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Adelaide Desalination Project

Plankton Characterisation Study - Phase 2

Paul van Ruth

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March 2012

Prepared for Adelaide Aqua

Government of South Australia

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1. Executive Summary

Adelaide Aqua has constructed and commissioned a seawater reverse-osmosis desalination plant at Port Stanvac in Gulf St Vincent (GSV) with a targeted 100 GL per annum production of fresh water. This development aims to provide a reliable potable water supply for the city of Adelaide, reducing the need to draw water from the River Murray. The first water was delivered from the plant in August 2011, but it is not yet operating at full capacity. Studies assessing the impact of desalination plants on marine ecosystems recognise that the intake of seawater can entrain plankton species. The brine discharge from the desalination process also affects marine ecosystems, ultimately leading to changes in species composition, species diversity and population density which may promote the emergence/dominance of opportunistic or harmful species.

Phytoplankton form the primary food source in oceanic food webs, underpinning marine ecosystems. Herbivorous zooplankton feed on phytoplankton, and are in turn preyed upon by carnivorous higher consumers such as ichthyoplankton and fish. Phytoplankton are thus gradually transformed into products available for consumption by higher trophic levels in marine food webs. The risks that entrainment of plankton and increased salinity from brine discharge pose to the marine environment off Port Stanvac, and the food web it supports, require investigation to ensure that any environmental impacts due to operations of the Adelaide Desalination Project (ADP) are minimised or prevented. This study was designed to characterise the plankton communities in the area around the intake and outlet pipes to be used by the ADP. It is the second phase in the ADP plankton characterisation study (PCS) and will be used together with information from phase 1 (van Ruth 2010) to conduct a preliminary examination of interannual variation in plankton dynamics in the Port Stanvac region.

Temporal patterns in phytoplankton, zooplankton and ichthyoplankton dynamics observed in 2010-11 differ from those identified in 2009-10 as regular seasonal patterns for the region(van Ruth 2010). There were differences in temporal patterns of phytoplankton abundance and community composition, including the presence and abundance of potentially harmful/toxic algae species, and differences in zooplankton and icthyoplankton abundances and community composition. The cycles of primary productivity and phytoplankton growth, and zooplankton grazing pressure were also different to those reported in 2009-10 (van Ruth 2010), suggesting that changes have occurred to the food web in the waters off Port Stanvac. In both phases of the study a summer increase in potentially harmful cyanobacteria appears to be a characteristic of the region. Ongoing monitoring of cyanobacterial populations off Port Stanvac via HPLC pigment analysis is recommended given that increased salinity in the vicinity of the ADP outflow structures has the potential to promote cyanobacterial blooms.

Monitoring the plankton community in the waters off Port Stanvac should be continued to assess the impact of the working desalination plant. It is possible that activities associated with the working plant, such as increased entrainment in the intake structure and brine discharge, will further change the ecology of the plankton in the region, and may promote blooms of harmful/toxic algal species which are identified as components of the local plankton community. The paucity of information available on temporal variation plankton community dynamics and food web structure in GSV dictate that further monitoring is required before the impact of the ADP on the plankton community off Port Stanvac can be adequately assessed. This will ensure that any variation in cycles of plankton abundance and biomass in the

region is properly understood so that changes in these parameters can be attributed to the correct cause, be it natural cyclical variation or impacts of the desalination process. Further, it will help to produce information and strategies that alleviate impacts and thereby ameliorate community concerns about the environmental impact of industrial seawater intake, and aid in the development of strategies to minimise and mitigate the impacts of the desalination process.

2. Introduction

The security of Adelaide's metropolitan water supply is affected by climate variability and by Adelaide's limited water storage capacity (approximately one years' demand). With the impact of anticipated climate change and increase in the frequency of extreme events including droughts likely to lead to greater variability and reduced average flows to local reservoirs (Garnaut, 2011), water security has become an issue of critical significance. In March 2007, the South Australian Minister for Water Security announced the formation of a Desalination Working Group to investigate implementation of desalination technology for the State to reduce reliance for water supply on the River Murray. Adelaide Aqua subsequently constructed a seawater reverse-osmosis desalination plant at Port Stanvac in Gulf St Vincent (GSV), which aims to provide 100 GL per annum of potable water for the city of Adelaide. First water (and associated brine discharge) was produced by the plant in August 2011 but the plant is still operating substantially below maximum production.

Desalination plants can entrain plankton (Latteman and Hopner, 2008). Modern intake designs attempt to minimise the chance of motile species being drawn into the feedwater by not placing intake structures in known nursery areas, covering intake structures with fine (~5 mm^2) mesh (Morton et al., 1996) and reducing intake velocity to equivalent to ambient currents. Animals that can swim against prevailing currents are therefore unlikely to be drawn into the desalination plant. Smaller, less motile organisms such as phytoplankton and smaller zooplankton, however, are still at risk of entrainment through the mesh of the intake structure (Latteman and Hopner, 2008), and the removal of these important organisms from the water column may have impacts on food web structure (Morton et al., 1996).

The brine discharge from the desalination process also affects marine ecosystems (Miri and Chouikhi, 2005; Latteman and Hopner, 2008). This discharge is of higher salinity than surrounding waters, and may contain chemicals and additives associated with the desalination process (such as antifouling/anti-scaling substances, biocides (chlorine), coagulants, heavy metals, and nutrients (nitrogen and iron)). Changes in salinity can affect phytoplankton physiological processes (Qasim et al., 1972; Miller and Kamykowski, 1986; Flameling and Kromkamp, 1994) and fish egg and larval development (Boeuf and Payan, 2001; Varsamos et al., 2005), ultimately leading to changes in species composition, species diversity and population density. The addition of nitrogen and iron to the water column may affect primary productivity since these nutrients are potentially limiting to phytoplankton growth (Martin and Fitzwater, 1988; Martin et al., 1989; Martin, 1990) These changes may promote the emergence/dominance of opportunistic or harmful species at heavily impacted sites (Miri and Chouikhi, 2005).

Phytoplankton are responsible for more than 95% of marine photosynthesis (Falkowski and Kolber 1995). Their health and productivity underpins marine ecosystems because they form the primary food source in oceanic food webs. Herbivorous secondary producers feed on phytoplankton, and are in turn preyed upon by higher consumers such as ichthyoplankton and fish. Light energy harnessed by the phytoplankton is thus transformed into products available for consumption by higher trophic levels in the marine food chain. All energy in food webs ultimately originates in photosynthetic organisms.

The risks posed by entrainment and increased salinity on regional food webs and ecosystem services including commercial and recreational fisheries require

investigation. This study provides data to characterise the phytoplankton, zooplankton and ichthyoplankton communities around the Adelaide Desalination Project (ADP) intake and outlet pipes, quantify the temporal variation in these communities, and quantify variation in primary and secondary productivity in the waters off Port Stanvac. This information will assist in identifying any risks posed to plankton communities and food web structure by the industrial works of the ADP by highlighting changes in seasonal/annual/interannual cycles. This is the second phase of the ADP Plankton Characterisation Study (ADP PCS). The first phase of the study described the temporal variation in plankton abundance, composition and productivity over a 12 month period at the Port Stanvac ADP site (van Ruth, 2010). This second phase is intended to continue these analyses toward the full commissioning of the Adelaide Desalination Plant. It provides information on interannual variability in plankton dynamics in the area of the ADP and help to quantify the impacts of the ADP construction process on plankton community composition and food web structure. The objectives of this study were to:

1. Identify plankton (phytoplankton, zooplankton and icthyoplankton) to the lowest taxonomic group possible
2. Asses the abundance, composition and temporal (intra- and inter-annual) variability of plankton
3. Examine seasonal variation in rates of primary and secondary productivity
4. Assess potential impacts of entrainment of plankton species and the presence of brine discharge on local marine scalefish, lobster, abalone, prawn and blue crab fisheries and protected fish taxa
5. Assess the impacts of entrainment of plankton and the presence of brine discharge on the local food web

This report provides Adelaide Aqua with detailed results of these surveys and an analysis of the likely environmental impacts to the plankton community of a desalination plant, to support robust decision making about management of the desalination plant's operation to support sustainable production of potable water for Adelaide.

3. Methods

Sampling took place from December 2010 to November 2011 at three sites: the intake pipe (station IRP), outlet pipe 4 (station ORP-4), and station PP situated midway between IRP and ORP-4 (Fig. 1). Each site was sampled eight times, 5 in summer/autumn (December 2010 – April 2011) and three in winter/spring (August, October and November 2011). Additional samples for seasonal studies of primary productivity were collected from station PP in January, April, August and October.

A Seabird SBE 19-plus conductivity, temperature and depth recorder (CTD) fitted with a Biospherical QSP-2300 underwater Photosynthetically Active Radiation (PAR) sensor with log amplifier (Biospherical Instruments Inc., San Diego, CA, USA) was used at each station during each sampling trip to provide information about sea surface temperature, salinity and irradiance for use in determinations of primary and secondary productivity. Samples for pigment analysis, phytoplankton community analysis, and investigations of primary productivity were collected at midday. Zooplankton and icthyoplankton samples were collected at midday and midnight, recognising that the abundance and species composition of these groups may display high diurnal variability.

Figure 1. Station locations for sample collection for the plankton characterisation study. IRP = 138.4679 E 35.09059 S; ORP-4 = 138.47129 E 35.09255 S; PP = 138.46961 E 35.09158 S; (WGS 84). Green line outlines Adelaide Desalination Project site location.

3.1 Phytoplankton biomass, abundance and community composition

The pigment composition of water samples was measured using High Pressure Liquid Chromatography (HPLC). At each station three litre samples were collected from 3 m depth, without replication, with a Niskin bottle and kept cool in the dark and returned to the laboratory within 3 hours. Samples were filtered through stacked mesh (to retain cells >5 µm) and Whatman GF/F filters (nominal pore size 0.4 µm, to retain cells <5 µm), allowing the examination of size fractionated phytoplankton biomass. Filters were frozen and stored at -80°C prior to HPLC analysis in the environmental chemistry laboratory at SARDI Aquatic Sciences.

A detailed inventory of taxa and their cell abundances was obtained from one litre samples collected with a Niskin bottle from 3 m depth, without replication, at midday at each station on each sampling trip (= 3 samples per trip). Samples were fixed with acidified Lugol's iodine solution. Enumeration and identification of phytoplankton to genus or species level was carried out using light microscopy by Microalgal Services, Victoria, Australia. Species richness (S) was measured as the number of species in a sample unit.

3.2 Zooplankton biomass, abundance and community composition

The structure of the zooplankton community was determined from samples collected via net tows with a 50 µm mesh net (30 cm net mouth diameter) lowered to within 1 m of the bottom and towed vertically to the surface. One sample was collected from each station on each sampling trip (= 1 midday sample from each station on each trip, and 1 midnight sample from each station on each trip). For each sample, the contents of the net were washed into a sample jar, topped up to 1 litre, and fixed with formalin (5% final volume). In the laboratory, samples were rinsed through a 35 µm mesh sieve. The contents of the sieve were rinsed into 100ml measuring cylinders and allowed to settle for 24 hours, after which settling volumes were recorded. Samples were then decanted into 120 ml jars and resuspended in 100ml of water (i.e. concentrated 10x). Samples were viewed, identified and enumerated with a compound microscope. After gently resuspending the sample, a 1 ml aliquot was taken for counting using a Sedgewick-Rafter chamber. Counts were continued until 100 specimens of the dominant taxa were counted. Organism numbers were recorded as individuals m^{-3} in the water column using the volume swept by the net, calculated as the depth of the tow multiplied by the area of the net mouth. Settling volumes were recorded as ml m^{-3} using the volume swept. Settling volumes were converted into displacement volumes using a factor for samples without gelatinous zooplankton (0.35, see Wiebe *et al.* 1975; Wiebe 1988). Displacement volumes were then converted to biomass (mg C) using a factor of 21 for samples with displacement volumes < 1 cm^3, and a factor of 41 for samples with displacement volumes 1-10 cm^3 (Bode *et al.* 1998). S was calculated as above.

3.3 Ichthyoplankton abundance and community composition

To assess abundance of fish larvae and eggs, five samples were collected from each station on each sampling trip (= 5 midday samples from each station on each trip, and 5 midnight samples from each station on each trip), using net tows with a 350 µm mesh bongo net (30 cm net mouth diameter). The net was lowered to ~mid-water depth (~10 m), towed 50 m horizontally and then brought to the surface. Flow

meter suspended in the mouth of the nets recorded distances travelled during sampling. While this technique predominantly sampled mid-water plankton, some surface plankton were collected as the net was retrieved. The contents of the nets were pooled into a sample jar, topped up to 1 litre with seawater, and fixed with formalin (5% final volume). The five samples from each station were pooled during rinsing through a 35 µm mesh sieve. The entire sample was sorted under a dissecting microscope at up to 60x magnification. Egg and larvae numbers were recorded as individuals m^{-3} in the water column using the volume swept by the net, calculated using distances travelled by the net multiplied by the area of the net mouth. Fish larvae and eggs were identified using Leis and Trnski (1989) and Neira *et al*. (1998).

