Regional Leader - Monitoring and Compliance, Canterbury Regional Council as a part of the annual reporting required under Condition 29.

26. The Consent Holder shall undertake a review of the groundwater and surface water monitoring required under Condition 23:

a. within the six month period that begins on the date that is five years after the Commencement Date, being the date which the Base Attribute State has been determined for all listed contaminants,

b. at any other time that may be determined by the Consent Holder; or

c. on making any change to Schedule CRC185469A that results in increasing the area managed by the scheme on a Property or adjoining Properties by more than 200 hectares over that occurring at the Commencement Date, provided that in the case of a review under this Condition 26(c), the review shall be limited to the effects of the change and the need to consider further groundwater and/or surface water monitoring sites.

27. If the Consent Holder is required to or elects to undertake such a review under Condition 26, the Consent Holder will engage a suitably qualified and experienced person to:

    a. Advise on any changes that might be made to add, remove or amend:

        i. Groundwater monitoring bores;

        ii. Surface water monitoring sites;

        iii. Contaminants;

        iv. how a Deterioration is determined; and

        v. Sampling frequency,

    b. Prepare a Water Monitoring Amendment Report that:

        i. Outlines the reasons for the change(s)proposed; and

        ii. Confirms that the additional, removal or amendment will continue to enable the Consent Holder to meet the objectives set out in Condition 22;

        iii. Consult with the Regional Leader - Monitoring and Compliance, Canterbury Regional Council in the preparation of the Water Monitoring Amendment Report; and

        iv. Provide a copy of the of the Water Monitoring Amendment Report to the Regional Leader - Monitoring and Compliance, Canterbury Regional Council for certification that any change(s) proposed meet the requirements of this Condition 27 and the objectives outlined in Condition 22.

28. The Consent Holder shall only implement the change(s) proposed to the monitoring required in Condition 23 if written certification is provided by the Regional Leader – Monitoring and Compliance, Canterbury Regional Council.

3.0 Groundwater Monitoring

The following sections provide details of the bores to be sampled along with the sampling methodology and requirements. A step-by-step summary of the groundwater sampling procedure that can be referred to while on site is provided in section 3.3 of this report.

3.1 Monitoring Bores

Table 1 below provides a list of the ten proposed bores that will be monitored for nitrate-nitrogen to satisfy condition 23a of the resource consent CRC185469. Figure A1 (Appendix A) shows the locations of these bores and photos of all the bores are included in Appendix B.

If groundwater samples cannot be collected from a bore for two consecutive quarters, ALIL, PDP and Environment Canterbury will review the adequacy of continuing with that bore as part of the ALIL groundwater monitoring programme.

Table 1: Details of proposed monitoring bores

ECan bore number	Bore owner	Address	Diameter (mm)	Depth (m bgl)	Screened interval (m bgl)	Water level (m bgl)*	NZTMX	NZTMY
K36/0018	Holmes, Gordon	Sivonholm Lodge - off Mount Hutt station Rd	150	52.1	-	42.7	1490094	5170165
K36/0090	Lilley R	Methven Highway - near intersection with Reynolds Road	150	34.8	-	6.1	1490289	5164155
K36/0104	Currie A	Ashburton-Rakaia Gorge Road RD6	100	24.0	-	15.0	1490382	5160392
K36/0131	Nixon W R	Braemar Lauriston Road	125	51.0	50 - 51	35.4	1493699	5153337
K36/0179	Taralea Farms Limited	Blands Road - near intersection with Methven Highway	600	9.0	-	2.4	1489705	5153626
K36/0200	Mr & Mrs L A & S A Glass	Pole Road	200	62.8	59.8 - 62.8	48.8	1493529	5156925

Table 1: Details of proposed monitoring bores

ECan bore number	Bore owner	Address	Diameter (mm)	Depth (m bgl)	Screened interval (m bgl)	Water level (m bgl)*	NZTMX	NZTMY
K37/0013	Peter Woods	765 Winchmore Dromore Road	150	30.0	28.8 - 29.9	17.5	1499229	5145737
L36/0970	Rowe G A	Dromore Methven Road (rapid # 765)	125	72.0	69.5 - 71.5	41.1	1501591	5150347
L36/2205	Mr & Mrs M R & L J Thomson	978 Dromore Methven Road	150	66.5	65 - 66.5	47.1	1500140	5151987
L37/1383	Mr & Mrs RH & SC Duncan	273 Mitcham Road	200	40.0	37 - 40	18.7	1500815	5143232

*Notes: *Water level sourced from ECan's online database – assumed to be static water level after bore was installed.*

Condition 23a of the resource consent requires that all groundwater samples are collected as per the requirements outlined within the national environmental monitoring standards (NEMS) document Water Quality Part 1 of 4: Sampling, Measuring, Processing and Archiving of Discrete Groundwater Quality Data (NEMS-WQ1), dated March 2019. The groundwater sampling procedures provided in this report are based on the NEMS-WQ1 (2019) document.

It is understood that all of the monitoring bores, except for K36/0090, are actively used for abstraction purposes and have a pump currently installed. ALIL will supply a pump for purging K36/0090.

It is important that the groundwater samples are representative of the groundwater in the aquifer from which the groundwater is abstracted from. Therefore, sampling points on the bore shall be located as close to the bore head works as possible (i.e. as close to the top of the bore where the pipe works come out of the ground). It is also important that the sampling point on all bores is upstream of any storage tanks and pressure tanks, as tanks can cause contamination of samples. The sampling point should consist of a tap (or similar) that can be easily accessed and turned on and off by the sampler. A photo of an example bore head with a sampling tap that is considered to be suitable for monitoring is shown in Figure 1, below. If there is no suitable sampling point already installed on the bore head works, then modifications of the bore head may be required so a tap can be installed on the bore.

It is also preferable that a groundwater level can be measured in each bore. This involves lowering a water level dipper into the bore and measuring the groundwater level (preferably while the bore is turned off) to measure the static groundwater level. This water level measurement will be used to assess the

volume of water required to be removed from the bore as part of the purging process. Most bore heads will have an access point through which groundwater levels can be measured, but if this access is not available, the bore head may have to be modified to accommodate a water level dipper. An example of a groundwater level access point is shown in Figure 1.

Figure 1: Photo of a bore with appropriate sampling tap installed on the bore headworks. Note this sampling point is as close to bore head as possible and upstream of any storage tanks or pressure tanks.

Details of the sampling methodology and purging process are provided below and are based on the assumption that all of the monitoring bores listed in Table 1 have an existing pump and can be modified to have a sampling point and groundwater level access installed on the bore head works.

3.2 Sampling Methodology: in Depth

Groundwater sampling from bores with existing pumps installed involves pumping water from the bore, assuming that the bore has not been pumped recently. The standing water within the casing can become contaminated from

surface influences or from accumulation of material within the bore and can affect the water quality of the bore. Therefore, purging removes this standing water within the bore casing to ensure that fresh water from the surrounding aquifer is flowing into the bore prior to sampling. In addition to purging, field water quality measurements are also undertaken to check that the water in the bore is fresh groundwater sourced from the aquifer and must be undertaken before samples can be collected. The details of these steps are provided below.

Bore Purging

It is preferable that a groundwater level in the bore to be sampled is measured upon arrival and the volume of standing water within the casing of the bore calculated. If the bore is not operating upon arrival and a water level is unable to be measured, then water level has to be assumed to be at 0 m bgl (i.e. water level is at ground level) for the purposes of purging, which will take additional time to purge, particularly if the bore is deep.

Unless the bore owner is able to confirm the duration that the bore has been operating prior to the sampler's arrival at the site, then it is recommended that the bore will need to be purged three bore casing volumes regardless of whether it was operating upon arrival. In this case, the measured water level, even while the bore is operating, can used to calculate the minimum purge volume.

The volume of water that will need to be removed as part of purging (minimum purge volume) is calculated using the equation below:

$$Min.Purge\ Volume = 3\ \times (3.14\ \times [bore\ depth - depth\ to\ water\ level] \times [bore\ radius]^2 \times 1000)$$

Note: all measurements are expressed in metres (m) and the purge volume is in litres (L).

During the initial sampling round, pumping rates should be measured (if no flow rate meter is installed on the bore), as understanding the flow rate and the volume of standing water within the bore casing will determine how long the bore will need to be pumped to purge three casing volumes before samples can be collected from the bore. Unless indicated otherwise by the bore owner, the pumping rate can be assumed to remain consistent and used for subsequent sampling rounds for calculating the time taken to remove the minimum purge volume from the bore.

Once the minimum purge volume has been calculated and the pumping rate of the bore is known, the time required to remove the minimum purge volume can be calculated using the equation below:

$$Time\ Taken\ to\ Purge\ Bore = \frac{Minimum\ Purge\ volume}{[Bore\ Pumping\ Rate\ \times 60]}$$

Note: '*Time Taken to Purge Bore*' expressed in minutes, '*Minimum Purge Volume'* express in litres (L) and '*Bore Pumping Rate*' expressed in litres per second (L/s).

It is important to record the time when the bore pump is turned on. If the pump is already operating upon arrival, record the time from when field water quality parameters began to be measured from.

All water level measurements, flow rates and minimum purge volumes should be recorded on the PDP groundwater purge and sampling form provided in Appendix C. A new form should be used for each bore and each sampling round and kept as a record of sampling undertaken for quality assurance and control purposes.

The purged water should be discharged away from the bore to avoid ponding of water around the bore head and sampling area. Ideally the purged water would be contained within the existing pipework/tank system at each property, although if this isn't possible then the purged water should be discharged as far from the bore head as possible. The sampling tap should only be used for measuring field water quality parameters and collecting samples from and not for discharging water.

Field Water Quality Measurements

During purging of the standing water in the bore casing, field water quality measurements need to be recorded. The purpose of the field water quality measurements is to confirm that standing water within the bore has been removed during purging and that fresh water sourced from the aquifer is flowing into the bore prior to sampling. The field water quality measurements should include pH, specific conductance (i.e. electrical conductivity at 25°C) and temperature at a minimum, but oxidation reduction potential (ORP), dissolved oxygen (DO) and turbidity can also be recorded. Note that if ORP and DO are measured, then a flow cell must be used to measure these parameters to avoid the groundwater mixing with the atmosphere prior to the measurement of ORP and DO.

Calibration of field water quality meters should be undertaken daily, prior to sampling on site. Calibration provides a check of the meter's sensors, as sensors (particularly pH sensors) can drift over time. Calibration involves submerging the sensors within a standardised solution of a known value and adjusting the sensor to match the solution's value for a particular water quality parameter. Specific details of the calibration procedures are not provided within this report as procedure will vary between water quality meters. However, PDP can provide onsite training for calibrating specific water quality meters if required.

A minimum of four lots of field water quality measurements (i.e. pH, specific conductance and temperature) should be recorded while removing the minimum purge volume of water from the bore. The period of time between field water quality measurements during purging is typically between 1 to 5 minutes but will depend on the calculated time required to purge the bore. The time of each field water quality measurement and the elapsed time since the start of the pumping

to remove the minimum purge volume should be recorded. All field water quality measurements should be recorded on the PDP groundwater purge and sampling form provided in Appendix C.

The field water quality measurements should be measured by placing the water quality meter probe within a clean vessel. A length of temporary tubing should be connected to the sampling tap on the bore head with the other end placed in the vessel next to the water quality meter probe. The open end of the tubing in the vessel should be placed near the bottom and remain submerged to reduce mixing the water with the atmosphere. When the tap is turned on while the bore is being purged, water will flow into the vessel via the tubing at the bottom of the vessel and water quality parameters can be measured. Water should be allowed to go over the top of the vessel and the overflowing water should be directed away from the bore head and work area to avoid ponding. Figure 2 below provides an example setup with tubing and the water quality meter probe within a vessel during purging.

Figure 2: Example of a typical field water quality measuring setup during purging of a bore for groundwater sampling. Note that the opening of the tubing is placed at the bottom of the vessel to reduce mixing of the water with the atmosphere. Image sourced from NEMS-WQ1 (2019).

It is important that the sampling tap is turned on after the bore pump is turned on and the sampling tap opened slowly to a point where a slow, laminar flow rate with continuous pressure can be maintained. Avoid excessive water pressure and turbulence (i.e. lots of air bubbles) in the tubing when measuring field parameters.

Ideally the length of tubing between the sampling tap and the vessel for measuring field water quality parameters should be dedicated to each bore and kept clean between each sampling round in a plastic bag labelled with the bore number that it is used at.

The vessel used to measure water quality parameters can be used for all monitoring bores but should be kept clean and should be rinsed prior to sampling using the water from the particular bore being sampled.

Sampling

Groundwater samples can only be collected once the following criteria has been met:

- The calculated minimum purge volume of water within the bore casing has been removed; and
- The tubing connected to the sampling point and the vessel in which field measurements have been made, has been rinsed at least three times their volume by water from the bore being sampled; and
- Water temperature, specific conductivity and pH have been measured at least four times and that the final three consecutive field measurements are within the following limits:
    - Water temperature: ± 0.2°C; and
    - Specific conductance: ± 3%, and
    - pH: ±0.1 pH unit.

Once the above criteria have been met, samples should then be collected into bottles supplied by the laboratory.

It is preferable to label all sample bottles prior to collection. All sample bottles shall be labelled clearly, noting the date and time of collection and the ECan bore number the sample was collected from. Bottle labels (sampling site name) can be labelled by the laboratory but if labelled at the sampling site, a permanent marker should be used to label the bottles to ensure the sample details are not erased during either the handling or transport of the sample bottles.

The sampler's hands should be clean and nitrile gloves should be worn during the filling of the sample bottles. The bottles should be filled using the tubing connected to the sampling tap by holding the tubing in one hand and the open sample bottle in the other hand. Care should be taken not to touch the threads of the sample bottle or the inside of the bottle cap. The bottle should be filled to a level at the base of the threaded section of the bottle. It is likely that the sample bottles will contain a preservative, so it is important that the sample bottle is not rinsed. This should be confirmed with the laboratory supplying the sample bottles prior to sampling.

Following the collection of the samples, the caps/lids of sample bottles should be firmly sealed to ensure water cannot leak out of the bottle and also so no water could ingress into the bottle. All sample bottles should be placed within a chilly bin (or similar storage container) to avoid exposure to sunlight, which can cause warming and photochemical degradation of the samples.

The samples should be cooled quickly and evenly within the storage container (i.e. chilly bin) to a temperature ideally between 4 and 10°C. Sealed chemical cooling packs (e.g. slicker pads supplied by the laboratory) can be used to cool the samples down. Alternatively, regular ice (i.e. ice that can be purchased at a service station etc) can be used in place of chemical ice packs, although the ice must be double bagged within ziplock plastic bags, or the sample bottles placed within zip lock bags, if the ice is loose within the storage container.

Samples should be submitted to the laboratory as soon as possible following collection and must be kept chilled between when they are collected at the bore and when they are submitted to the laboratory. Ideally, all samples should be submitted to the laboratory on the same day as collection, but they can be refrigerated overnight (maintained between 4 and 10°C), if same day submission is not possible. Samples should be submitted to the laboratory within 24 hours at the latest.

A chain of custody (CoC) form should be completed during each sampling round and accompany all groundwater samples when they are submitted at the laboratory. The CoC provides a trail that can be audited and can be used for quality assurance to check sample integrity. The CoC form should include the sample number, date and time of sample collection and the requested determinands required to be tested at the laboratory. An example CoC form is provided in Appendix C (sourced from NEMS-WQ1, 2019) but most laboratories typically provide a CoC form when sample bottles are ordered from the laboratory that can be used instead of the CoC provided in Appendix C.

3.3 Sampling Methodology: Step-by-step Procedure

This section provides a step-by-step procedure for the sampling of groundwater is provided below and is a summary of the information described in Section 3.2 of this report. The summary below is provided in a format that can be referred to while in the field.

1. Pre-site work:
    a. Prepare field equipment including sample bottles provided by laboratory (see full list of recommended equipment in Appendix C).
    b. Calibrate field water quality meter.

c. Confirm access to bores and whether bores will be operating prior to arrival.

2. Upon arrival at bore, check if bore pump is currently operating or turned off. If the bore owner can confirm that the bore has been operating for some time, check if the minimum purge volume of water has been purged from the bore (equation in Step 4). If the minimum purge volume has already been removed, measure groundwater level, note that bore is operating and proceed to Step 6, otherwise proceed to Step 3. If the bore is operating but it is not known how long it has been operating, assume that the minimum purge volume needs to be removed and proceed to Step 3.
3. Measure groundwater level in bore.
4. Calculate minimum purge volume for bore using the equation below:

$$Min.\,Purge\ Volume = 3\ \times (3.14\ \times [bore\ depth - depth\ to\ water\ level] \times [bore\ radius]^2 \times 1000)$$

Note:

- All measurements are expressed in metres.
- The purge volume will be expressed in litres (L).
- If groundwater level is unable to be measured, assume "*depth to water level*" = 0.

5. Ensure sampling point is closed and turn bore pump on. Record the time that the pump was started.
6. Connect temporary sampling tubing to sampling point (i.e. tap) on bore to measure field water quality parameters. Place other end of tubing in a clean open vessel along with water quality meter probe.
7. With nitrile gloves covering hands, open sampling point tap slowly to allow water to flow into the vessel with the water quality meter probe via the tubing. The sampling tap should be opened to point where a continuous and laminar flow rate can be maintained. Avoid excessive water pressure and turbulence in the temporary pipework when measuring field parameters.
8. Record pH, specific conductance and temperature and the time of measurement (see attached PDP groundwater purge and sampling form in Appendix C).
9. If pumping rate of the bore is unknown, measure the flow rate (in L/s) at a suitable location downstream of the bore headworks and sampling point using a large bucket/container and measuring the time taken to fill the bucket/container.

10. Based on the minimum purge volume calculate in Step 4 and bore pumping rate, calculate the time taken to purge the bore using the equation below:

$$Time\ Taken\ to\ Purge\ Bore = \frac{Minimum\ Purge\ volume}{[Bore\ Pumping\ Rate\ \times 60]}$$

Note:

- "*Time Taken to Purge Bore*" expressed in minutes.
- "*Minimum purge volume*" expressed in litres (L).
- "*Bore Pumping Rate*" expressed in litres per second (L/s).

11. Record field water quality measurements at approximately evenly spaced time increments (i.e. typically between 1 to 5 minutes but dependant on time required to remove minimum purge volume) so at least four water quality measurements have been recorded during the period taken to remove the minimum purge volume. The time of each water quality measurement should also be recorded. The measurements should be recorded on the groundwater purge and sampling form in Appendix C.
12. Continue to pump water from bore and record field water quality parameters until:
    a. The minimum purge volume is removed from the bore; and
    b. The tubing connected to the sampling point and the vessel in which field measurements have been made, has been rinsed at least three times their volume by water from the bore being sampled; and
    c. Water temperature, specific conductivity and pH have been measured at least four times and that the last three consecutive field measurements are within the following limits:
        i. Water temperature: ± 0.2°C; and
        ii. Specific conductance: ± 3%, and
        iii. pH: ±0.1 pH unit.
13. Once all of the requirements of Step 12 are met, samples can be collected from the bore. The sampler's hands should be clean and nitrile gloves should be worn during sample collection. Sample bottles should be labelled with the bore number along with the time and date of when the samples were collected using a permanent marker. The sample bottles should be filled using the tubing connected to the sample tap on the bore head works.

14. After bottles have been filled, ensure bottle caps and lids are secured on tightly to avoid leakage. The bottles should then be placed within a chilly bin containing sealed ice packs (i.e. chemical cold packs or double bagged loose ice) as soon as possible and the lid of the chilly bin closed, to reduce exposure to sunlight.
15. Details of the samples should be recorded on the CoC form (see Appendix C for an example CoC form).
16. Once sample bottles have been filled and stored in the chilly bin, close sampling tap and turn the bore pump off (if required). Disconnect all sampling tubing etc and pack up equipment.
17. The samples should be submitted to the laboratory along with the CoC ideally on the same day as collection but no more than 24 hours after collection. Samples can be stored in a refrigerator overnight if required at a temperature between 4 and 10°C.
18. The final field water quality measurements (including the date and time) should be recorded in a database, so they can be compared alongside the laboratory water quality results.