3.4 Primary productivity

Primary productivity in the water column was measured based on methods outlined by Parsons et al. (1984), Lohrenz et al.(1992), and Mackey et al. (1995). Three independent 2 litre samples were collected from 3 m depth at midday at station PP in August and October 2009, and January and April 2010. Samples were collected in opaque bottles and kept cool with light excluded until return to the laboratory within 3 hours. The samples were then exposed to light. Seven irradiance levels were used, by modifying the amount of natural sunlight reaching the experimental bottles via shading with shade cloth. Irradiances included 0% (dark), 0.4%, 1.2%, 1.5%, 6.5%, 50%, and 100% of natural sunlight. From each independent water sample, 1 x 250ml polycarbonate bottle was prepared for each irradiance level. A ^{14}C stock solution with an activity of 200 µCi ml^{-1} was prepared by adding 1ml of sodium bicarbonate (GE Life Sciences $NaH^{14}CO_3$, 2 mCi ml^{-1}, 55 mCi $mmol^{-1}$) to 9 ml of Na_2CO_3 solution (concentration 0.12 g L^{-1}). A known quantity of $NaH^{14}CO_3$ (20 µCi) was mixed into each replicate bottle via the addition of 0.1 ml of ^{14}C stock solution. Bottles were then incubated in a flow-through water bath for 24 hours at *in-situ* water temperatures in sunlight. Irradiance was measured every minute with a Licor Li-1400 data logger and quantum sensor, with the mean irradiance logged every 30 minutes over the 24 hour period, then integrated to provide daily integral irradiances. Post-incubation, samples were filtered at low vacuum pressure through 25mm Whatman GF/F filters, rinsed with filtered seawater, placed into 5ml scintillation vials and frozen until further analysis. Filters were thawed at room temperature and exposed to 200µl of 5N HCl for 12 hours to drive off any remaining $^{14}CO_2$. Four millilitres of scintillation fluid (Ultima Gold high flashpoint LSC cocktail) was then added to each vial and, after 24 hours, radioactivity was determined as disintegrations per minute using a scintillation counter (Packard Tricarb 2100TR). Total CO_2 concentration in the samples was estimated from salinity using the method of Parson et al. (1984). Measured photosynthetic rates were fitted to the hyperbolic tangent equation of Jasby and Platt (1976):

$$P^b = P^b_{MAX} * \tanh(\alpha * I / P^b_{MAX}) \qquad (1)$$

Where P^b_{max} is the maximum biomass specific photosynthetic rate, α is the photosynthetic efficiency, and I is irradiance. Rates were fitted to the above equation in Microsoft Excel using Solver to provide estimates of α, P^b_{max}, and irradiances corresponding to the onset of light saturation of photosynthesis (I_k). These data were used to examine seasonal variations in daily integral productivities according to Tallings model (Talling 1957):

$$\sum P = P^{b}_{\max} / k_d * Ln(I'_o / 0.5I_k) \quad (2)$$

Where ΣP is the integral productivity, P^{b}_{max} is the maximum specific photosynthetic rate, I'_o is photosynthetically active radiation (PAR) available just below the sea surface (as measured by CTD), k_d is the attenuation coefficient of downwelled irradiance (the slope of the line when Ln PAR is plotted against depth). Integral productivity was multiplied by daylength (D_{irr} obtained from astronomical information on the Geoscience Australia website (www.ga.gov.au/geodesy/astro)) and a correction factor of 0.9 to compensate for the decreasing incoming irradiance either side of solar mid-day to provide daily integral productivity (Talling 1957). Phytoplankton turnover time was calculated as standing stock of Chl *a* (determined using a Carbon to chlorophyll ratio of 40:1) over maximum photosynthetic rate (P_{max} = P^{b}_{max} * Chl *a*). The gross phytoplankton growth rate was calculated as the inverse of the turnover time.

3.5 Secondary productivity

Meso-zooplankton grazing pressure was estimated from zooplankton biomass. Potential growth of the meso-zooplankton was estimated via the empirical relationship of Huntley and Boyd (1984):

$$G'_{\max} = 0.0542e^{(0.110T)} \quad (2)$$

Where T is temperature (CTD measured sea surface temperature (SST)) and G'_{max} is the maximum mass-specific food-saturated growth rate, which can be used to estimate the assimilative capacity (AC) of the meso-zooplankton community via:

$$AC = 0.7G'_{\max} \quad (3)$$

Where 0.7 is the estimate of 70% assimilative efficiency proposed by Conover (1978). The assimilative capacity was multiplied by biomass to give an estimate of the potential grazing rate of the meso-zooplankton community.

4. Results

4.1 Phytoplankton biomass, abundance and community composition

Temporal patterns in total Chl *a* concentrations were similar for all stations sampled (Fig. 2). Chl *a* concentrations were generally between 0.2 and 0.4 µg L^{-1}, with a peak of ~0.6 µg L^{-1} in October 2011. There was a general decline in Chl *a* concentrations from December 2010 into January 2011 with a gradual increase through February/March/April. Concentrations decreased to a low of ~0.2 µg L^{-1} in August 2011, rising to the October peak before a sharp decline back to ~0.3-0.4 µg L^{-1} in November 2011. Most of the phytoplankton biomass occurred in the small size fraction (<5 µm, Fig. 3). The temporal pattern in total Chl *a* concentration closely followed that of the small size fraction of phytoplankton biomass. Chl *a* concentrations in the large size fraction (>5 µm) were generally < 0.1 µg L^{-1}, contributing little to the peaks in Chl *a*.

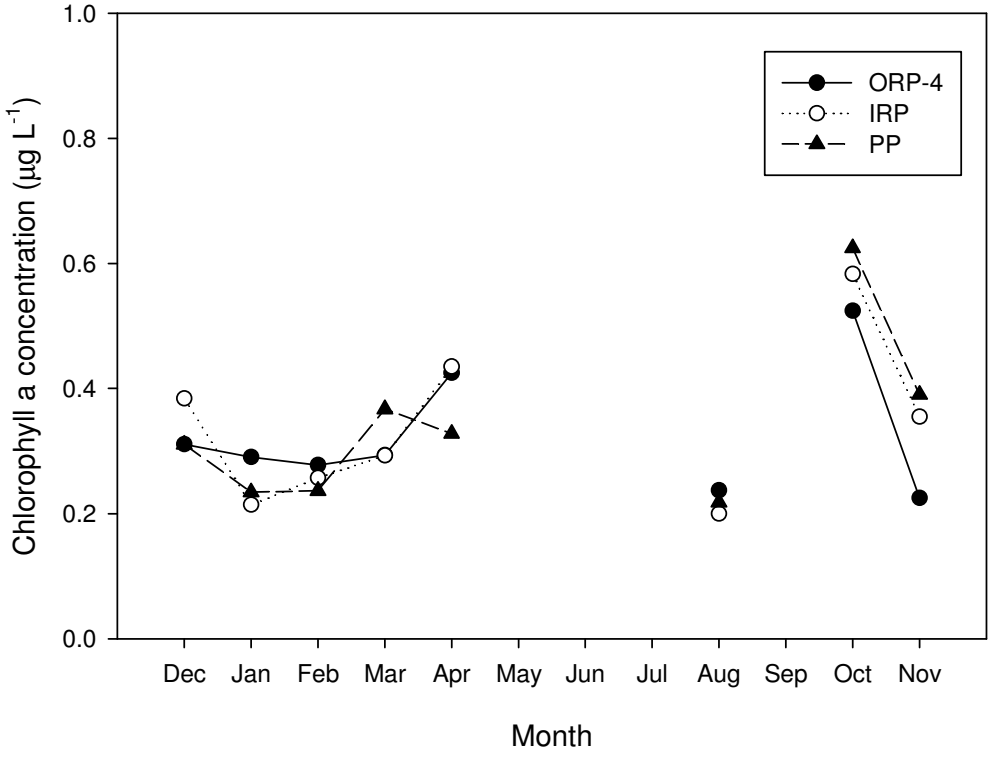

Figure 2. Monthly variation in chlorophyll a concentration during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011. See Fig. 1 for station locations.

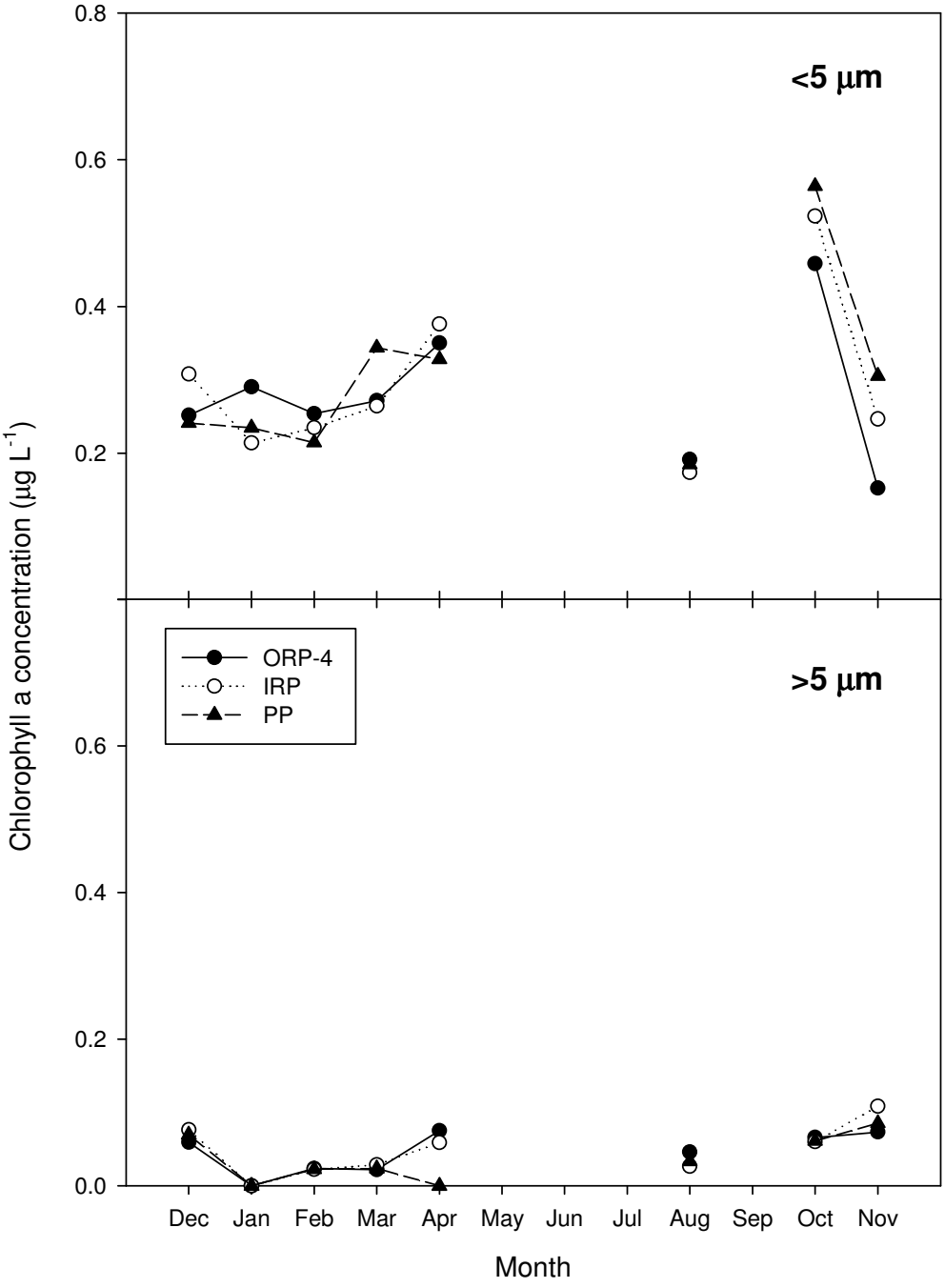

Figure 3. Monthly variation in size fractionated chlorophyll *a* during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011. See Fig. 1 for station locations.

An examination of marker pigments normalised to Chl *a* suggests a strong seasonality in the types of phytoplankton found in the study area (Fig. 4). There was a spike in zeaxanthin (an indicator of the presence of cyanobacteria) through summer

to a peak in January/February, with a decline in its presence through Autumn to lows in winter and spring. Chlorophyll *b* peaked in April and October 2011, which suggests an increase in the presence of prasinophytes in the community during these months (Jeffrey and Wright, 2006). The relative presence of 19-hex-fucoxanthin (an indicator of the presence of haptophytes) in total pigment in the region decreased from a high in December 2010 through to August 2011 before a increasing through October to a peak in November 2011.

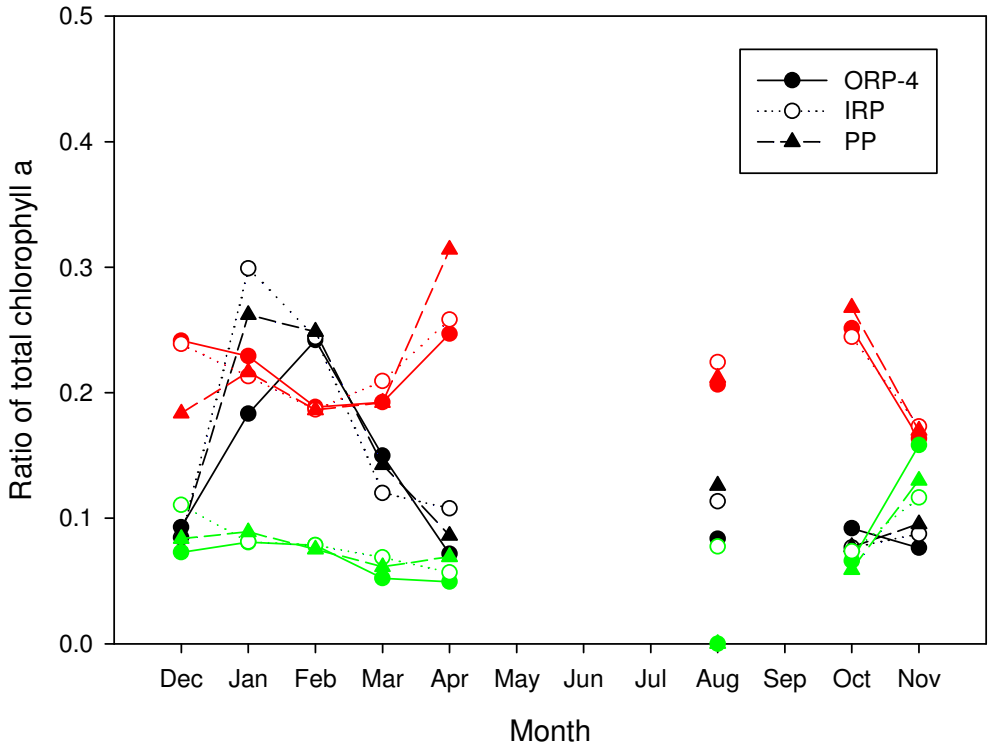

Figure 4. Monthly variation in the concentration of selected marker pigments normalised to chlorophyll a during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011. Black data points indicate Zeaxanthin, red data points indicate chlorophyll b, green data points indicate 19-hex-fucoxanthin. See Fig. 1 for station locations.