Note that the vessel used to measure the field water quality parameters should be dedicated for water quality sampling purposes and kept clean. In addition, it is preferable to have dedicated sampling tubing and any associated fittings for each monitoring bore to reduce the potential for cross contamination of samples, although this is not a requirement of NEMS-WQ1 (2019).

It is recommended that PDP accompany ALIL groundwater sampling staff during at least the initial round of groundwater sampling to ensure that the sampling methodology is being applied correctly. Following the initial sampling round, quality control can be undertaken in the form of regular audits of groundwater sampling by PDP to ensure that groundwater sampling is being undertaken as per the requirements of the resource consent.

4.0 Surface Water Monitoring

4.1 Monitoring Sites

4.1.1 Site Reconnaissance and Selection

Site reconnaissance was undertaken by PDP at each of the watercourses listed in CRC185469 on 28 October 2021. The objective of the site reconnaissance was to identify appropriate in-stream conditions for water quality and ecological monitoring at each site. Methods are described in order of occurrence below.

1) Stream width and depth measurements were taken at a single representative point, and a reach measuring 5-times the wetted stream width was defined for each site.

2) Reach-scale rapid habitat assessments were completed for each reach, including depth measurements, assessments of flow and bed substrate composition, and general observations, in order to conform each reach met the following criteria:

   a) at no point did the water depth within the reach exceed 60 cm depth;

   b) 'run' flow conditions[1] were dominant throughout the reach;

   c) the upstream extent of the reach was sufficiently distanced from the influence of point sources, tributary stream and drain confluences, and dead zones (e.g. backflow eddies) such that reasonable mixing would have occurred[2]; and,

   d) with consideration to the above-mentioned criteria, the reach was considered representative of the watercourse as a whole.

3) GPS coordinates were recorded and photographs taken at the upstream extent of each reach, including key identifying features (i.e., mature vegetation and structures) to facilitate easy location for repeat monitoring.

4) Monitoring reaches were given a numeric site identifier, indicating the order in which sites are to be visited. For streams with multiple monitoring sites, the order of sampling begins at the furthest downstream site and proceeds upstream.

Monitoring site locations are shown in Figure A2 (Appendix A). Identifying photographs for each monitoring site are provided in Appendix D. A brief description of each monitoring site, based on site reconnaissance is provided below.

4.1.2 Wakanui Creek

There has been question as to the exact alignment of Wakanui Creek due to the stock water inputs above Farm Road and the eastern alignment known as 'Mill Creek' through and downstream of the Ashburton township. During site reconnaissance, it was considered that although flow from Wakanui Creek does flow to the east, as part of the 'Mill Creek' alignment, some flow is diverted through a channel (exact alignment still to be determined) that discharges into the Ashburton River. As effects to the lower Hakatere/Ashburton River and its

[1] Low to moderate depth, slow to moderate water velocity, uniform to slightly variable current, surface smooth-rippled and unbroken.
[2] As per the Canterbury land and Water Regional Plan, the Mixing Zone of any discharge to surface water is defined as a being no longer than 200 m.

hapua were the primary reason for the Wakanui system to be monitored (as determined at the consent hearing), it is considered that the most appropriate location downstream location to sample is at Cochranes Road. It must be noted however, that water quality and ecological conditions at this site will be impacted by urban input.

4.1.2.1 Upstream Monitoring Site

The upstream monitoring site for Wakanui Creek is located downstream from its intersection with Farm Road. Wakanui Creek forms the eastern border of Argyle Park at this site, located in North Ashburton, and is publicly and easily accessible. The site has stable banks, with a gentle gradient. Potential point-source discharges (likely small stormwater outfalls) are present on both the true right and left banks, which may be a source of urban contaminants to the site following rainfall events.

At the time of the site visit, the maximum water depth at the site measured approximately 20 cm and the maximum wetted width was 2.6 m. A reach length of 20 m was defined in order to sufficiently capture instream variation for most instream assessments. Suitable downstream habitat is available onsite for the establishment of a 50 m (i.e., 20x wetted width) reach for annual macroinvertebrate sampling. The upstream extent of the reach is located at the first tree on the true left bank when accessing the site from Farm Road. Photographs of the monitoring reach are shown in Figure D8-D9 (Appendix D).

The stream bed substrate composition was dominated (>50%) by cobble and gravel substrate types; however, high levels of fine sediment deposition (i.e., approximately 30% cover of sand/silt) was recorded throughout. Water was clear but slightly opaque; however, the stream bed was visible throughout.

4.1.2.2 Downstream Monitoring Site

As discussed above, the downstream monitoring site for Wakanui Creek flows parallel to Cochranes Road, approximately 5 km south-east of Ashburton. The site can be accessed by walking over the small bridge at the entrance to the dirt biking/4WD park at the terminus of Cochranes Road and travelling a short distance along the true right bank of Wakanui Creek in an upstream direction.

At this site, Wakanui Creek is channelised, with a steep built-up true-left bank and a lower true right bank with a gentle gradient. Bank vegetation is dominated by rank grass with interspersed weed species. Of note is the large amount of willow tree cuttings that have been placed throughout the length of the channel to encourage new growth. The willow tree cuttings have suspended flow at many points and will likely do so further in the future, with likely effects to aquatic ecology. Flow conditions are otherwise relatively uniform, consisting of shallow riffle/run mesohabitat types.

At the time of site visit, the maximum water depth at the site measured 13 cm and the maximum width was approximately 2.5 m. A reach length of 20 m was defined in order to sufficiently capture instream variation for most instream assessments and the majority of willow tree cuttings within the channel were avoided. The upstream extent of the reach is located approximately 95 m upstream from the access bridge to the dirt bike/4WD park. There is sufficient downstream habitat for the establishment of a 50 m reach (i.e., 20x wetted width) onsite for annual macroinvertebrate assessments. Photographs of the monitoring reach are shown in Figure D10-D11 (Appendix D).

At the time of the site visit, stream bed substrate composition was dominated (>50%) by cobble and gravel substrate types; however, the site also has a high cover of deposited fine sediment (approximately 40% cover) which will likely increase during warm, low flow periods when aquatic plant cover is likely to increase. The instream cover of macrophytes was approximately 10%, while long filamentous periphyton cover was approximately 30%. Water at the site was clear and colourless, and the stream bed was easily visible.

4.1.3 Mt Harding Creek

4.1.3.1 Upstream Monitoring Site

The upstream monitoring site on Mt Harding Creek runs parallel to Mt Harding Road, approximately 2 km north-west from the Methven town centre (lineal distance). The site runs parallel to a recently established footpath and banks have recently been planted out with native vegetation. Bank instability is evident from recent slips that have settled in the channel. The channel is channelised and incised, with steep banks measuring approximately 80 cm high. Flow conditions instream were relatively uniform, dominated by deep riffle/run mesohabitat types.

At the time of the site visit, the maximum water depth at the site measured 33 cm and the maximum width 1.9 m. A reach length of 20 m was defined in order to sufficiently capture instream variation for most instream assessments. Sufficient downstream habitat is available onsite for the establishment of a 40 m (i.e., 20x wetted width) reach for annual macroinvertebrate sampling. The upstream extent of the reach is located at the third fence post on the true left bank following the streams intersection with Mt Harding Road. Photographs of the monitoring reach are shown in Figures D1-D3 (Appendix D).

Stream bed substrate composition within the reach was dominated (>50%) by cobble and gravel substrate types. Flowing water had a milky-grey colouration and very low clarity, reflecting the conditions of the Ashburton River on the day (Mt Harding Stream is fed by the Ashburton River). Periphyton monitoring can only be conducted when water is flowing clear; therefore, it is recommended

that water clarity at the Ashburton River is assessed prior to monitoring to ensure that conditions are appropriate for sampling.

4.1.3.2 Downstream Monitoring Site

The downstream monitoring site for Mt Harding Creek is located at the end of Aikens Road (45 Aikens Road), approximately 14 km north-west of Ashburton. Coordination with the property owner will be required prior to accessing this site. A farm road continues on from the Aikens Road terminus through the property, towards the Ashburton River. Mt Harding Creek intersects the farm road at a bridge located approximately 250 m from the entrance to the property.

The selected monitoring site is located downstream from the farm road bridge, where there is currently no fencing, and the site is easily accessible. The site has mature native riparian plantings, interspersed with weedy exotic species, and has low banks of a steady gradient. The channel is straight and flow conditions relatively uniform, consisting of deep riffle/run mesohabitat types.

At the time of the site visit, the maximum water depth at the site measured 27 cm and the maximum width was approximately 5 m. A reach length of 25 m was defined in order to sufficiently capture instream variation for most instream assessments. The upstream extent of the reach is located approximately 10 m downstream from the farm bridge. There appears to be sufficient downstream habitat for the establishment of an up to 100 m reach onsite for annual macroinvertebrate assessments; however, this reach length can be truncated if required. Photographs of the monitoring reach are shown in Figures D4-D5 (Appendix D).

Within the monitoring reach, the stream bed substrate composition was dominated (>50%) by cobble and gravel substrate types. The flowing water had a milky-grey colouration and low clarity, reflecting the conditions of the Ashburton River on the day. Periphyton monitoring can only be conducted when water is flowing clear; therefore, it is recommended that water clarity at the Ashburton River is assessed prior to monitoring this site to ensure that conditions are appropriate.

4.1.4 Ashburton River South Branch at Digbys Bridge

Monitoring of the Ashburton River will be completed downstream from Digbys Bridge, which passes over the Ashburton River approximately 5.5 km north-west of central Ashburton. Access to the Ashburton River is available via a small dirt road that runs parallel to the bridge (east-to-west).

The Ashburton River is a braided river and, as such, has highly dynamic flow, channel positioning, and bed characteristics. On the day of site reconnaissance, Ashburton River flows were slightly elevated. As a result, water had a milky grey colouration and poor visibility, and was deep and swiftly flowing. The site was

not wadable and field staff were therefore not able to locate a suitable reach for instream periphyton monitoring. Whether this site will be suitable for instream periphyton monitoring at any time throughout the year is currently unclear, and more generalised bankside visual assessments may need to be conducted instead, with percent bed cover assessments, as outlined in section 4.2.2.1, completed when flow conditions allow for safe monitoring.