There was little variation in total phytoplankton abundances between stations, following the general pattern observed in pigment concentrations although with some notable exceptions (Fig. 5). Highest abundances were observed in December 2010 (up to ~1,000,000 cells L^{-1} at station ORP-4), with abundances decreasing in January 2011 to <100,000 cells L^{-1} at all stations by February. There was a small spike in abundance at all station to ~200,000 cells L^{-1} in March before a decline to <100,000 cells L^{-1} in April that persisted through winter. Abundances increased to ~200,000cells L^{-1} again in the spring (October/November). There was a distinct pattern in the type of phytoplankton driving peaks in total abundance (Fig. 5). The December peak was driven by high abundance of both diatoms and flagellates. The spike in March abundance was due to an increase in flagellate numbers while diatoms drove the October/November increase in abundance. Dinoflagellates were present in abundances <50,000 cells L^{-1} throughout the study. December peaks in diatom abundance were due to an increase in the presence of *Chaetoceros* spp. (>12,000 cells

L^{-1}), *Dactyliosolen fragilissimus* (up to 30,000 cells L^{-1}), *Leptocylindrus danicus* (up to 102,000 cells L^{-1}), and *L. minimus* (up to 50,000 cells L^{-1}), while the flagellate peak was due to increased abundances of the haptophytes *Emiliania huxleyi* (up to 12,000 cells L^{-1}) and *Phaeocystis* sp. (up to 300,000 cells L^{-1}), and the cryptophytes *Hemiselmis* sp. (up to 18,000 cells L^{-1}) and *Plagioselmis prolonga* (up to 14,000 cells L^{-1}). The March spike in flagellate abundance was driven by increases in the haptophytes *Chrysochromulina* sp. (>25,000 cells L^{-1}) *Emiliania huxleyi* (>12,000 cells L^{-1}) and *Phaeocystis pouchetti* (20,000 cells L^{-1}), and the cryptophytes *Hemiselmis* sp. (>25,000 cells L^{-1}) and *Plagioselmis prolonga* (>50,000 cells L^{-1}). Diatom abundances increased slightly in November due to a greater presence of *Chaetoceros* spp. (up to 86,000 cells L^{-1}).

Phytoplankton species richness (S) generally ranged between 28 and 36 species (Fig. 6). While there was no clear pattern evident between stations there was a general pattern of decreasing S from a December high through January, then increasing to another high in April. S at station IRP decreased to a low of 26 species in March 2011 before increasing to a peak of 43 species in April 2011. All stations had a decrease in S from August to October with an increase in November. A peak S of 36 occurred at station PP in April 2011, with the peak of 40 species at ORP-4 occurring in December 2010. A full list of phytoplankton species identified during the study is provided in Appendix 1. Temporally persistent diatoms included *Chaetoceros* spp., *Cocconeis* spp., naviculoid spp., and *Thalassiosira cf. mala*, which were all present in all months of the study, but rarely in abundances >6,000 cells L^{-1} except for *Chaetoceros* spp. *Cylindrotheca closterium* was present in summer/autumn (December to April) in numbers <6,000 cells L^{-1}, but absent in winter/spring. In contrast, *Asterionellopsis glacialis* and *Bacteriastrum elegans* were not found in summer/autumn but were present through winter/spring in abundances <6,000 cells L^{-1}. *Leptocylindrus danicus* and *L. minimus* were present in high numbers in December 2010, as mentioned above, but were not present in significant numbers again until winter/spring (August to Novemebr) when they were observed at abundances between 1,000 and 8,000 cells L^{-1}. The rest of the diatoms listed in Appendix 1 occurred infrequently, and at abundances < 6,000 cells L^{-1}. The dinoflagellates gymnodinioid spp., *Gyrodinium* spp., *Heterocapsa rotundata*, were present in all months of the study, and dominated the dinoflagellate community. They were generally found in numbers <5,000 cells L^{-1} but were present at >20,000 cells L^{-1} on rare occasions between January and April. *Protoperidinium* spp. and *Scripsiella* spp. were also found throughout the study but always in numbers <2,000 cells L^{-1}. Other dinoflagellates listed in Appendix 1 were found on a few occasions, and only in abundances < 5,000 cells L^{-1}. Flagellates present year round included the potentially icthyotoxic haptophyte *Chrysochromulina* spp., which reached abundances of >10,000 cells L^{-1} in January, March, April, October and November, with highs of ~30,000 cells L^{-1} in March 2011. The haptophyte *Emeliana huxleyi* was also found year round, and in abundances >10,000 cells L^{-1} in December, January and March. The cryptophytes *Hemiselmis* spp., *Leucocryptos marina*, and *Plagioselmis prolonga* were present in relatively high numbers in all months of the study except January and February. The prasinophytes *Pyramimonas* spp. and *Tetraselmis* spp. were also present in relatively high abundances through the study period.

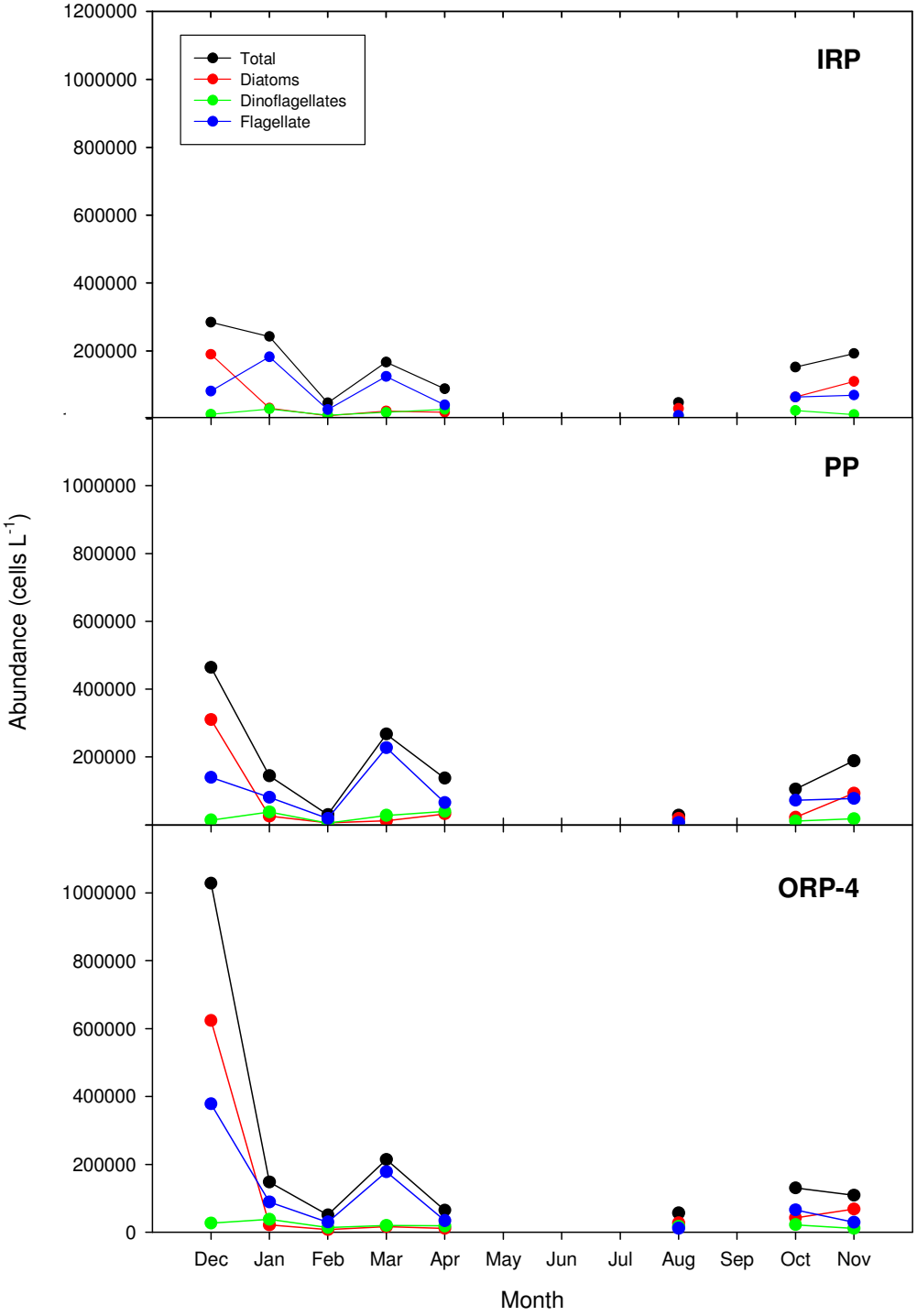

Figure 5. Monthly variation in phytoplankton abundance during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011. See Fig. 1 for station locations.

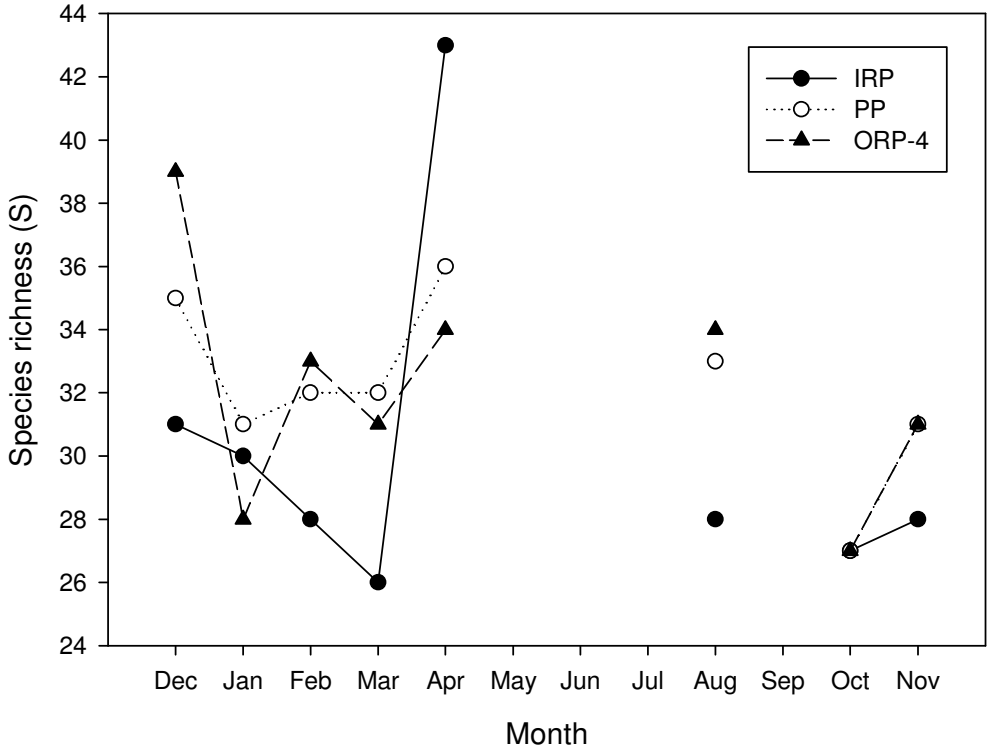

Figure 6. Monthly variation in phytoplankton species richness (S) during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011. See Fig. 1 for station locations.

4.2 Zooplankton biomass, abundance and community composition

There were no clear patterns in zooplankton biomass between stations or times of day (Fig. 7). There was a general increase in biomass and abundance from December 2010 to a peak in March 2011. Both biomass and abundance decreased through April and August into October before an increase in biomass in November that was not associated with an increase in abundance. Abundances in night samples were generally higher than abundances in day samples, with no clear patterns in biomass between night and day samples.

There were no clear patterns in zooplankton species richness between stations or times of day. Species richness (S) decreased from December 2010 to January 2011 before increasing to a peak ~10 species in April (Fig. 8). There was a decrease in S through August into October before an increase to near peak levels in November 2011. Copepoda and Copepoda nauplii (see Appendix 2 for common names of all zooplankton taxa) dominated the zooplankton community during the study (Fig. 9). Appendicularia and Bivalvia were also prominent. In figure 9, zooplankton taxa which contributed <3% to the total community were grouped together as "other". These taxa included barnacle nauplii, Cyphonautes larvae of Bryozoa, Chaetognatha, Cladocera, Cnidaria, Ctenophora, Decopoda, Gastropoda, Ostracoda, Polychaetae, and pluteus larvae. Copepoda, Copepoda nauplii, and Appendicularia were present in

all months of the study. Bivalvia were prominent in the region April, October and November 2011.