An approximately 50 m stretch of run habitat was identified as a suitable ongoing monitoring reach. Photographs of the monitoring reach are shown in Figure D6-D7 (Appendix D). The reach is located on the main braid, accessible from the true left river bank, beginning (upstream) 50 m downstream from Digbys Road Bridge. All ongoing water quality and ecological monitoring should be conducted over this reach. Proposed instream works at this site (i.e., annual macroinvertebrate monitoring) should target low flow conditions at least three weeks following any major rainfall event.

4.2 Sampling Methodology

4.2.1 Surface Water Quality Monitoring

On a monthly basis, the sampler will collect a single surface water grab sample for nitrate-nitrogen, dissolved reactive phosphorus, and the faecal indicator bacteria *Escherichia coli* (*E. coli*) at predetermined monitoring sites on each of the site locations listed in CRC185469. A list of required field equipment for the assessment of surface water quality is presented in Appendix E.

An International Accreditation New Zealand (IANZ) laboratory must be used for the analysis of samples. As per the procedure recommended by Hill Laboratories (an IANZ accredited laboratory), a sterile, unpreserved 400 mL PET container will be used to collect the sample for *E. coli* analysis, and an unpreserved 1,000 mL polyethylene container will be used to collect the sample for the remaining parameters. Appropriate sample containers can be sourced from Hill Laboratories. If another provider is used, ALIL must contact the laboratory to determine any differences in procedure.

Surface water quality monitoring shall follow the methodology outlined in the National Environmental Monitoring Standards (NEMS) Water Quality Part 2 of 4: Sampling, Measuring, Processing and Archiving of Discrete River Water Quality Data (NEMS-WQ2), dated March 2019. Recommended sampling methodology is detailed below, in accordance with the NEMS-WQ2 procedure.

1) A standard form will be used to record site metadata prior to sample collection on each sampling occasion. The following key parameters must be included:

    a) the name of the water body

    b) the name of the site location

c) any unique sample ID number(s) assigned

d) the name(s) of the field personnel

e) date and time (in NZST) of field measurements

f) the weather conditions at the time of measurements

g) flow state (i.e., 'low', 'moderate', 'high')

h) general sample colouration (clear/colourless, turbid or brown) and if the water has any unusual smell

i) the presence of any scums, foams or floatables

j) general notes of any other factors that may influence the data being collected; for example, potential waterfowl or stock influence, land-use changes, or channel alterations.

2) Samples can be collected by either wading into flowing water, with samples being collected from upstream of the sampler; or, for deep and/or swift water sites, samples may be collected from the bank, upstream from the sampler, using a telescopic water quality sampler (e.g., the 'Mighty Gripper'). To reduce risk of injury, it is recommended that the latter method is followed if sampling is to be completed by a single sampler.

3) It is important that samples are not contaminated by the sampler during the collection process; therefore, a new set of nitrile gloves are to be worn for the collection of samples at each sampling site. In addition, care should be taken to not touch the inner surface of the sample container or lid, and the sample container lid must be stored away from any sources of contamination (e.g., within a zip-lock bag) throughout the collection process.

4) The 1000 mL sampling container must be rinsed before sample collection. Rinsing requires the container be plunged into the water, approximately 20 cm below the surface, and rotated so that the opening faces upstream and allows the container to fill (Figure 3). The contents of the container are then discarded on the stream bank. This process is repeated three times to complete the rinsing process.

5) Following rinsing, the sample is taken by again filling the container following the same method. As the smaller, 400 mL container is sterile, this container does not need rinsing and can be filled immediately following the same methodology. Both sample containers must be filled completely, allowing for a small air gap for the sterile container only, and lids secured tightly.

6) After both samples have been collected, it is important that they are stored immediately in a chilled container before proceeding with further on-site assessments. During this process, exposure to direct sunlight must be avoided as far as practicable by the sampler, as UV exposure can

substantially impact *E. coli* counts and skew results. A large, insulated container (e.g., 'chilly bin') is recommended, with sufficient storage capacity for samples from multiple sites, as well as a sufficient number of pre-frozen icepacks or ice-slush to keep the samples chilled for up to 24 hours. Multiple chilly bins can be used if many samples are to be collected over a single day.

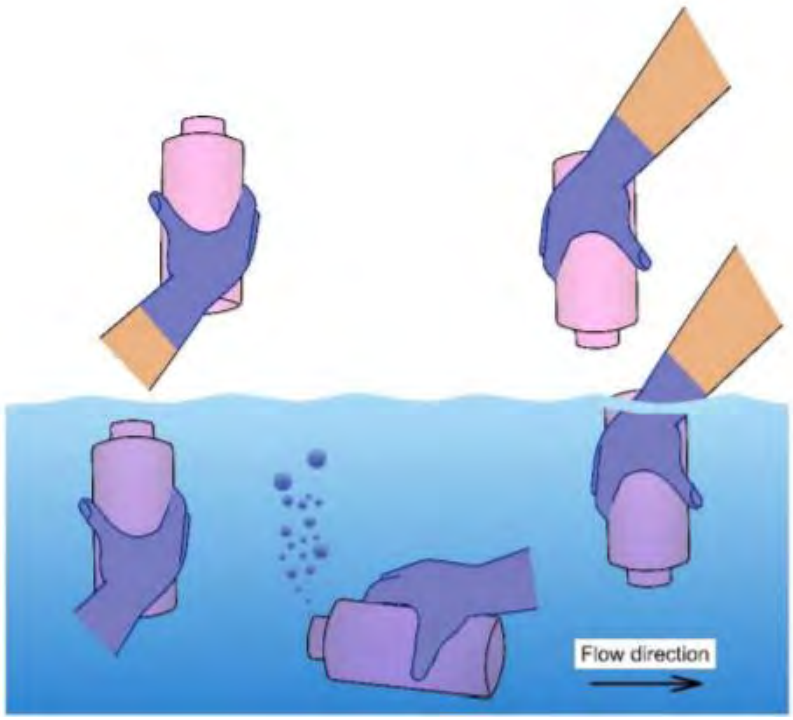

Figure 3. Recommended water quality sampling technique. Adapted from NEMS-WQ2 (2019).

7) For accurate measurement of *E. coli* counts to be achieved at the laboratory, it is required that samples are analysed within 24 hours of their collection time. It is thus recommended that, at the end of the sampling day, samples are delivered to the laboratory before the stated closing time. If samples cannot be delivered to the laboratory directly by the sampler, samples can be delivered to the laboratory using an overnight courier service. If the latter option is used, care should be taken by the sampler to ensure that the first sample is taken sufficiently after the stated opening time for the laboratory (i.e., >2 hours), thereby allowing sufficient time for analysis to be completed within the critical 24-hour timeframe. Each packed chilly bin weight should not exceed 25 kg to meet courier acceptance criteria.

8) A Chain of Custody (CoC) form must be competed and included with samples upon delivery to the laboratory. Standard format CoC forms are typically

available through the laboratory and will be included with the delivery of sample containers. The CoC allows the laboratory to identify what samples have been collected and when, which analyses are required for each sample, and to whom results should be sent following analysis. A copy of the Hill Laboratories CoC is provided in Appendix E. If an alternate provider is used, a CoC will need to be provided.

4.2.2 Periphyton and Stream Bed Properties

Following surface water quality sampling at each site, periphyton monitoring will be completed. As per Table CRC185469-2, this will include the assessment of periphyton percentage bed cover, as well as the analysis of chlorophyll-*a* biomass. An assessment of the stream bed composition and deposited fine sediment cover (<2 mm) will be completed concurrently. A list of sampling equipment required for each of these tasks is presented in Appendix E.

Periphyton and stream bed composition assessments will be conducted in accordance with the National Environmental Monitoring Standards (NEMS) Periphyton – Sampling and Measuring Periphyton in Wadeable Rivers and Streams (NEMS-Periphyton), dated November 2020. As per the guidance provided in the NPS-FM 2020, deposited fine sediment cover assessments will be completed following the procedure described in pages 17-20 of Clapcott et al. (2011). Key sampling procedures outlined in each of these guiding documents are detailed below.

4.2.2.1 Periphyton Visual Cover, Bed Substrate Composition, and Deposited Fine Sediment Assessments

This ecological sampling is specialised and will require a suitably qualified ecologist to either conduct the sampling, or train ALIL staff to a suitable standard. If ALIL choose to conduct this sampling (after appropriate training), ongoing auditing of field surveys will be required to provide assurance that results will be defensible.

1) At each of the predefined monitoring sites (see section 4.1.1), the monitoring reach (measuring 5-times the wetted stream width or 50 m (whichever is shorter)) will be partitioned into four evenly spaced transects, incorporating the full reach length, over which periphyton visual assessments will be conducted. The location of each premeasured transect should be marked on the stream bank using a wooden stake or a distinctive pile of stones. Chlorophyll-*a* assessments will also be conducted over these transects, but only the second and fourth furthest upstream transects will be used.
2) Beginning at the downstream-most marker, the assessor must wade into the stream on a 90° angle from the stream bank. The assessor will then estimate five evenly spaced monitoring points along the transect, covering the full

width of the stream, at which visual periphyton and deposited fine sediment assessments will be conducted.

3) At the nearest monitoring point, the bathyscope (underwater viewer) will be positioned upstream from the sampler and submerged up to 20 cm below the water's surface. Using the field measurement form provided in Appendix E, the percentage cover of each periphyton category should be recorded (see Figure 4 and Figure 5)[3]. In conjunction with the visual periphyton assessment, the bathyscope will be used to assess the percent cover of deposited fine sediment on the stream bed at each monitoring point. Repeat sequentially for each of the five monitoring points along the transect. Care should be taken to not veer from the transect when commuting between measurement points.

4) An assessment of substrate composition will then be completed at the transect scale. Substrate sizes can be estimated visually from the bank or in-stream; measurements are not necessary. At a minimum, this assessment will describe the percent cover of the following substrate size classes[4]:

   a) silt (<0.06 mm)

   b) sand (0.61-2 mm)

   c) gravel (2-64 mm)

   d) small cobble (66-128 mm)

   e) large cobble (129-256 mm)

   f) boulder (>256 mm)

   g) bedrock

5) Step 2-4 will be completed at each of the predefined transects, working sequentially in an upstream direction.

[3] The NIWA Periphyton Identification Guide provides a comprehensive guideline for categorising New Zealand periphyton types, and it is recommended that this document is taken into the field with the assessor (See https://niwa.co.nz/sites/niwa.co.nz/files/Periphyton%20ID%20Guide.pdf)

[4] Substrate size classifications should be given based on the 'B' axis of the stone (the second longest axis); this axis size indicates the smallest mesh size of which the stone would feasibly pass through.