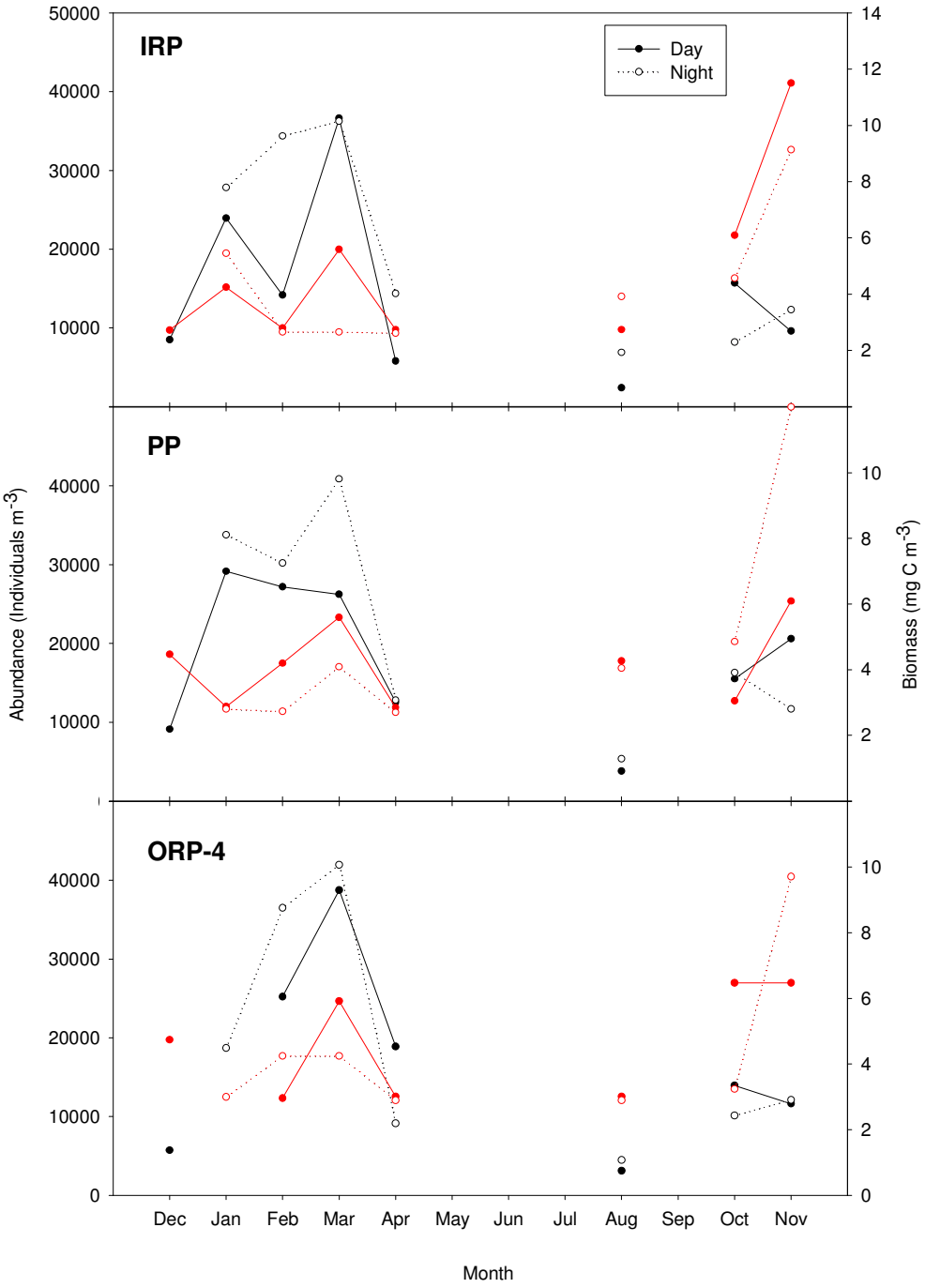

Figure 7. Temporal variation in zooplankton abundance and biomass during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011. Black data points represent abundance (left y-axis), red data points represent biomass (right y-axis). See Fig. 1 for station locations.

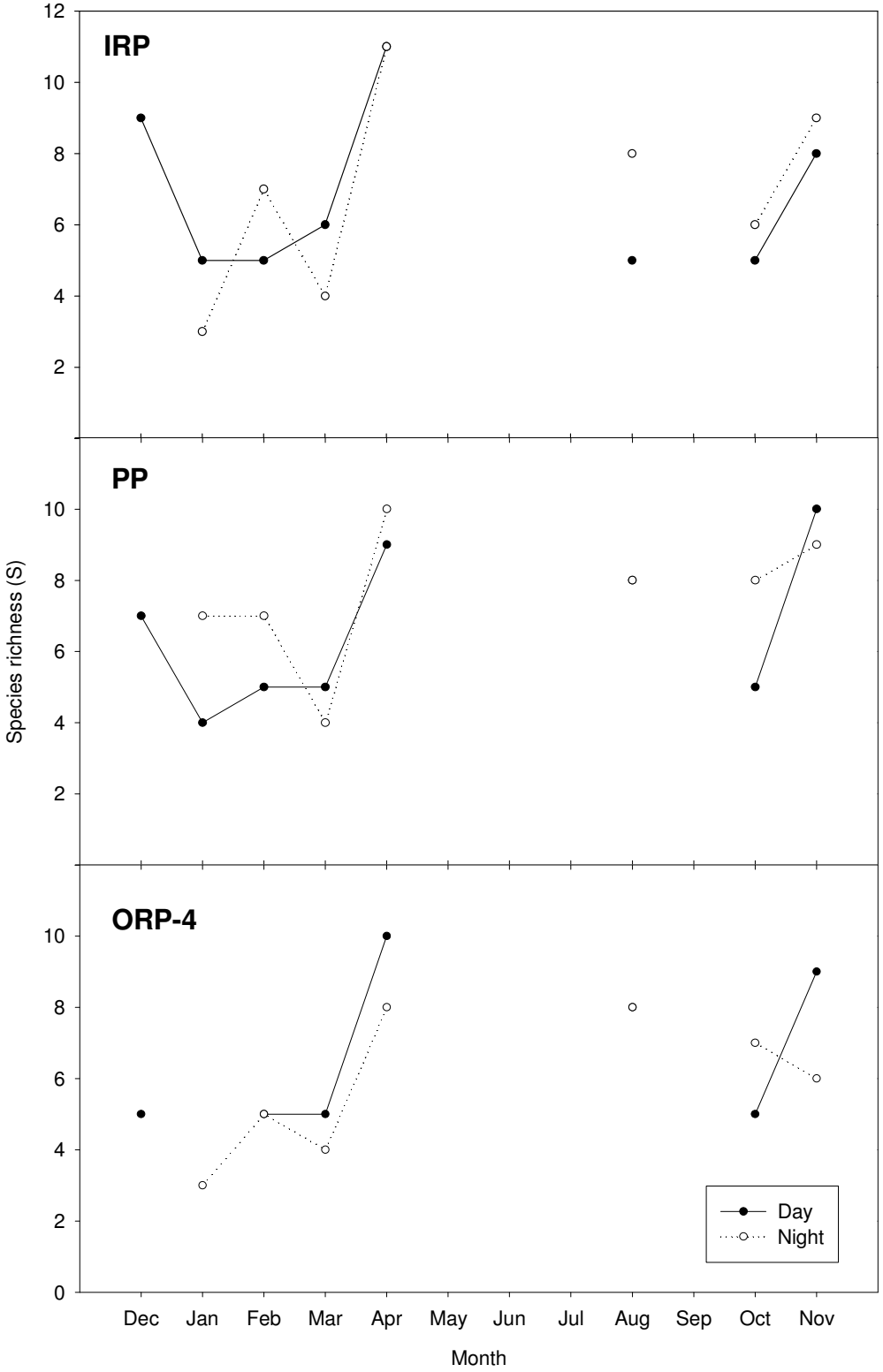

Figure 8. Temporal variation in zooplankton species richness (S) during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011. See Fig. 1 for station locations.

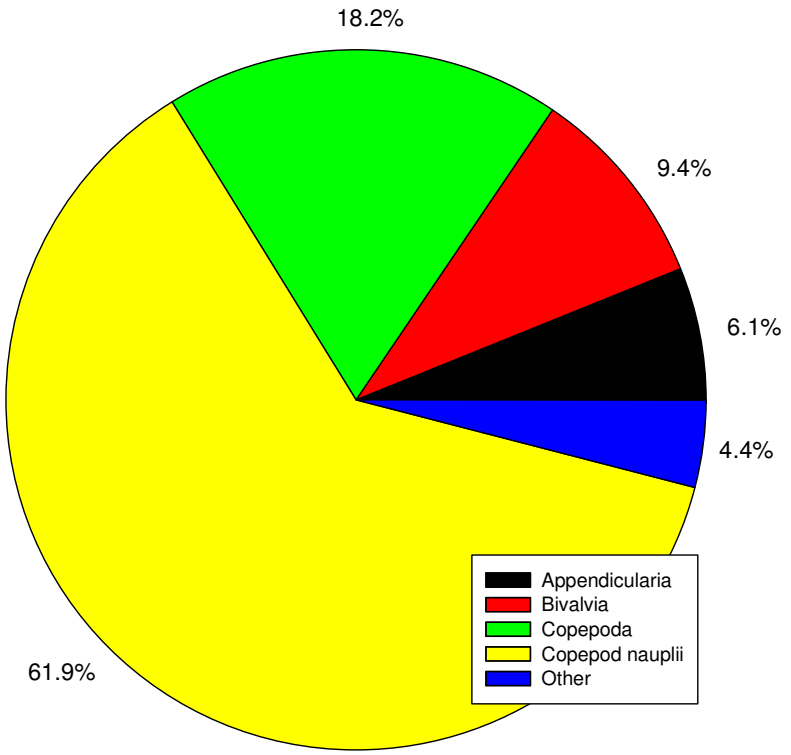

Figure 9. Proportional composition, based on abundance, of the zooplankton community during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011.

4.3 Ichthyoplankton abundance and community composition

The pattern of egg abundance sharply decreased from 18 eggs m^{-3} at station IRP, 10 eggs m^{-3} at station PP, and 8 eggs m^{-3} at station ORP-4 in December 2010 through to zero eggs at all stations in March 2011 (Fig. 10). There was a slight increase in April at all stations and a peak at 15-20 eggs m^{-3} in November.

A large proportion of eggs in the ichthyoplankton community were unidentified (Fig. 11). Unidentified eggs were found year round, with highest abundances in December 2010, and October and November 2011. Anchovy eggs were present in all months except August-November 2011, with highest abundances at IRP in December 2010 (6.1 eggs m^{-3}). Sardine eggs were present throughout in all months of the study except February, March, and October, and were in highest numbers at IRP in December 2010 (4.7 eggs m^{-3}).

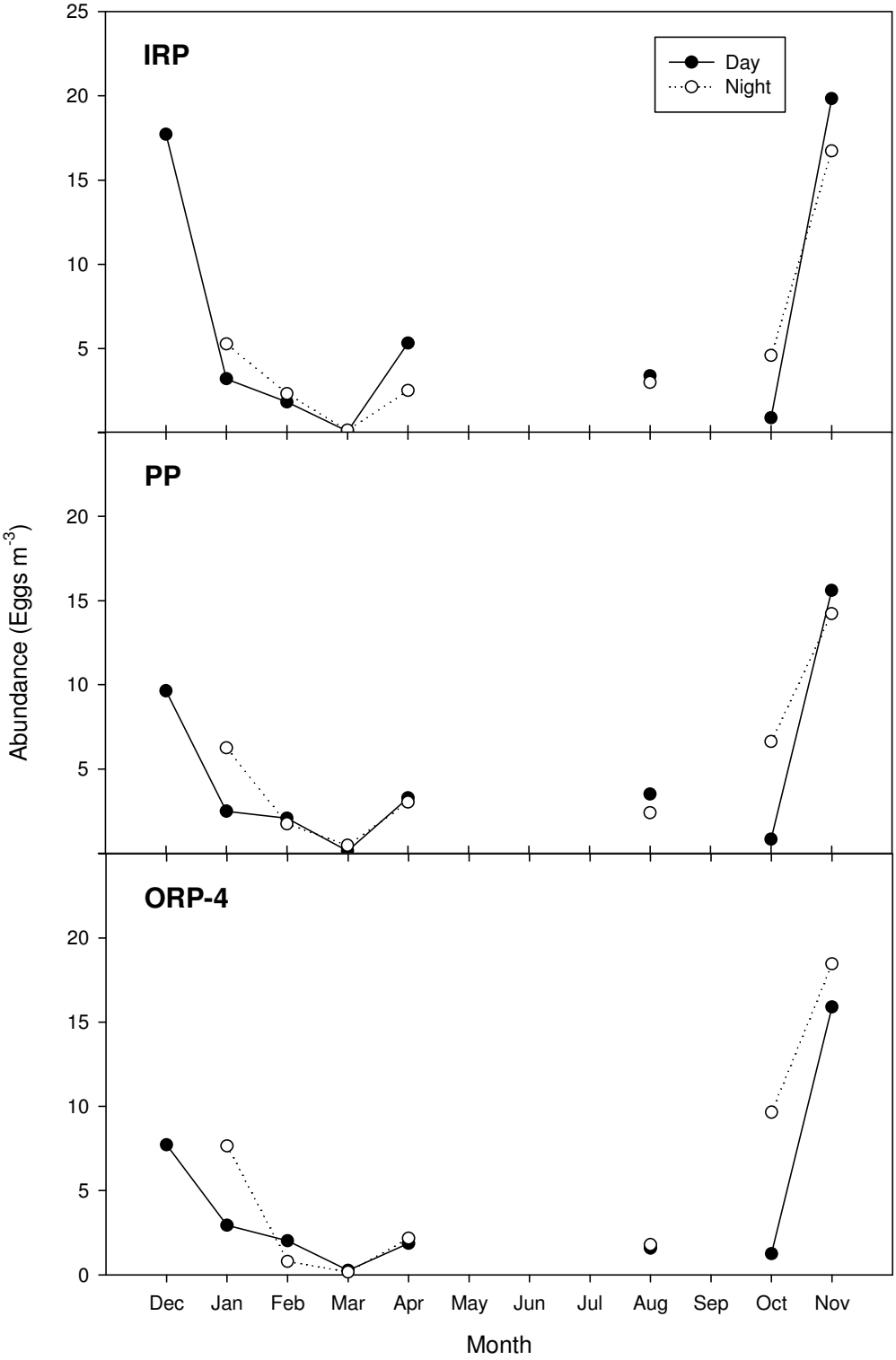

Figure 10. Temporal variation in egg numbers during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011. See Fig. 1 for station locations.

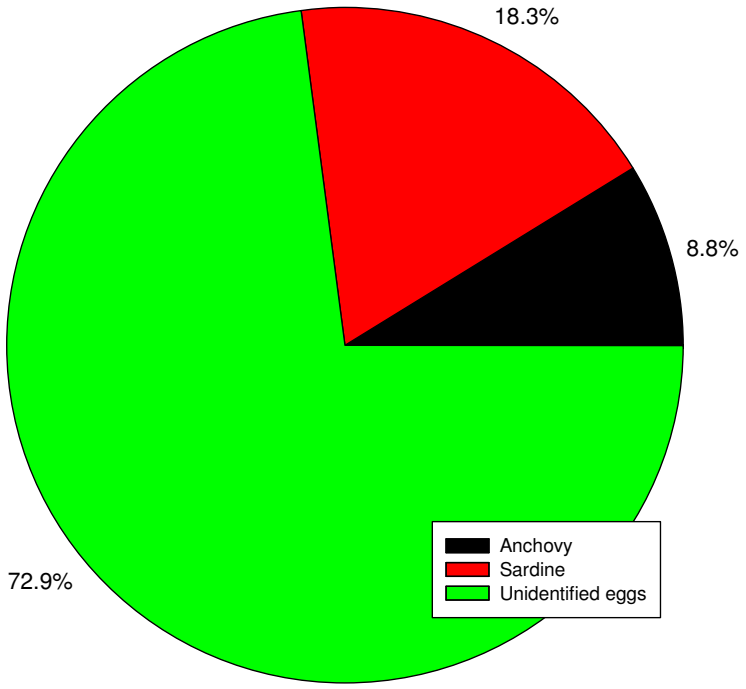

Figure 11. Proportional composition of egg types in the community during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011.

Midnight abundances were higher than midday abundances (Fig. 12). The general temporal pattern saw abundances <2 larvae m^{-3} throughout the year, with a sharp increase to peaks of 7-8 larvae m^{-3} in November. Unidentified larvae dominated the community throughout the study, with anchovy, blue sprat and sardine larvae also present (Fig. 13). Unidentified larvae were present year round, with highest abundances in midnight samples in November 2011 (>6.5 larvae m^{-3}). Anchovy larvae were found at night in December and January in low abundance (0.1-0.4 larvae m^{-3}). Sardine larvae were present in all months except February, March, October and November, in abundances <0.25 larvae m^{-3}. Blue sprat larvae were found in January to March, and November 2011, at abundances <0.1 larvae m^{-3}. Sygnathids were present in January, March, April, August, October and November, generally at station ORP-4 at very low abundances (0.01-0.04 larvae m^{-3}).

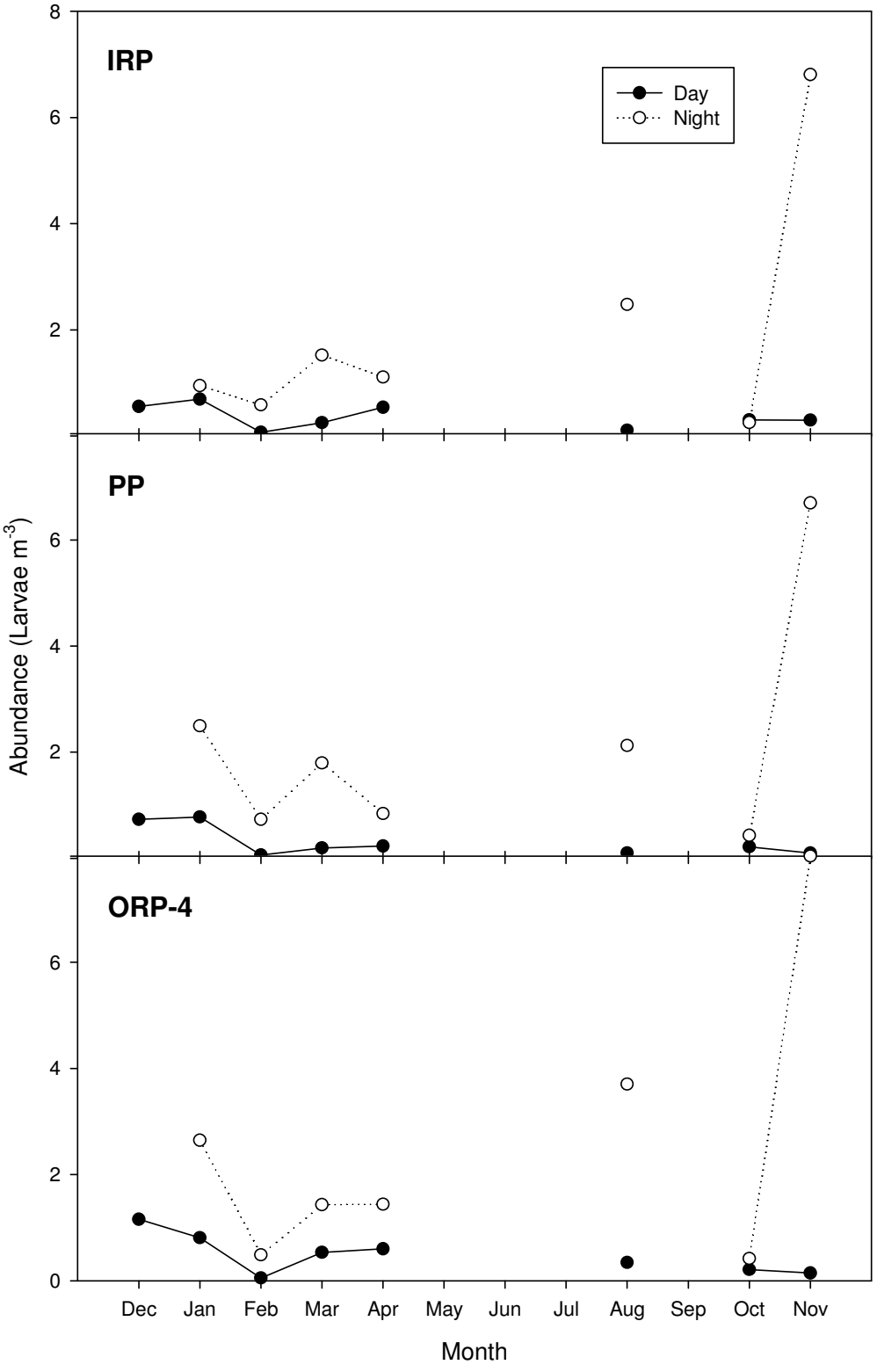

Figure 12. Temporal variation in larval abundance during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011. See Fig. 1 for station locations.

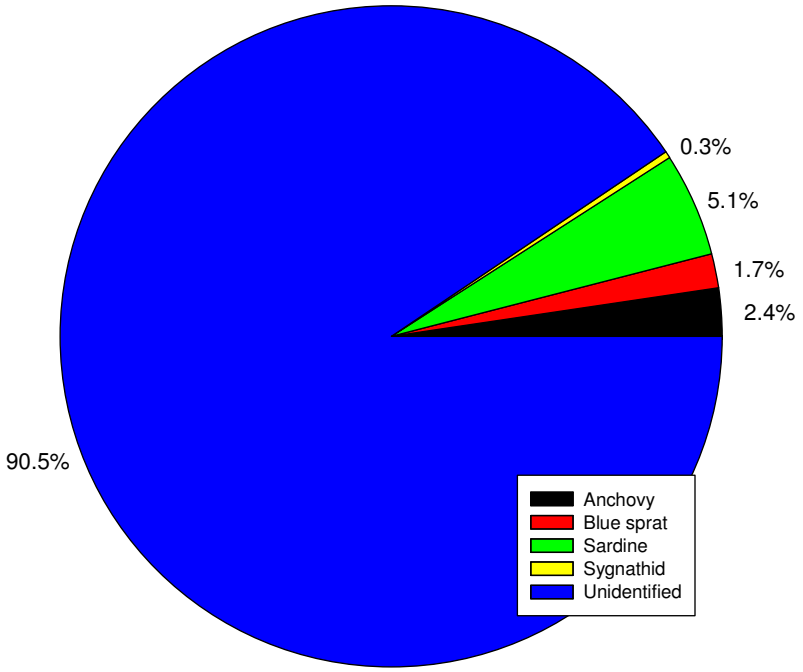

Figure 13. Proportional composition of icthyoplankton larval community during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011.

4.4 Primary productivity

Modelled rates of primary productivity agreed well with measured results (Fig. 14), but there was much temporal variation in photosynthetic parameters (Table 1). P^b_{max} ranged between 2.27 and 3.33 mg C (mg chl)$^{-1}$ hr^{-1}, with highest values in January and lowest values in August. Photosynthetic efficiency ranged between 0.011 and 0.052 mg C (mg chl)$^{-1}$ hr^{-1} (µmol m^{-2} s^{-1})$^{-1}$, with highest α in August and lowest in January. Daily integral productivity ranged from 89.3 to 303.6 mg C m^{-2} d^{-1}, with the highest productivity in October > three times the value if the lowest productivity which occurred in April (Table 1, Fig. 15). Phytoplankton turnover times decreased gradually from January to a peak in October. Highest gross phytoplankton growth rates occurred in August and October (Fig. 15).

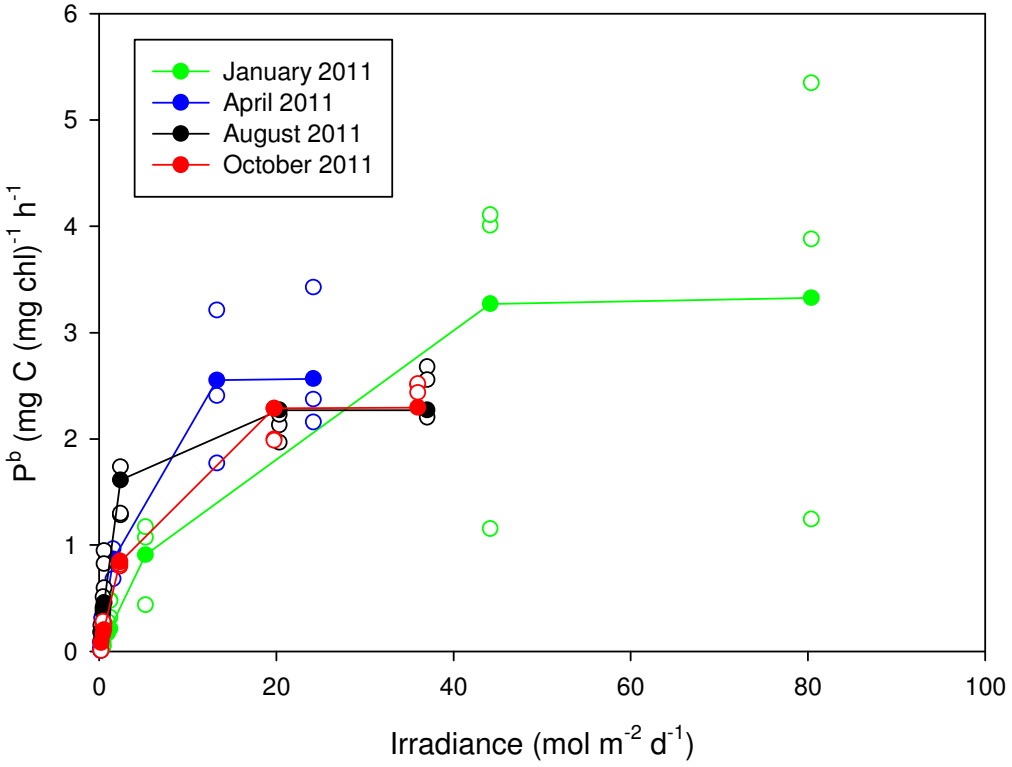

Figure 14. Seasonal variation in photosynthesis – irradiance curves from ^{14}C experiments conducted during the Adelaide Desalination Project plankton characterisation study on water collected from station PP. Open circles indicate measured rates of primary productivity, closed circles indicate rates of primary productivity modelled according to Jasby and Platt (1976). See Fig. 1 for station location.

Table 1. Seasonal variation in photosynthetic parameters and daily integral productivity during the Adelaide Desalination Project plankton characterisation study. D_{irr} is daylength is in decimal hours, I'_o is the irradiance just below the sea surface (µmol m^{-2} s^{-1}), K_d is the attenuation coefficient of downwelled irradiance (m^{-1}), Chl a is surface extracted chlorophyll a concentration (µg L^{-1}), I_k is the irradiance corresponding to the onset of light saturation of photosynthesis (µmol m^{-2} s^{-1}), α is the photosynthetic efficiency (mg C (mg chl)$^{-1}$ hr^{-1} (µmol m^{-2} s^{-1})$^{-1}$), P^b_{max} is the biomass specific maximum photosynthetic rate (mg C (mg chl)$^{-1}$ hr^{-1}), and DIP is the daily integral productivity (mg C m^{-2} d^{-1}).