Figure 4. Examples of the periphyton categories (with subcategory descriptions in brackets). Adapted from NEMS-Periphyton (2020).

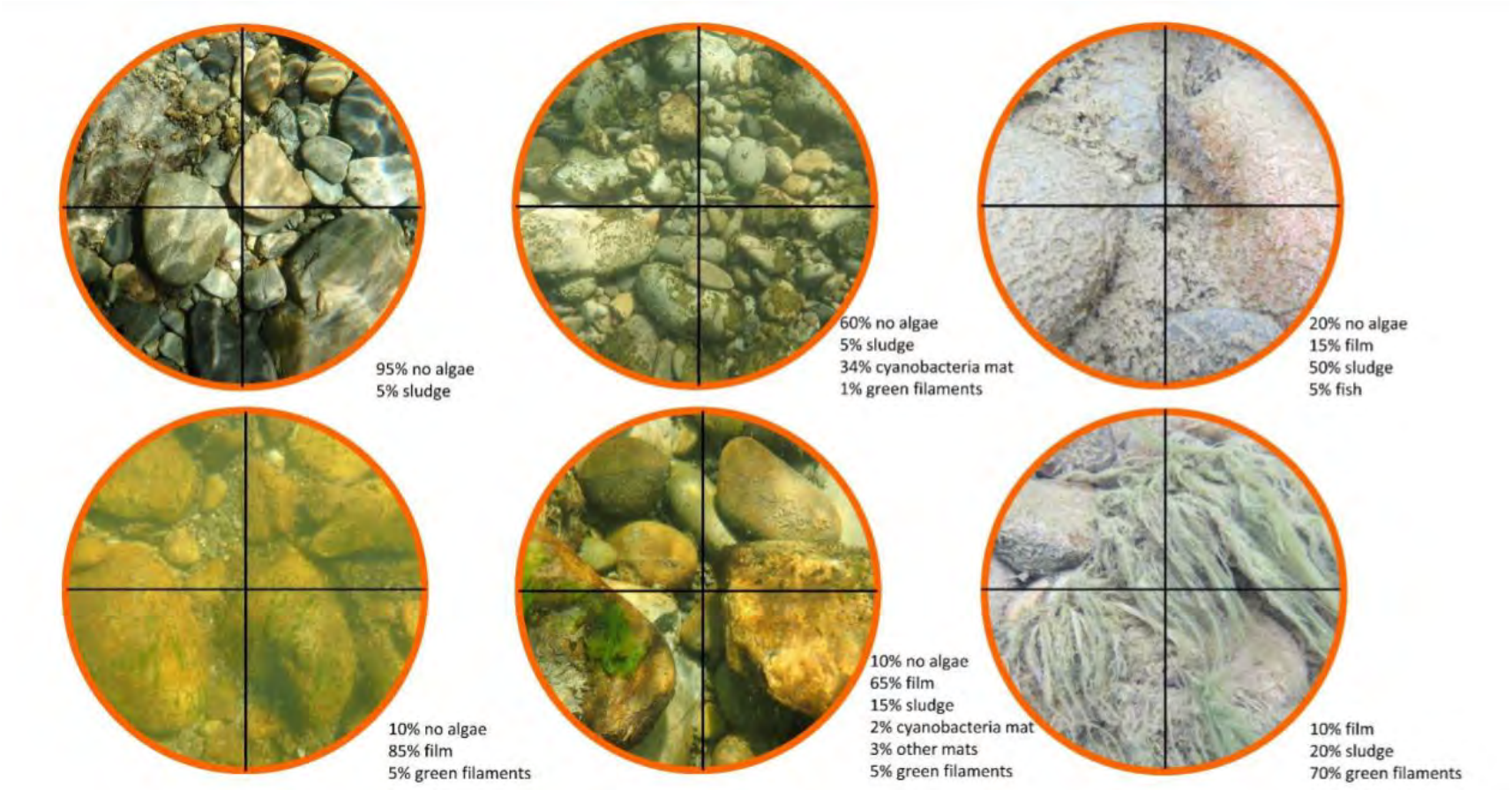

Figure 5. Examples of different levels of streambed periphyton cover. Adapted from NEMS-Periphyton (2020).

4.2.2.2 Periphyton Biomass Samples

1) Select two random transects along the reach length. To simplify the site selection process, it is recommended that the second and fourth transects (previously established for visual assessments) along the stream reach are used as a reference.

2) At the furthest downstream transect, access the stream on a 90° angle from the stream bank, approximately 1 meter upstream from the previously established transect line (to avoid the area disturbed from the visual periphyton assessment).

3) A single stone will be collected from five points spaced evenly across the full transect length. Care must be taken to ensure stones are collected randomly; as such, at each point along the transect, the sampler will place their index finger onto the stream bed without looking. The first stone that the index finger touches will be the stone from which the periphyton sample will be collected[5]. Stones should be placed in a large container, in the same orientation as they were in the stream and kept moist with a spray bottle.

4) Repeat steps 2-3 at the second transect.

5) Upon collection of ten stones from the stream bed, the sampler will collect a composite periphyton sample from the upper surface of each, covering a fixed surface area. This is completed by placing a ring (e.g., a sample bottle cap) of a known surface area onto a representative location on the upwards-facing surface of the stone. The material covering the upper surface of the stone (excluding the area covered by the cap) is then removed by scrubbing the stone with a wire brush. The dislodged periphyton sludge is then cleared using a spray bottle and discarded on the stream bank, or alternatively (and preferentially), it can be washed off by placing the stone into the flowing stream water (being careful to keep the ring secured in-place). Ensure residual water running off the stone runs clear before proceeding to the next step.

6) When the upper surface is clear from periphyton cover and residual water running off the stone is clear, the ring can be removed from surface of the stone, revealing a circular patch of periphyton. Over a large, clean, white plastic container, a second, clean wire brush is then used to dislodge the remaining circle patch of periphyton cover from the stone's surface. Ensure

[5] If the upper surface of the randomly selected stone is smaller than the surface area covered by the measuring ring, select the closest nearby stone of sufficient size. If all stones at the sampling point are smaller than the measuring ring (e.g., gravel bed streams), press the cap into the stream bed at an undisturbed area of the sampling point, scoop underneath the cap with paint scraper and collect all encapsulated material in a spare 400 mL (microbes) water quality sample container.

that when scrubbing the stone surface that all dislodged periphyton is captured within the large white collection container. Scrubbing will continue until all periphyton is dislodged from the stones surface. The spray bottle will be used to intermittently spray both the brush and the stone, until residual water runs clear. Care must be taken to ensure that all residual water is captured within the large white collection container. When the upper surface of the stone is cleared of periphyton cover, place the stone back into the stream.

7) Periphyton will be collected from smaller stones (collected within the 400 mL sample container) by one-quarter filling the sample container with water, securing the lid tightly, and vigorously shaking for 30-seconds. The resulting slurry is then decanted into the large white collection container. The small stones are then at least twice rinsed, by one-quarter filling the 400 mL sample container with water and gently swishing for approximately 10 seconds, before decanting into the large white collection container.

8) Repeat steps 7 and/or 8 on all gathered stones to form a composite sample of periphyton in the large white container.

9) Once complete, decant the contents of the large white collection container into a 500 mL sample container, spraying the internal walls of the container to ensure all dislodged material is removed.

10) Decontaminate the large white container and brushes by washing/spraying with stream water and detergent and using hands to dislodge any stubborn grime. Ensure all detergent is rinsed away with stream water after decontamination.

4.2.3 Macrophytes Cover Assessments

An assessment of macrophyte cover will be completed monthly as per Table CRC185469-2. Visual macrophyte assessments will be completed subsequent to the assessment of periphyton cover and biomass, at the scale of the full reach established in Section 4.1.1. The percentage of macrophyte cover will be assessed based on what is visible to the 'birds-eye'; that is, no attempt will be made to determine the level of cover of submerged macrophytes that are visually obscured by emergent macrophytes when viewed from above.

The assessment will be preferentially conducted from the bankside, with the assessor walking up the full reach length and estimating what proportion of the stream bed is obscured by the presence of either emergent or submerged macrophyte species. Care should be taken to ensure that the full wetted width of the stream is included in the visual assessment, ensuring that the cover of marginal species is appropriately captured. The proportionate cover of emergent and submerged macrophyte species is to be recorded separately.

4.2.4 Macroinvertebrates

On an annual basis during summer (December-March inclusive) low flow conditions, a benthic macroinvertebrate survey will be completed at each site following the National Environmental Monitoring Standards (NEMS) Macroinvertebrates – Collection and Processing of Macroinvertebrate Samples from Rivers and Streams (NEMS-Macroinvertebrates), dated November 2020.

Due to the specialised nature of this sampling, it must be completed by a suitably qualified freshwater ecologist. A second person will also be required for health and safety reasons (working in water). This second person can be from ALIL.

Sampling will be conducted no less than 14-days following any major stream flow that results in the following:

- significant bed-movement in a hard-bottomed stream (e.g., >25% of sampling habitat);
- >50% removal/scour of macrophytes, woody debris or other stable mesohabitat in a soft-bottomed stream; or,
- >50% movement of fine bottom sediments (e.g., sand and silt) greater than what is typical at baseflow.

Stream flows measuring >3-fold base flow conditions from a nearby representative flow monitoring station can be used as an indicator of major flow events at each monitoring site.

The following standard methodology will be followed for the collection of macroinvertebrate samples from each monitoring site:

1. Using a 100 m measuring tape, a monitoring reach will be defined at each monitoring site. The monitoring reach will preferentially measure 20-fold the wetted width of the watercourse at a representative site, with a minimum length of 20 m (for narrower sites) and a maximum length of 100 m (for wider sites). The monitoring reach will be inclusive of the 5-fold wetted width reach used for periphyton monitoring above.
2. The sampler will walk along the bank and take note of the proportionate cover of macroinvertebrate mesohabitat types within the reach.
3. The sampler will access the reach at the downstream end of the monitoring reach and move progressively upstream to ensure sampling areas are not disturbed by material dislodged from upstream.
4. All suitable macroinvertebrate mesohabitats present need to be included (e.g. riffle, run, bank margins, submerged woody debris, aquatic

macrophytes) in proportions equal to their presence across the monitoring site (reach).