Month	D_{irr}	I'_o	K_d	R^2	Chl a	I_k	Alpha	P^b_{max}	DIP
Jan-11	14.18	1943	0.19	0.95	0.24	298.8	0.01	3.33	137.7
Apr-11	11.33	315	0.21	0.95	0.33	72.0	0.04	2.57	89.3
Aug-11	11.32	935	0.16	0.94	0.22	43.5	0.05	2.27	119.6
Oct-11	12.57	314	0.10	0.99	0.63	97.8	0.02	2.29	303.6

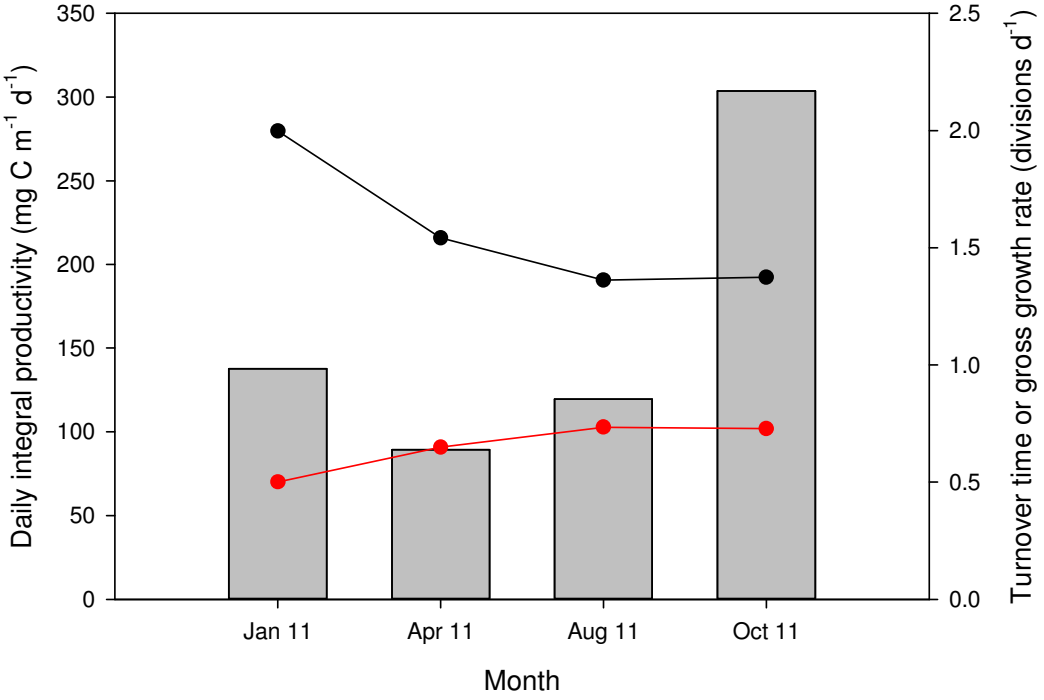

Figure 15. Seasonal variation in daily integral productivity during the Adelaide Desalination Project plankton characterisation study. Black line represents turnover time, red line represents gross phytoplankton growth rate.

4.5 Secondary productivity

G'_{max} increased with increasing SST during this study to peak in January 2011 before declining with SST in autumn (Table 2). There were no clear patterns in zooplankton grazing rate between stations or times of day (Table 2, Fig. 16). Grazing rate remained at ~2 to 4 mg C m^{-3} d^{-1} from December 2010 through to April 2011, decreasing to ~1 mg C m^{-3} d^{-1} in August, before rising to a peak at ~5 to 7 mg C m^{-3} d^{-1}in November.

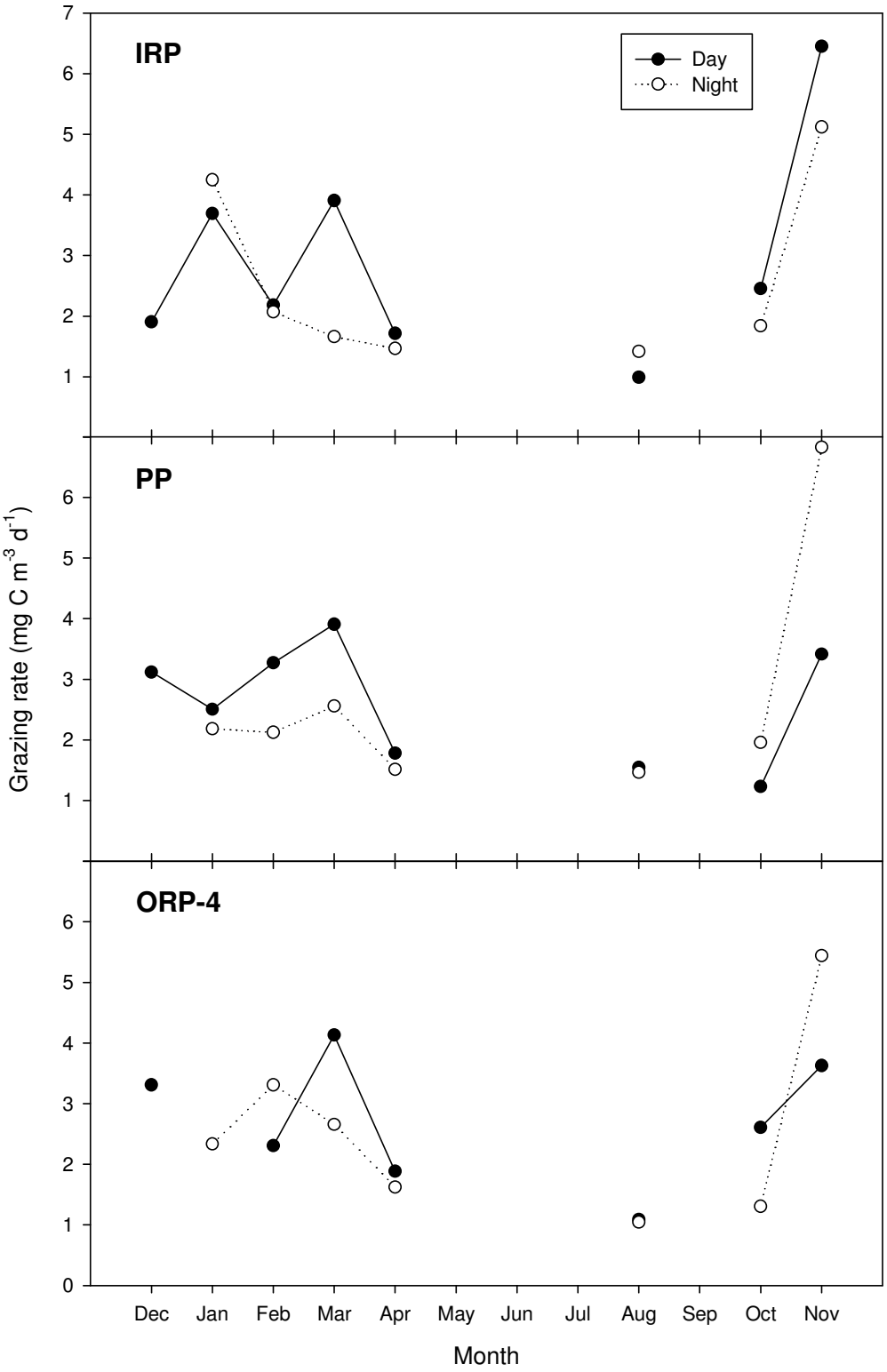

Figure 16. Temporal variation in zooplankton grazing rate during the Adelaide Desalination Project plankton characterisation study between December 2010 and November 2011. See Fig. 1 for station locations.

Table 2. Temporal variation in zooplankton settling volume (ml m^{-3}), biomass (mg m^{-3}), and grazing rate (mg C m^{-3} d^{-1}) during the ADP PCS. SST = CTD measured sea surface temperature (ºC), G'_{max} = potential growth rate (d^{-1}). See Fig. 1 for station locations.

Station	Month	Time	Settling vol	Biomass	SST	G'_{max}	Grazing rate
IRP	DEC	Day	0.5	2.7	20.0	0.49	1.90
PP	DEC	Day	0.75	4.5	20.0	0.49	3.12
ORP4	DEC	Day	0.75	4.7	20.0	0.49	3.31
IRP	JAN	Day	0.75	4.2	22.0	0.61	3.70
IRP	JAN	Night	1	5.4	21.0	0.55	4.25
PP	JAN	Day	0.5	2.9	22.0	0.61	2.50
PP	JAN	Night	0.5	2.8	21.0	0.55	2.18
ORP4	JAN	Night	0.5	3.0	21.0	0.55	2.33
IRP	FEB	Day	0.5	2.8	21.0	0.55	2.18
IRP	FEB	Night	0.5	2.7	21.0	0.55	2.07
PP	FEB	Day	0.75	4.2	21.0	0.55	3.27
PP	FEB	Night	0.5	2.7	21.0	0.55	2.13
ORP4	FEB	Day	0.5	3.0	21.0	0.55	2.31
ORP4	FEB	Night	0.75	4.2	21.0	0.55	3.31
IRP	MAR	Day	1	5.6	20.0	0.49	3.91
IRP	MAR	Night	0.5	2.7	19.0	0.44	1.66
PP	MAR	Day	1	5.6	20.0	0.49	3.91
PP	MAR	Night	0.75	4.1	19.0	0.44	2.56
ORP4	MAR	Day	1	5.9	20.0	0.49	4.13
ORP4	MAR	Night	0.75	4.2	19.0	0.44	2.66
IRP	APR	Day	0.5	2.7	19.0	0.44	1.71
IRP	APR	Night	0.5	2.6	18.0	0.39	1.47
PP	APR	Day	0.5	2.8	19.0	0.44	1.78
PP	APR	Night	0.5	2.7	18.0	0.39	1.51
ORP4	APR	Day	0.5	3.0	19.0	0.44	1.88
ORP4	APR	Night	0.5	2.9	18.0	0.39	1.62
IRP	AUG	Day	0.5	2.7	14.0	0.25	0.99
IRP	AUG	Night	0.75	3.9	14.0	0.25	1.42
PP	AUG	Day	0.75	4.3	14.0	0.25	1.54
PP	AUG	Night	0.75	4.0	14.0	0.25	1.46
ORP4	AUG	Day	0.5	3.0	14.0	0.25	1.09
ORP4	AUG	Night	0.5	2.9	14.0	0.25	1.04
IRP	OCT	Day	1	6.1	15.0	0.28	2.46
IRP	OCT	Night	0.75	4.6	15.0	0.28	1.84
PP	OCT	Day	0.5	3.0	15.0	0.28	1.23
PP	OCT	Night	0.75	4.9	15.0	0.28	1.96
ORP4	OCT	Day	1	6.5	15.0	0.28	2.61
ORP4	OCT	Night	0.5	3.2	15.0	0.28	1.30
IRP	NOV	Day	2	11.5	18.0	0.39	6.45
IRP	NOV	Night	1.5	9.1	18.0	0.39	5.12
PP	NOV	Day	1	6.1	18.0	0.39	3.41
PP	NOV	Night	2	12.2	18.0	0.39	6.83
ORP4	NOV	Day	1	6.5	18.0	0.39	3.63
ORP4	NOV	Night	1.5	9.7	18.0	0.39	5.4

5. Discussion

The April/May/June peak in phytoplankton biomass that was identified in phase one of this study (van Ruth 2010) was also observed in this second phase of the study. Although sampling was not conducted during June due to adverse weather conditions, there was a trend of increasing Chl *a* concentrations through February/March/April in the region, suggesting a peak in June followed by lower concentrations in spring/summer. In contrast to the first phase of the study, however, when Chl *a* concentrations remained ~0.2 μg L^{-1} through spring and summer before increasing in April to peak at ~0.8 μg L^{-1} in June (van Ruth 2010), concentrations in this study spiked from ~0.2 μg L^{-1} in August to ~0.6 μg L^{-1} in October before falling again in November. Unlike the first phase of the study, patterns in phytoplankton abundance did not follow patterns in Chl *a* concentration. In 2009-10, phytoplankton abundances peaked at ~500,000 to 600,000 cells L^{-1} in April and June, with abundances ~100,000 to 200,000 cells L^{-1} in December. The April peak was driven by increased numbers of diatoms and the June peak was driven by flagellates (van Ruth 2010). In 2010-11, abundances peaked in December at up to 1,000,000 cells L^{-1} largely due to increases in diatom and flagellate numbers. There was also a downward trend in abundances from the flagellate driven March spike into April when abundances were <100,000 cells L^{-1}.

Temporal patterns in phytoplankton community composition were also consistent with patterns observed in stage 1 of this study. The April and October 2011 peaks in Chl *b*, corresponding with increases in the abundance of haptophytes in the community, were higher in 2010-11 than in 2009-10 (~30% of total Chl *a* in this study compared to ~20% of total Chl *a* in 2009-10 (van Ruth 2010)). In contrast, the summer increase in cyanobacteria (as indicated in increases in the fraction of zeaxanthin in the community to ~30% of total Chl *a* in January/February 2011) was slightly lower than that observed in phase 1 (zeaxanthin peaking at ~50% of total Chl *a* in February 2010 (van Ruth 2010)). Cyanobacteria are generally more tolerant of environmental extremes than phytoplankton, and are often the only phototrophic organisms found in extreme environments, such as hypersaline waters (Madigan et al., 1997). They produce potent neurotoxins, and blooms can rapidly kill marine organisms (Madigan et al., 1997). Ongoing monitoring of cyanobacterial populations off Port Stanvac by HPLC pigment analysis is recommended because increased salinity in the vicinity of the ADP outflow structures has the potential to promote cyanobacteria blooms.

Several potentially harmful/toxic harmful algal bloom (HAB) phytoplankton species were detected in the community off Port Stanvac, most of which were also detected in 2009-10 (van Ruth 2010), but some of which were new. There were also differences in abundance of HAB species between 2009-10 and 2010-11. HAB species detected in both stage 1 and stage 2 included species harmful/toxic to fish, and species potentially toxic to humans. The diatom *Chaetoceros* can cause harm to fish even at low cell densities due to the siliceous spikes (setae) and barbs which characterise the genus and can break off and penetrate the gill membranes of fish (Hallegraeff, 2002). All *Chaetoceros* species possess setae, but it is only the species that have setae with barbs (*C. concavicornis* and *C. convolutus*) that are regarded as harmful to fish (Hallegraeff, 2002). While *Chaetoceros* spp. were present in all months of this study, potentially harmful species were not identified. Toxic diatoms found in the waters off Port Stanvac include *Pseudonitzschia delicatissima*, and *P. fraudulenta/australis*, which are likely producers of the toxins that cause amnesic shellfish poisoning (Hallegraeff, 2002; Baugh et al., 2006). *Rhizosolenia* spp. have

been linked to shellfish mortality, with inflammation and degeneration of digestive glands and a bitter taste affecting shellfish marketability (Hallegraeff, 2002). Toxic dinoflagellates were also found. *Alexandrium catenella* is a producer of toxins that cause paralytic shellfish poisoning in humans (Hallegraeff, 2002; Jester, 2008), while *Karenia papilionacea* contains toxins responsible for neurotoxic shellfish poisoning, and can kill fish (Hallegraeff, 2002). *Prorocentrum cordatum* and *P. rhathymum* have been associated with shellfish mortality (Hallegraeff, 2002; Pearce et al., 2005), and *Dinophysis acuminata* produces the toxin that causes diarrhetic shellfish poisoning in humans (Hallegraeff, 2002; Sutherland, 2008). Ichthyotoxic flagellates identified during this study include the haptophytes *Chrysochromulina* spp., a dominant flagellate throughout the study, and *Heterosigma* sp. With the exception of *Chaetoceros* spp. and *Chrysochromulina* spp., these harmful/toxic species of phytoplankton were found infrequently at low abundances (< 2,000 cells L^{-1}). In 2010-11, *Chaetoceros* spp. were present in abundances averaging 14,260 cells L^{-1} over the 21 times the species was detected, compared to an average of 5,273 cells L^{-1} over the 11 times it was detected in 2009-10. Average *Chrysochromulina* spp. abundances were ~30% lower in 2010-11 than abundances observed in 2009-10. HAB species detected in the Port Stanvac region in 2010-11 for the first time included the dinoflagellate *Karenia micimotoi*, a known cause of neurotoxic shellfish poisoning. While detected, however, this species was rarely present. *Phaeocystis* sp., a non toxic species which forms gelatinous blooms that can cause fish kills by clogging the gills of fish and bivalves (Hallegraeff, 2002) was present from December 2010 to March 2011 in abundances >5,000 cells L^{-1}, reaching bloom proportions of up to 119,000 cells L^{-1} in December.

The continued presence of cyanobacteria and HAB species, the continued detection of new HAB species and increases in HAB species abundances warrants ongoing monitoring. There is increasing evidence that HAB events are becoming more common at local, regional and global scales (Hallegraeff, 1993; Anderson et al., 2002; Gilbert et al., 2005a), with the increasing impact of anthropogenic activities on coastal ecosystems a major cause (Gilbert et al, 2005b; Cochlan et al, 2008; Kudela et al., 2008). These activities, which may include desalination plant construction and operation, cause changes in nutrient concentrations and irradiance, through increased terrestrial run-off, wastewater outflow or resuspension of sediments due to dredging during construction work, which may drive changes in phytoplankton community composition and may promote HABs. A cyanobacterial bloom/HAB event represents a significant public health risk, not only to users of the marine environment, but to consumers of desalinated water if noxious/toxic substances associated with the HAB are not completely removed in the desalination process (Caron et al., 2010). HABs may also lead to increases in turbidity, total suspended solids, and total organic content that can pose issues for the whole desalination process through the clogging of reverse osmosis membranes (Caron et al 2010). A detailed understanding of spatial and temporal variation in HAB ecology may aid in the efficiency of the desalination process through the early detection of HAB events such that pre-determined strategies (pre-treatment activities, chemical additions, maintenance activities) can be implemented to maintain optimal production capacity (Caron et al., 2010). Ongoing monitoring of cyanobacteria/HAB species in the waters off Port Stanvac may help prevent or minimise the impact of HABs on the desalination process and the quality of desalinated water. Early detection of HAB events would also ensure that mitigation strategies can be enacted to prevent significant detrimental impacts occurring to marine life and public health.

As observed in 2009-10 (van Ruth 2010), peaks in zooplankton biomass and abundance roughly corresponded to peaks in phytoplankton biomass and abundance. The exception was found in December 2010 when despite phytoplankton abundance and biomass were high, zooplankton abundance and biomass were low. The size structure of the zooplankton community may have caused this exception: the December peak in phytoplankton abundance was driven by high numbers of large diatoms, at a time when the Port Stanvac zooplankton community was dominated by small organisms such as copepod nauplii. Diatoms are too large to be consumed by small zooplankton, hence the low zooplankton abundances in December 2010. In contrast to 2009-10, however, the December diatom peak did not result in an increase in numbers of larger zooplankton in the Port Stanvac community. The March spike in zooplankton abundance and biomass was due to a large increase in numbers of copepod nauplii (to up to five times the abundances in December), which were responding to the spike in numbers of small flagellates in the phytoplankton community, an ample food source for smaller zooplankton. In general, zooplankton abundances in 2010-11 were ~one third lower than those observed in 2009-10 (van Ruth 2010), with biomass ~ one sixth lower in stage 2 than stage 1. No lobster, abalone, prawn or blue crab larvae were found in the study area. Although Decapoda and Gastropoda larvae were found in zooplankton samples, they were a small proportion of the total zooplankton, contributing <0.2% and <2% to the total community, respectively. When compared to results from 2009-10 (van Ruth 2010), zooplankton species richness was lower, peaking at ~10 species compared with peaks of 14-17 species. There were nine species contributing >3% to the total zooplankton community in 2009-10 of this study but only 4 species doing so in 2010-11.

Peak ichthyoplankton egg and larval abundances were higher in 2010-11. While temporal patterns in egg abundance were generally similar, abundances were at least double, sometimes >triple those observed in 2009-10 (van Ruth 2010; November/December abundances ~10-20 eggs m^{-3} compared to ~2-6 eggs m^{-3}). Midnight samples collected in April, however, had abundances of ~6-12 eggs m^{-3} in 2009-10 (van Ruth 2010), compared to <2 eggs m^{-3} in 2010-11. In contrast to results from 2009-10, in which larval abundances were always observed at <1.0 larvae m^{-3}, midday abundances were generally <1.0 larvae m^{-3} while midnight samples were generally >1.0 larvae m^{-3}. November/December peaks were observed in both 2009-10 and 2010-11, with the April peak present in the 2009-10 absent in 2010-11. The November peak in larval abundance reported in results in this document was ~13 times the November/December peak reported in van Ruth (2010).

Unidentified eggs and larvae made up a large proportion of the icthyoplankton community during this study. There is a paucity of information on larval fish taxonomy in South Australia, and many fish larvae have not been described. If funding was available, it would be prudent to investigate icthyoplankton further, using DNA analysis techniques to confirm identifications and provide data on the identity of unidentified samples. This would result in more accurate quantification of the potential impacts of the desalination process on early life history stages of ecologically and commercially important fish species, providing information that may help to alleviate community concerns about this highly sensitive issue.

Ichthyoplankton communities varied throughout the study. There were more sardine eggs observed in 2010-11 than in 2009-10 (an average of 0.95 eggs per sample compared to 0.79 eggs per sample), with more anchovy eggs also found (0.46 eggs per sample compared to 0.15 eggs per sample). There were more sardine larvae in 2010-11 than in 2009-10 (an average of 0.06 larvae per sample compared to 0.02

larvae per sample), but less anchovy larvae (0.03 larvae per sample compared to 0.12 larvae per sample). Blue sprat have been recorded throughout GSV in abundances of up to 0.2 larvae m^{-3} (Rogers et al., 2003), and were recorded in 2010-11 in abundances of 0.4 to 2.9 larvae m^{-3}. These larvae were not observed in the community 2009-10 (van Ruth 2010). Leather jacket larvae were not observed in this part of the study but comprised 11.7% of the total larval community in 2009-10. Sygnathids were not present 2009-10 but were observed at low abundances in 2010-11. No other commercially important marine scale-fish larvae or larvae of protected fish taxa were identified during this project although they may be present in the large proportion of eggs and larvae that could not be identified . It appears that ichthyoplankton dynamics may also be changing around the Port Stanvac desalination plant. These changes may have adverse affects on commercially and recreationally important species in the region, and ecologically important rare and protected species (eg. such sygnathids as the leafy sea dragon *Phycodurus eques*). Given community sensitivity to this issue, and the paucity of information regarding egg and larval abundance in GSV, further investigation is required to ensure that temporal cycles in egg and larval abundance are properly understood, and impacts from the ADP are quantified and mitigated.

Seasonal primary productivity varied between 2009-10 (van Ruth 2010) and 2010-11. Peak productivity occurred in January in 2009-10 and October in 2010-11. Gross phytoplankton growth rate peaked at ~0.75 divisions d^{-1}, also in October 2011, 50% lower than the peak in 2009-10 of 1.5 divisions d^{-1} that occurred in April 2010 (van Ruth 2010). G'max was comparable to values recorded for SWSG in van Ruth et al. (2009), increasing through spring into summer with increasing SST, but peak grazing rates were an order of magnitude lower due to the lower zooplankton biomass measured in this study. G'max and grazing rates were similar to those reported for stage 1 of this project (van Ruth 2010).

Rates of daily integral primary productivity measured in the waters off Port Stanvac were very low by global standards, comparable to the oligotrophic waters of the eastern Great Australian Bight (<400 mg C m^{-2} d^{-1}, van Ruth, 2009), the Leeuwin current off south west Western Australia (110-530 mg C m^{-2} d^{-1}, Hanson et al., 2005), and the north and south Atlantic sub-tropical gyres (18-362 mg C m^{-2} d^{-1}, Maranon et al., 2003). Rates were comparable to the lowest rate of primary productivity measured in South Western Spencer Gulf (SWSG ~200 mg C m^{-2} d^{-1} in October 2007) by van Ruth et al. (2009), and were similar to rates reported for the waters off Port Stanvac in van Ruth (2010) (95.9 to 227.1 mg C m^{-2} d^{-1}). Rates of primary productivity alone are insufficient to explain temporal variation in phytoplankton biomass and abundance, and must be examined with information about phytoplankton growth rates, and zooplankton grazing. Highest primary productivity occurred in October 2011, with the highest phytoplankton growth rate also observed at this time. Grazing rates were low in October, which allowed phytoplankton biomass to accumulate, driving the October peak in phytoplankton Chl *a* concentrations (Fig. 2). This biomass triggered an increase in zooplankton grazing pressure in November, and fish spawning and increased ichthyoplankton larval activity, that caused a sharp decline in concentrations of Chl *a*. Lowest productivity and low growth rates were measured in April, but low grazing pressure allowed the slow accumulation of phytoplankton biomass creating the upward trend in Chl *a* concentrations (Fig. 2).

6. Summary and conclusions

Temporal patterns in phytoplankton, zooplankton and icthyoplankton dynamics identified in 2010-11 differ from those identified in 2009-10. There were differences in temporal patterns of phytoplankton abundance and community composition between the two phases of this study, including the presence and abundance of potentially harmful/toxic algae species, and differences in zooplankton and icthyoplankton abundances and community composition. The cycles of primary productivity and phytoplankton growth, and zooplankton grazing pressure also differed between 2009-10 and 2010-11, although rates of primary productivity remained similar, and relatively low on a global scale.

The characteristic summer increase in cyanobacteria, together with the continued presence of HAB species, the continued detection of new HAB species and increases in HAB species abundances warrants ongoing monitoring, not just for reasons of public health, but to ensure optimal operating efficiency of the ADP. Plankton biomass and abundance data suggest that the patterns of monthly/seasonal phytoplankton succession in the Port Stanvac region are changing, with associated changes in zooplankton and icthyoplankton community dynamics which have implications for commercially and recreationally important fish species, as well as socially important protected fish species. ADP operations may have driven changes in nutrient concentrations and irradiance through increased terrestrial run-off, wastewater outflow or resuspension of sediments due to dredging during construction work. These changes can cause the kind of alterations observed in this study. The paucity of historical data on seasonal/annual/inter-annual variation in plankton abundance and biomass in the region, indeed in Gulf St Vincent in general, makes it difficult to draw accurate conclusions on the environmental effects of the ADP. Further data collection is required to ensure that any variation in cycles of plankton abundance and biomass in the region is properly understood so that changes in these parameters can be attributed to the correct cause, either natural cyclical variation or impacts of desalination. More accurate conclusions drawn from a greater range of data will provide information that may be a valuable means of alleviating environmental impacts and community perceptions and concerns about the impacts of the desalination process, and will aid in the development of strategies to minimise and mitigate the impacts of, and improve the efficiency of, the desalination process.

7. References

Anderson, D. M., Gilbert, P. M., and Burkholder, J. M. (2002) Harmful algal blooms and eutrophication: nutrient sources, composition and consequences. Estuaries 25 (4B): 704-726.

Baugh, K. A., Bush, J. M., Bill. B. D., Lefebvre, K. A., and Trainer, V. L. (2006) Estimates of specific toxicity in several *Pseudonitzscia* species from the Washington coast, based on culture and field studies. African Journal of Marine Science 28 (2): 403-407.

Bode, A., Alvarez-Ossorio, M. T., Gonzalez, N. (1998) Estimations of meso-zooplankton biomass in a coastal upwelling area off NW Spain. Journal of Plankton Research 20(5): 1005-1014.

Boeuf G. & Payan P. (2001) How should salinity influence fish growth? Comparative Biochemistry and Physiology C 130: 411-423.