5. The sample collection procedure is illustrated in Figure 6 for hard bottomed substrate and Figure 7 for soft-bottomed substrate and involves the following steps (modified from Protocols C1 and C2 of Stark et al., 2001, respectively).
6. Select a set area (0.1–0.15 m^2) of mesohabitat to sample (ideally in flowing water) using the following procedures.

- **Cobbles and gravels (in runs and riffles)** – place the kick-net on the streambed facing directly into the direction of flow to direct organisms into the net and disturb the surface substrate immediately in front of the net by foot-kicking (and by hand where necessary) to dislodge macroinvertebrates and detritus on and under the substrate and to scrape the underlying bed. *Note: For a kick-net 30 cm wide, disturbance of the streambed to a distance of approximately 30 cm in front of the mouth of the net represents an area of ~0.1 m^2. The disturbed distance in front of the mouth of the net should not exceed ~0.4 m to avoid organisms drifting outside of the net. Avoid scraping more than 10 cm below the bed and adjust kicking effort according to the degree of substrate embeddedness (e.g. loose gravel requires significantly less effort to turn over than large cobbles).*
- **Bedrock** – place the kick-net on the bed facing directly into the direction of flow to direct organisms into the net and disturb the surface of the bedrock immediately in front of the net by foot-kicking as well as by hand to dislodge macroinvertebrates and detritus.
- **Woody debris** – select a single, small point with submerged and partially decayed woody debris (>50 mm diameter preferred). Place the wood over the mouth of the bucket or sieve bucket. Pour water over the wood while brushing it gently by hand to remove organisms. Larger pieces of wood may be sampled in situ by brushing the log while holding the net directly behind it.
- **Bank margins** – locate a single (~0.5 m^2) area of bank with good structure and aggressively jab the net into the bank to dislodge organisms, followed by two cleaning sweeps to collect organisms in the water column.

- **Macrophytes (aquatic plants)** – jab at, or sweep the net through, a single point within a macrophyte bed on the flowing water side to dislodge organisms, followed by two cleaning sweeps to collect organisms in the water column. Alternatively, place the net downstream of the macrophytes and shake them by hand to dislodge the invertebrates.

a. Soft mud/sand/leaf litter – carefully and lightly disturb the top oxygenated layer of substrate immediately in front of the net with your hand or foot. *Note: Avoid deeply dredging the net through anoxic mud as well as sampling stagnant backwaters.*

7. Transfer the material from the net into a white tray, bucket or sieve bucket. Large fragments of macrophyte caught in the net can be picked out first and washed within the net to catch any macroinvertebrates. *Note: This is only required when the net begins to get clogged.*

8. Repeat steps 6 and 7 above until a total area of 0.6 to 0.9 m^2 has been sampled (i.e. typically between four and eight unit efforts depending on the net dimensions). *Note: For a hard-bottomed monitoring site comprising 80% runs and 20% riffles, the number of unit efforts collected for a 40 cm wide kick-net should be 3-4 and one in run and riffle mesohabitat, respectively.*

9. Holding the net in the water face up, remove any larger stones and vegetated material, ensuring any macroinvertebrates are first washed off into the net.

Figure 6. Kick-net sampling in a hard-bottomed stream. Photo: © EOS Ecology Taken from NEMS-Macroinvertebrates (2020).

Figure 7. Kick-net sampling in a soft-bottomed stream. Photos: ausrivas.ewater.org.au (left) and NIWA. Taken from NEMS-Macroinvertebrates (2020).

Sample Preservation and Sorting

After collection, rinse and remove any unwanted large debris items (e.g. stones, sticks, leaves) that may not fit into the sample container or will absorb and diminish the effectiveness of the preservative. Excessive gravel can be removed after tipping the sample into a bucket, stirring the content and floating off organic matter back into the net. This process may need to be repeated a few times. As far as practicable, rinse and remove any fine sediment prior to transferring to the sample container.

Then transfer the material to a pre-labelled sample container(s) via a 0.5 mm sieve if a sieve bucket is not used. Inspect the sieve or sieve bucket and return any macroinvertebrates to the sample container. The sample container should not be more than half full of material and should contain minimal amounts of water to prevent excessive dilution of preservative; use another container(s) for any remaining sample material.

To preserve the sample for sorting, isopropanol or ethanol preservative should be added to the sample container immediately upon completion and sorting of the sample. A 70–80% concentration is required, with an air gap of approximately 1 cm left for safe transport. Ensure the container lid is tightly sealed before gently inverting the container to mix the preservative throughout the sample. *Note: The container should not be more than half full of sample material and the amount of stream water present should be minimised to ensure that the preservative is effective. If a large amount of fresh organic matter (e.g. macrophytes) is present in the sample, it may be necessary to change the*

preservative after 24-48 hours to ensure the desired concentration is maintained prior to storage and dispatch.

An internal label (on waterproof paper) needs to be added to the sampling container and sampling details entered onto a Field Record Form, including records of the reach length, the types and proportions of mesohabitat sampled (e.g. 80% run and 20% riffle or 50% wood, 25% banks and 25% macrophytes), the collector's name, and preservative used.

Preserved macroinvertebrate samples should be stored in cool (< 15°C) conditions within a hazardous storage facility prior to dispatch to the laboratory for processing. The container seals should be checked prior to storage to ensure preservative has not been lost. It is recommended that samples are dispatched to the processing laboratory in a sealed, sturdy plastic bin promptly after collection where dedicated storage conditions are available. ids of all sample containers need to be tightly sealed, carefully packed, and the containers stored upright (vertical) to avoid leakage during transit. Each container should have a hazardous substances sticker on the outside. *Note: To avoid loss of sample, transport in cardboard boxes is not recommended. If plastic bins are used, they must be very sturdy and well-sealed to support the weight of sample containers without leakage. A courier approved for shipment of hazardous samples is required to transport the samples to the laboratory for processing. The courier ticket number(s) should be recorded on the Field Record Form to assist with prompt tracing of any bins lost or delayed in transit to the laboratory.*

A Chain of Custody (CoC) Form should be completed and inserted (within a sealed, waterproof bag) inside the bin and include a list of each sample in the shipment, including the sample site name, site number, the date and method of collection, the number of containers used and the name of the sampler.

5.0 Water Quality Database

ALIL shall maintain a water quality database to store all groundwater and surface water quality results from the monitoring required by CRC185469. The database should include the final field water quality measurements (including the date and time) and all laboratory results.

Results should be delineated by site and enable results for all parameters to be easily graphed, so that trends can easily be determined.

Results should typically be entered into the database as soon as they are received from the laboratory. That way any 'abnormal' or 'outlier' results can quickly be identified and, if necessary, ALIL can collect another sample to confirm these.

6.0 Deterioration in Surface Water Quality

As outlined in condition 25 of CRC185469, a 'Deterioration' in surface water quality must be investigated by a suitably qualified and experienced person and a Remediation and Response Plan must be prepared.

The criteria for determining whether there is a Deterioration is listed in Table CRC185469-2. The following contaminants have Deterioration criteria:

- Nitrate toxicity
- Dissolved reactive phosphorus (DRP)
- *Escherichia coli* (*E. coli*)
- Macroinvertebrates
- Deposited fine sediment (percentage cover)
- Periphyton (chlorophyll-*a*)

Before a Deterioration can be determined, ALIL must first establish the 'Base Attribute State' for each of these contaminants. As per Table CRC185469-2, the Base Attribute State is calculated from the first five years of monitoring data.

7.0 Annual Reporting

As outlined in condition 24 of CRC185469, ALIL must prepare an annual report by 1 December (starting 1 December 2022) that includes the following:

- Sampling results from the previous 12-month period;
- Discussion of any identified Deteriorations; and
- Discussion of any changes to periphyton and macrophytes percent cover over time, including any unexpected declines.

8.0 Review of this Monitoring Plan

As outlined in conditions 26 – 28 of CRC185469, this monitoring plan can be reviewed at any time by ALIL to add, remove or amend:

- Groundwater monitoring bores;
- Surface water monitoring sites;
- Contaminants;
- How a Deterioration is determined; and
- Sampling frequency.

ALIL are also required to review the environmental monitoring five years after the commencement date. Consent CRC185469 was granted on 28 June 2021, so a review will be required in the six-month period following 28 June 2026.

Finally, if the area managed by ALIL (as recorded in Schedule CRC185469A) increases by more than 200 ha from the commencement date, then a review is required.

Any review will require the preparation of a Water Monitoring Amendment Report (as detailed in condition 27(b) of CRC185469) and written approval from Environment Canterbury before any changes can be implemented.

9.0 References

Clapcott, J.E., Young, R.G., Harding, J.S., Matthaei, C.D., Quinn, J.M., and Death, R.G. 2011. Sediment assessment methods: protocols and guidelines for assessing the effects of deposited fine sediment on in-stream values. Cawthron Institute: Nelson, New Zealand.

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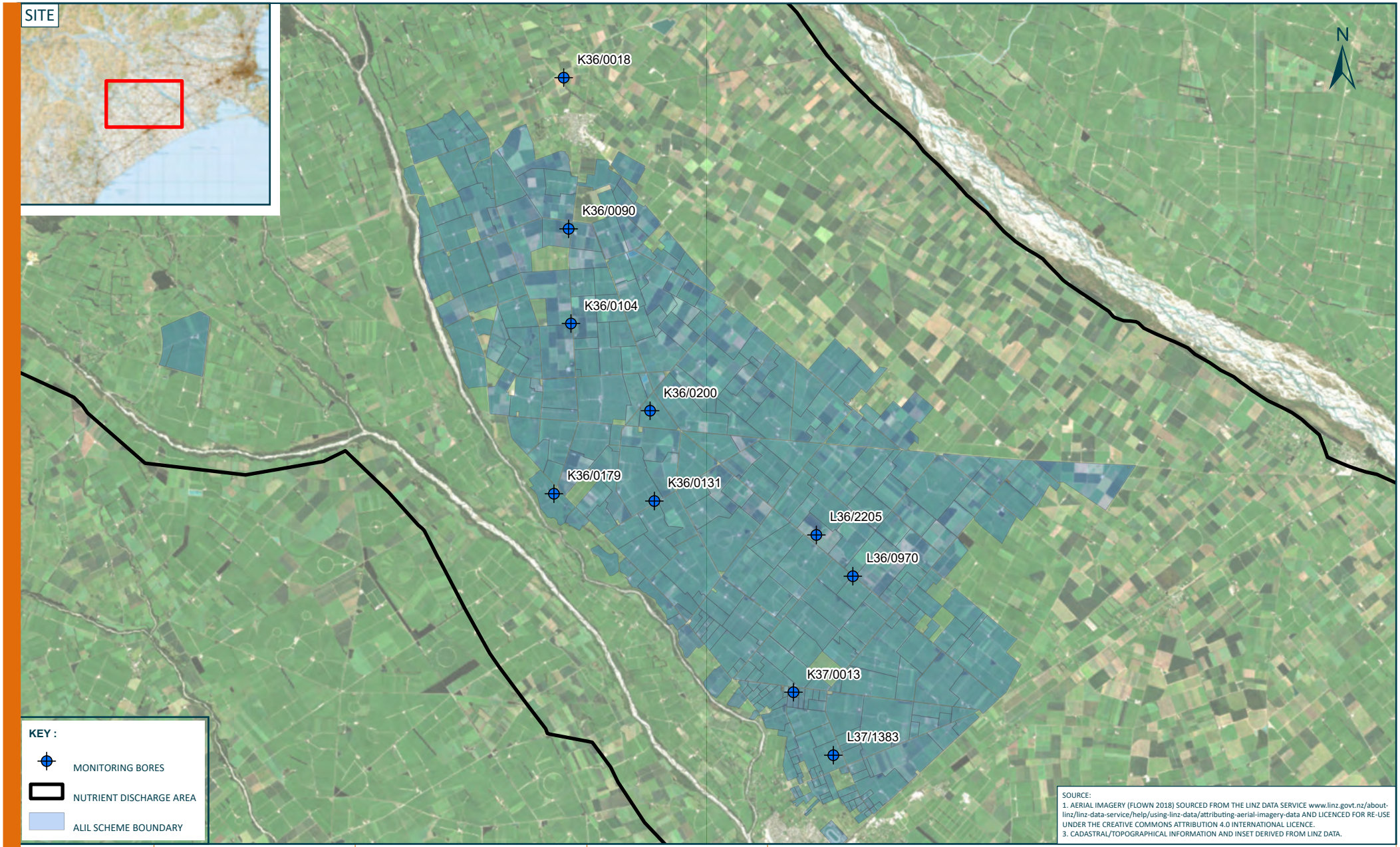
Stark, J.D.; Boothroyd, I.K.G; Harding, J.S.; Maxted, J.R.; Scarsbrook, M.R. 2001. *Protocols for sampling macroinvertebrates in wadeable streams*. New Zealand Macroinvertebrate Working Group Report No. 1. Prepared for the Ministry for the Environment. Sustainable Management Fund Project No. 5103. 57p.

Appendix A

Figures



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METRES

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A	ISSUED	DEC 21	CF

CLIENT

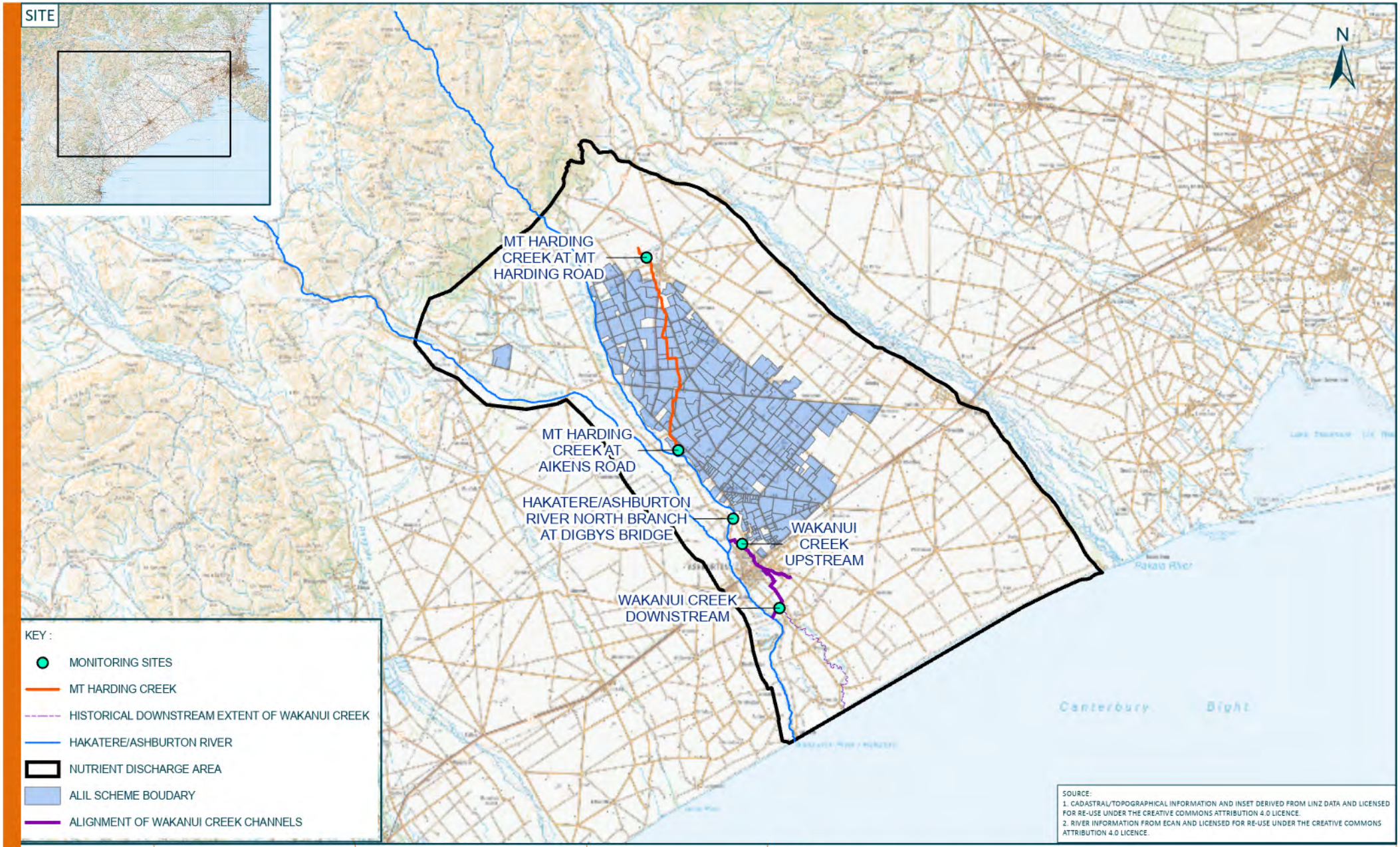
ALIL
ASHBURTON LYNDHURST IRRIGATION

FIGURE

FIGURE A1: GROUNDWATER MONITORING BORES

PROJECT

ALIL ENVIRONMENTAL MONITORING PLAN



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FOR ISSUE
20/12/2021

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CLIENT
ALIL
ASHBURTON LYNDHURST IRRIGATION

FIGURE
FIGURE A2: SURFACE WATER MONITORING SITES

PROJECT
ASHBURTON LYNDHURST IRRIGATION SCHEME

Appendix B

Photo of Groundwater Monitoring Bores

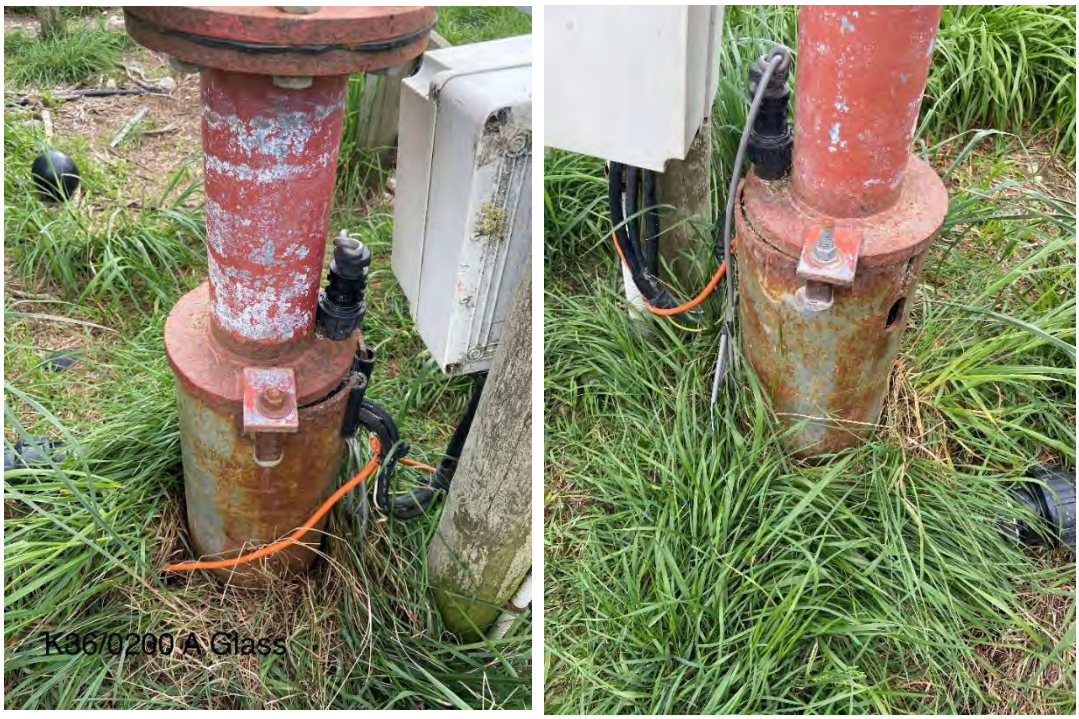
K36/0018 (52 m deep), located at Sivonholm Lodge - off Mount Hutt Station Road

K36/0090 (35 m deep), located at Methven Highway - near intersection with Reynolds Road

K36/0104 (24 m deep), located at Ashburton-Rakaia Gorge Road

K36/0131 (51 m deep), located at Braemar Lauriston Road

K36/0179 (9 m deep), located at Blands Road - near intersection with Methven Highway



K36/0200 (63 m deep), located at Pole Road

K37/0013 (30 m deep), located at 765 Winchmore Dromore Road

L36/0970 (72 m deep), located at Dromore Methven Road (Rapid # 765)

L36/2205 (67 m deep), located at 978 Dromore Methven Road

L37/1383 (40 m deep), located at 273 Mitcham Road

Appendix C

Groundwater Sampling Equipment

PATTLE DELAMORE PARTNERS LTD

PDP WELL PURGING AND SAMPLING FORM

Site: _____ Well ID: _____

Job Number: _____ Date(s): _____

Weather: _____ Sampler Name(s): _____

Purging method: _____

Sampling Equipment: _____

WELL DETAILS:

PID reading in neck of well: _____ (ppm)

Well casing diameter: _____ (mm)

Total Depth of Well: _____ (m)

Distance of PVC casing above/below ground level _____ (m)*

DEPTH TO WATER*:

Before Purging ("static water level"): _____ (m)

After Sampling: _____ (m)

Depth to Product: _____ (m)

Product Thickness: _____ (m)

Product measured by: _____ interface probe / bailer / product bailer

Volume of Product Removed: _____ L

*** Water Level Measurement**

Reference Point: Top of PVC Casing / Top of Well (circle as appropriate)

Toby Key Type: triangular? / allen key? / padlock?