Caron, D. A., Garneau, M., Seubert, E., Howard, M. D. A., Darjany, L., Schnetzer, A., Cetinic, I., Filteau, G., Lauri, P., Jones, B., and Trussell, S. (2010) Harmful algae and their potential impacts on desalination operations off southern California. Water Research 44: 385-416.

Cochlan, W. P., Herndon, J., and Kudela, R. M. (2008) Inorganic and organic nitrogen uptake by the toxigenic diatom *Pseudonitzschia australis* (Bacillariophyceae). Harful Algae 8 (1): 111-118.

Conover, R. J. (1978) Transformations of organic matter. *In* Kinne, O. (Ed.), *Marine Ecology V. 4. Dynamics*. John Wiley, Chichester, 221-456.

Falkowski, P. G., and Kolber, Z. (1995). Variations in chlorophyll fluorescence yields in phytoplankton in the world oceans. *Australian Journal of Plant Physiology* **22:** 341-355.

Fleming, I. A., and Kromkamp, J. (1994) Responses of respiration and photosynthesis of *Scenedesmus protuberans* Fritsch to gradual and steep salinity increases. Journal of Plankton Research 16: 1781-1791.

Garnaut, R. (2011) The Garnaut review 2001: Australia in the global response to climate change. Cambridge University Press, Melbourne.

Gilbert, P. M., Anderson, D. M., Gentien, P., Graneli, E., Sellner, K. G. (2005a) The global, complex phenomena of harmful algal blooms. Oceanography 18 (2): 137-147.

Gilbert, P. M., Seitzinger, S., Heil, C. A., Burkholder, J. M., Parrow, M. W., Codispoti, L. A., and Kelly, V. (2005b) The role of eutrophication in the global proliferation of harmful algae blooms. Oceanography 18 (2): 198-209.

Hallegraeff, G. M. (1993) A review of harmful algal blooms and their apparent global increase. Phycologia 32 (2): 79-99.

Hallegraeff, G. M. (2002) Aquaculturalists' Guide to Harmful Australian Microalgae. School of Plant Science, University of Tasmania, Hobart.

Hanson, C. E., Pattiaratchi, C. B. and Waite, A. M. (2005). Sporadic upwelling on a downwelling coast: Phytoplankton responses to spatially variable nutrient dynamics off the Gascoyne region of Western Australia. Continental Shelf Research **25**: 1561-1582.

Huntley, M., Boyd, C. M. (1984) Food-limited growth of marine zooplankton. The American Naturalist 124: 455-478.

Jassby, A. D., Platt, T. (1976) Mathematical formulation of the relationship between photosynthesis and light for phytoplankton. Limnology and Oceanography 21(4): 540-547.

Jeffrey, S. W., Wright, S. W. (2006) Photosynthetic pigments in marine microalgae: insights from cultures and the sea. In Subba Rao, D. V. (Ed.), Algal Cultures, Analogues of Blooms and Applications. Science Publishers, 33 - 90.

Jester, R. (2008) An investigation into the prevalence of *Alexandrium* derived toxins in marine food webs. Ph. D., University of California, Santa Cruz, pp. 116.

Kudela, R. M., Lane, J. Q., and Cochlan, W. P. (2008) The potential role of anthropogenically derived nitrogen in growth of harmful algae in Califronia, USA. Harmful Algae 8 (1): 103-111.

Latteman, S., and Hopner, T. (2008) Environmental impact and impact assessment of sea water desalination. Desalination 220: 1-15.

Leis, J.M., Trnski, T. (1989) The Larvae of Indo Pacific Shorefishes. New South Wales University Press, Sydney

Lohrenz, S. E., Wiesenberg, D. A., Rein, C. R., Arnone, R. A., Taylor, C. D., Knauer, G. A., Knap, A. H. (1992) A comparison of *in-situ* and simulated *in-situ* methods for estimating oceanic primary production. Journal of Plankton Research 14(2): 201-221.

Mackey, D. J., Parslow, J., Higgins, H. W., Griffiths, F. B., O'Sullivan, J. E. (1995) Plankton productivity and biomass in the western equatorial pacific: Biological and physical controls. Deep Sea Research Part 2 42(2-3): 499-533.

Madigan, M. T., Martinko, J.M., Parker, J. (1997) Brock Biology of Microorganisms. Prentice-Hall of Australia Pty. Limited, Sydney.

Maranon, E., Behrenfeld, M. J., Gonzalez, N., Mourino, B., Zubkov, M. V. (2003) High variability of primary production in oligotrophic waters of the Atlantic ocean: uncoupling from phytoplankton biomass and size structure. Marine Ecology Progress Series 257: 1-11.

Martin, J. H., 1990. Glacial-interglacial carbon dioxide change: The iron hypothesis. Paleoceanography 5, 1-13.

Martin, J. H., Fitzwater, S. E., 1988. Iron deficiency limits phytoplankton growthin the north-east Pacific subarctic. Nature 331, 341-343.

Martin, J. H., Gordon, R. M., Fitzwater, S., Broenkow, W. W., 1989. VERTEX: phytoplankton/iron studies in the Gulf of Alaska. Deep Sea Research 36, 649-680.

Miller, R. L. and Kamykowski, D. L. (1986) Short-term photosynthetic responses in the diatom *Nitzscia Americana* to a stimulated environment. Journal of Plankton Research 8: 305-315.

Miri, R., Chouikhi, A. (2005) Ecotoxicological marine impacts from seawater desalination plants. Desalination 182: 403-410.

Morton, A. J., Callister, I. K., and Wade, N. M. (1996) Environmental impacts of sea water distillation and RO processes. Desalination 108: 1-10.

Neira, F. J., Miskiewicz, A., Trnski, T. (1998) Larvae of temperate Australian fishes: laboratory guide for larval fish identification. University of Western Australia Press, Nedlands, W.A.

Parsons, T. R., Maita, Y., Lalli, C. M. (1984) A Manual of Chemical and Biological Methods for Seawater Analysis Pergamon Press Ltd., Oxford.

Pearce, I., Handlinger, J. H., Hallegraeff, G. M. (2005) Histopathology in Pacific oyster (*Crassostrea gigas*) spat caused by the dinoflagellate *Prorocentrum rhathymum*. Harmful Algae 4: 61-74.

Qasim, S. Z., Bhattathiri, P. M. A., and Devassy, V. P. (1972) The influence of salinity on the rate of photosynthesis and abundance of some tropical phytoplankton. Marine Biology 12: 200-206.

Rogers, P. J., Geddes, M., and Ward, T. M. (20036) Blue sprat *Spratelloides robustus* (Clupeidae: Dussumieriinae): a temperate clupeoid with a tropical life history strategy? Marine Biology 142: 809-824.

Sutherland, C. M. (2008) Diarrhetic shellfish toxins linked to local *Dinophysis* populations in the California coastal waters of Monterey Bay. Dissertation, University of California, Santa Cruz

Talling, J. F. (1957) The phytoplankton population as a compound photosynthetic system. The New Phytologist 56: 133-149.

van Ruth, P. D. (2010). Adelaide desalination project plankton characterisation study, prepared for Adelaide Aqua. SARDI Publication. South Australian Research and Development Institute (Aquatic Sciences), Adelaide. SARDI Publication No. F2010/000378-1. SARDI Research Report Series No. 487. 39 pp.

van Ruth, 2009. Spatial and temporal variation in primary and secondary productivity in the eastern Great Australian Bight. PhD thesis, the University of Adelaide, 192pp.

van Ruth, P., Thompson, P., Bonham, P., Jones, E. (2009) Primary productivity and zooplankton ecology in the Lincoln Offshore Subtidal Aquaculture Zone. Technical report, Aquafin CRC Project 4.6, FRDC Project 2005/059. Aquafin CRC, Fisheries Research & Development Corporation and South Australian Research & Development Institute (Aquatic Sciences), Adelaide. SARDI Publication No F2008/000789-1, SARDI Research Report Series No 343, 58 pp.

Varsamos S., Nebel C. & Charmantier G. (2005) Ontogeny of osmoregulation in postembryonic fish: a review. Comparative Biochemistry and Physiology - Part A: Molecular and Integrative Physiology 141: 401-429.

Wiebe, P. H. (1988) Functional regression equations for zooplankton displacement volume, wet weight, dry weight, and carbon. A correction. Fisheries Bulletin 86: 833-835.

Wiebe, P. H., Boyd, S., Cox, J. L. (1975) Relationship between zooplankton displacement volume, wet weight, dry weight and carbon. Fisheries Bulletin 73: 777-786.

Appendix 1: Phytoplankton species detected at Port Stanvac between December 2010 and November 2011.

Genus	Species	Genus	Species	Genus	Species
Diatoms		**Diatoms**		**Chrysophytes**	
Amphora	*sp.*	*Stauroneis*	*sp.*	*Calycomonas*	*sp.*
Asterionellopsis	*glacialis*	*Striatella*	*unipunctata*	*Dinobryon*	*sp.*
Bacillaria	*paxillifera*	*Thalassionema*	*sp.*	*Ochromonas*	*spp.*
Bacteriastrum	*elegans*	*Thalassiosira*	*sp.*		
Ceratoneis	*closterium*	*Thalassiosira*	*cf. mala*	**Haptophytes**	
Chaetoceros	*spp.*			*Calciopappus*	*caudatus*
Climacodium	*sp.*	**Dinoflagellates**		*Chrysochromulina*	*spp.*
Cocconeis	*spp.*	*Alexandrium*	*sp.*	*Corymbellus*	*sp.*
Coscinodiscus	*spp.*	*Amphidinium*	*sp.*	*Emiliania*	*huxleyi*
Cyclotella	*spp.*	*Amylax*	*sp.*	*Gephyrocapsa*	*oceanica*
Cylindrotheca	*closterium*	*Ceratium*	*furca*	*Hemiselmis*	*sp.*
Dactyliosolen	*fragilissimus*	*Ceratium*	*fusus*	*Leucocryptos*	*marina*
Diploneis	*sp.*	*Ceratium*	*lineatum*	*Phaeocystis*	*pouchetii cells*
Encyonema	*sp.*	*Ceratium*	*macroceros*	*Phaeocystis*	*sp.*
Entomoneis	*sp.*	*Ceratium*	*tripos*	*Plagioselmis*	*prolonga*
Fallacia	*sp.*	*Dinophysis*	*acuminata*	*Prymnesium*	*sp.*
Fragilaria	*sp.*	*Dinophysis/Phalachroma*	*mitra*	*Rhodomonas*	*salina*
Grammotophora	*serpentina*	*Gonyaulax*	*spp.*	*Teleaulax*	*acuta*
Guinardia	*delicatula*	*Gymnodinioid*	*spp.*		
Guinardia	*flaccida*	*Gyrodinium*	*spp.*	**Cryptophytes**	
Guinardia	*striata*	*Heterocapsa*	*rotundata*	*Hemiselmis*	*sp.*
Leptocylindrus	*danicus*	*Heterocapsa*	*triquetra*	*Leucocryptos*	*marina*
Leptocylindrus	*minimus*	*Karenia*	*mikimotoi*	*Plagioselmis*	*prolonga*
Licmophora	*sp.*	*Karenia*	*papilionacea*	*Teleaulax*	*acuta*
Lioloma	*sp.*	*Karlodinium*	*sp.*		
Mastogloea	*sp.*	*Katodinium*	*sp.*	**Prasinophytes**	
Minidiscus	*trioculatus*	*Noctiluca*	*scintillans*	*Micromonas*	*pusilla*
Minutocellus	*sp.*	*Oxyrrhis*	*marina*	*Nephroselmis*	*sp.*
Amphora	*sp.*	*Peridinium*	*sp.*	*Pyramimonas*	*spp.*
Asterionellopsis	*glacialis*	*Pronoctiluca*	*spinifera*	*Tetraselmis*	*spp.*
Naviculoid	*spp.*	*Prorocentrum*	*sp.*		
Nitzschia	*spp.*	*Prorocentrum*	*cordatum*	**Euglenophyta**	
Nitzschia	*longissima*	*Prorocentrum*	*gracile*	*Eutreptiella*	*spp.*
Nitzschia	*sigmoidea*	*Prorocentrum*	*micans*	*Trachelomonas*	*spp.*
Paralia	*sulcata*	*Prorocentrum*	*triestinum*		
Plagiotropis	*sp.*	*Protoceratium*	*reticulatum*	**Other**	
Pleurosigma	*sp.*	*Protoperidinium*	*spp.*	*Apedinella*	*spinifera*
Proboscia	*alata*	*Scrippsiella*	*spp.*	*Chattonella*	*spp.*
Pseudo-nitzschia	*delicatissima group*	*Takayama*	*pulchella*	*Dictyocha*	*fibula*
Pseudo-nitzschia	*fraudulenta/australis*	*Torodinium*	*sp.*	*Stephanoeca*	*sp.*
Pseudo-nitzschia	*turgidula*			*Unidentified*	*amoeba*
Rhizosolenia	*setigera*			*Unidentified*	*bodonids*
Skeletonema	*costatum/pseudocostatum*			*Unidentified*	*choanoflagellates*
				Mesodinium	*rubrum*

Appendix 2: Common names of zooplankton taxa detected at Port Stanvac between December 2010 and November 2011.

Taxa	Common name
Appendicularia	Larvacean
Barnacle nauplii	Barnacle larvae
Bivalvia	Bivalve
Bryozoa	Moss animals
Chaetognatha	Arrow worm
Cladocera	Water flea
Cnidaria	Jellyfish larvae
Copepoda	Copepod
Copepoda nauplii	Copepod larvae
Ctenophora	Comb jelly
Decapoda	Decapod (crab)
Gastropoda	Gastropod
Ostracoda	Ostracod
Polychaeta	Polychaete worm
Pluteus larvae	Sea urchin larvae
Veliger larvae	Gastropod larvae