Well Cap Type: H-cap? / screw cap? / push-fit?

Minimum Purge Volume (L): (**3** well vol.)

for 50mm dia. well= (total depth[m] - depth to water[m]) x 6
for non-50mm diameter wells see formula below.

NOTE: purge at least 3 well volumes **AND** until well has stabilized using field parameters below (or well is dry).

Key Stabilisation Criteria: pH ± 0.1, EC ± 3%, T ± 0.2 degree
Additional Stabilisation Criteria: DO ± 0.3 mg/L
Minimum volume between readings: 1/2 well volume

	Time Elapse	Time	Volume Removed (L)	Water Temp. (°C)	pH	EC ((µS/cm)	ORP (mV)	Dissolved Oxygen (mg/L)	Water Level (m)*	Water Appearance†
Before										
During										
During										
During										
During										
During										
During										
During										
During										
During										
During										
During										
After										

† CL=clear, CO=cloudy, TU=turbid, SI=silty, SA=sandy

Comments

Well Volume Calculation
1 well volume (L) = (total depth[m] – depth to water[m] x3.141 x d^2 / 4000
Where d = internal well casing (PVC) diameter in **mm**

1 Well Volume (L) =

Field Filtered (metals only)?: Y / N

Analyses Required:

Sample Bottles Collected:

Lab Quote No.

* = needs to be recorded each time you take a set of parameters

Annex D – Example Chain of Custody Form

Note: This form is a guide only and will need to be modified in consultation with the laboratory provider.

Chain of Custody Form for Water Samples

Client Name _____ **Client Contact** _____

Client Reference _____ **Email*** _____

Quote/Order No. _____ ** For return of CoC*

Client to complete

Sample Dispatch Date _____ Time (NZST) _____

Name _____ Signature _____

Additional Notes _____

_____

_____

Laboratory to complete

Sample Arrival Date _____ Time (NZST) _____

Name _____ Signature _____

Sample Condition o Room temperature o Chilled o Frozen

Temperature _____ °C

Comments *(e.g. appropriate bottle used, headspace requirements met)*

_____

_____

_____

Job. No. _____

Sample Details *(Client to complete)*

Type o Groundwater o River / Stream o Lake o Estuarine / Coastal

Have samples been field filtered? o Y o N Details _____

No.	*Sample Name*	*Sample Date & Time (NZST)*	*Tests Required*
1			
2			
3			
4			
5			
6			
7			
8			
9			
10			

Groundwater Sampling Equipment List

- Sample bottles
- Sampling procedure
- Field purging and water quality form (see Appendix C)
- Field water quality meter and calibration solutions
- Clean vessel for measuring water quality parameters in (i.e. a 3 L plastic measuring jug) or a flow cell
- Sample tubing and appropriate tap/tubing fittings
- Water level probe for measuring groundwater level
- 10 or 20 L bucket with volume measurements (i.e. 1 L increments)
- Nitrile gloves (powder-free latex)
- Toolbox, including basic tools such as screwdrivers, adjustable spanners, hammer, pliers, groove joint pliers, pipe/Stillson wrench, electrical tape, thread tape)
- Assorted plumbing fittings, hose clips and plugs
- Calculator
- Stopwatch
- Pen, pencil and permanent marker
- Cell phone
- First aid kit
- Paper towels
- Hand sanitizer
- Zip-lock plastic bags (for grouping bottles from each monitoring site)/double bagging loose ice in chilly bin
- Chilly bins for storing/transporting samples
- Ice and/or chemical cooling packs (i.e. slicker pads etc) for cooling samples in chilly bin
- A groundsheet/tarpaulin (if the area around the bore is muddy/dusty)

Appendix D

Photo of Surface Water Sites

Figure D1: Mt Harding Stream at Mt Harding Road (upstream). View looking downstream from the upstream end of the monitoring reach.

Figure D2: Mt Harding Stream at Mt Harding Road (upstream). View looking upstream from the downstream end of the monitoring reach

Figure D3: Mt Harding Stream at Mt Harding Road (upstream). Bank erosion on the true right bank.

Figure D4: Mt Harding Stream at Aikens Road (downstream). View looking downstream from the upstream end of the monitoring reach.

Figure D5: Mt Harding Stream at Aikens Road (downstream). View looking upstream towards the bridge from the upstream end of the monitoring reach.

Figure D6: Ashburton River at Digbys Bridge. View looking downstream from the upstream end of the monitoring reach.

Figure D7: Ashburton River at Digbys Bridge. View looking upstream towards Digbys Bridge from the upstream end of the monitoring reach.

Figure D8: Wakanui Stream at Farm Road. View looking downstream from Farm Road bridge - upstream end of the monitoring reach starts at the first large tree on the true left bank.

Figure D9: Wakanui Stream at Farm Road. View looking upstream towards Farm Road bridge - downstream end of the monitoring reach.

Figure D10: Wakanui Stream at Cochranes Road. View looking downstream towards the footbridge from the upstream end of the monitoring reach. Site chosen to avoid the majority of willow trunks placed on the true right bank.

Figure D11: Wakanui Stream at Cochranes Road. View looking upstream from downstream end of the monitoring reach. Note willow trunks on true right bank.

Appendix E

Surface Water Sampling Equipment

Surface Water Sampling Equipment List

General and PPE:

1) Waders, life jackets and other personal protection equipment
2) Waterproof marker pens and pencils
3) Cool, dark storage for samples (e.g. Chilly bin with ice packs or slush)
4) Disinfectant and associated gear for completing “check, clean, dry” procedures
5) Digital camera
6) GPS receiver
7) 50 m measuring tape
8) Metal metre ruler

Job specific equipment for water quality sample collection:

1) Field record forms to record visit metadata and field measurements
2) A fixed-length or telescopic sampling pole (i.e., ‘mighty gripper’)
3) Sample bottles
4) Nitrile gloves (1x pair per site)

Job specific equipment for periphyton assessments and sample collection:

1) Field forms to record site and visit metadata and field measurements
2) Underwater viewer (figure 1) for assessing periphyton cover
3) A circular ring(s) or equivalent (e.g. Sample container lid) of known diameter(s) between 30 and 70 mm
4) A thin sheet of stiff plastic or a metal spatula large enough to completely cover the sampling ring when sampling gravelly and/or sandy substrate
5) Blade(s) and scissors for scraping/cutting off thick algae
6) Small scrubbing brushes for scrubbing thin, tightly attached algae (e.g. Wire brush or firm toothbrushes)
7) Two or three open containers (e.g. 2-litre square food containers, deep-sided laboratory trays)
8) Squirt bottle containing stream water
9) Small disposable pipettes, and sample containers of 400 mL to 1,000 mL volume with watertight lids

Job specific equipment for macroinvertebrate sample collection:

1) Field record forms to record visit metadata and any field measurements
2) D-framed hand (or kick) net (0.5 mm mesh)
3) Cut resistant gloves
4) White plastic tray
5) Sieve/sieve bucket (0.5 mm mesh)

6) Tweezers (for picking off invertebrates caught on the net)
7) Water bottle (for rinsing material from the sampling net)
8) Plastic screw-top sample containers (500–1000 mL volume)
9) Isopropyl alcohol or ethanol (>500 mL per sample, for preservation of macroinvertebrate samples)
10) Waterproof sample container labels (inner container labels)
11) 100 m measuring tape (for measurement of sampling reach)
12) Rapid habitat assessment forms

Hill Laboratories
TRIED, TESTED AND TRUSTED

ANALYSIS REQUEST

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Private Bag 3205
Hamilton 3240, New Zealand

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T +64 7 858 2000
E mail@hill-labs.co.nz
W www.hill-laboratories.com

Office use only
(Job No)

Quote No

Primary Contact

Submitted By

Client Name

Address

Postcode

Phone *Mobile*

Email

Charge To

Client Reference

Order No

Results To *Reports will be emailed to Primary Contact by default. Additional Reports will be sent as specified below.*

☑ *Email Primary Contact* ☐ *Email Submitter* ☐ *Email Client*

☐ *Email Other*

☐ *Other*

CHAIN OF CUSTODY RECORD

Sent to Hill Laboratories
Date & Time:
Name:
☐ *Tick if you require COC to be emailed back*
Signature:

Received at Hill Laboratories
(Refer to Lab created Job No above)
Date & Time:
Name:
Signature:

Condition
☐ *Room Temp* ☐ *Chilled* ☐ *Frozen*
Temp:

ADDITIONAL INFORMATION / KNOWN HAZARDS

Priority ☐ **Low** ☑ **Normal** ☐ **High**
☐ **Urgent** (ASAP, extra charge applies, please contact lab first)

Requested Reporting Date:

No.	Sample Name	Sample Date	Sample Time	Sample Type	Tests Required (if not as per Quote)
1					
2					
3					
4					
5					
6					
7					
8					
9					
10					
11					
12					

Continued on next page

No.	Sample Name	Sample Date	Sample Time	Sample Type	Tests Required (if not as per Quote)
13					
14					
15					
16					
17					
18					
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21					
22					
23					
24					
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