



Adelaide Desalination Project Plankton Characterisation Study



Paul van Ruth

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SARDI Aquatic Sciences
PO Box 120 Henley Beach SA 5022

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Prepared for Adelaide Aqua



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South Australian Research and Development Institute

SARDI Aquatic Sciences
2 Hamra Avenue
West Beach SA 5024

Telephone: (08) 8207 5400

Facsimile: (08) 8207 5406

<http://www.sardi.sa.gov.au>

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Author(s): Paul van Ruth

Reviewer(s): Marty Deveney and Sasi Nayar

Approved by: Jason Tanner
Principal Scientist – Marine Environment & Ecology

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1. Executive Summary

Adelaide Aqua has begun the development of a 100 GL per annum seawater reverse-osmosis desalination plant at Port Stanvac in Gulf St Vincent (GSV). This development aims to provide a reliable water supply for the city of Adelaide, reducing the need to draw water from the River Murray. Studies assessing the impact of desalination plants on marine ecosystems recognise that the intake of seawater has the potential to entrain numerous plankton species. The brine discharge from the desalination process has also been shown to affect 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 underpin the marine ecosystem, since they form the primary food source in the oceanic food web. Herbivorous zooplankton feed upon the phytoplankton, and are in turn preyed upon by carnivorous higher consumers such as ichthyoplankton and fish. Thus, phytoplankton are gradually transformed into products available for consumption by higher trophic levels in the marine food chain. The risks that entrainment of plankton/increased salinity from brine discharge pose to the marine environment off Port Stanvac, and the food chain it supports, require investigation. This study was designed to contribute data to this investigation by characterising the plankton communities in the area around the intake and outlet pipes to be used by the Adelaide Desalination Project (ADP).

There were high phytoplankton abundances in the waters off Port Stanvac, with an April/May/June peak in abundances that appears to be a regular occurrence in the region. This pattern may be regional (within gulfs) and is similar to patterns identified in data from south-western Spencer Gulf (SWSG). However, there was a difference between SWSG and the Port Stanvac region in the high abundances of flagellates off Port Stanvac, which result in low chlorophyll *a* concentrations and low primary productivity. Zooplankton biomass and grazing impact was low, although abundances were relatively high due to a large number of smaller zooplankton in the community. Ichthyoplankton abundances were also low, similar to findings from previous studies in the region. A summer increase in the abundance of potentially harmful cyanobacteria was identified as a characteristic of the waters off Port Stanvac. In addition, several other potentially harmful/toxic harmful algal bloom (HAB) phytoplankton species were identified in the community off Port Stanvac, although infrequently and at low abundances.

Monitoring of 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 brine discharge and entrainment in the intake structure, will change the ecology of the plankton in the region, and may promote blooms of harmful/toxic algal species which have been identified as components of the plankton community off Port Stanvac. Further monitoring is required before the impact of the Adelaide desalination plant on the plankton community off Port Stanvac can be adequately assessed.

2. Introduction

The security of Adelaide's metropolitan water supply is affected by climatic variability and by Adelaide's limited water storage capacity (approximately one years' demand). With the impact of severe droughts and anticipated climate change likely to lead to reduced flows and greater variability of flows to local reservoirs, 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 desalination technology and other potential water security measures for the State in the future. Adelaide Aqua has since begun the development of a 100 GL per annum seawater reverse-osmosis desalination plant at Port Stanvac in Gulf St Vincent (GSV), which aims to provide a reliable water supply for the city of Adelaide, reducing the need to draw water from the River Murray.

Assessments of the impacts of desalination plants on marine ecosystems recognise that the intake of seawater has the potential to entrain a large number of different plankton species. 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 mesh ($\sim 4 \text{ mm}^2$) and reducing intake velocity, such that it is equivalent to ambient currents. Larger fish or larvae which are able to swim against the current are 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, and the removal of these important organisms from the water column may have impacts on food web structure.

The brine discharge from the desalination process has also been shown to affect marine ecosystems. This discharge has increased salinity compared with surrounding waters, and may contain small amounts of chemicals associated with desalination plant maintenance (such as antifouling/anti-scaling substances). An increase in salinity can affect phytoplankton physiological processes and lead to the death of sensitive larval species, ultimately leading to changes in species composition, species diversity and population density. 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 underpin the marine ecosystem, since they form the primary food source in the oceanic food web. Herbivorous secondary producers feed upon the phytoplankton, and are in turn preyed upon by carnivorous higher consumers such as ichthyoplankton and fish. Thus, light energy harnessed by the phytoplankton is gradually transformed into products available for consumption by higher trophic levels in the marine food chain. The risks posed by entrainment/increased salinity from brine discharge on regional food chains and ecosystem services including commercial and recreational fisheries require investigation. This study was designed to provide data to characterise the plankton communities in the area around the Adelaide Desalination Project intake and outlet pipes, to assist in identifying community structure, and identifying risk by characterising the plankton communities, quantifying the temporal variation in these communities, and quantifying variation in primary and secondary productivity in the region. There are few published studies on phytoplankton, zooplankton and productivity in GSV. Data available consist of chlorophyll *a* (chl *a*) concentrations, phytoplankton identifications and zooplankton settling volumes collected in southern

GSV during summer (February/March) sardine biomass surveys conducted by SARDI Aquatic Sciences (summarized in van Ruth, 2008, and SARDI Sardine spawning biomass reports e.g. Ward et al., 2009). Nayar et al. (2009) provide some seasonal information regarding total phytoplankton abundance and dominant species as part of the Adelaide Coastal Waters Study (ACWS) off Tennyson in mid GSV. Limited information on zooplankton and ichthyoplankton is available in Bryars (2003). More detailed information on ichthyoplankton (eggs and larvae) abundance and distribution within GSV can be found in the SARDI Sardine spawning biomass reports (e.g. Ward et al., 2009), and the studies of Dimmlich et al., 2004, Rogers et al., 2003, Rogers and Ward, 2007, and Ward et al., 2001. There is little information regarding temporal variation in plankton abundance and composition in GSV, and no information that is specific to the Port Stanvac ADP site. The objectives of this study were:

1. To identify plankton species (phytoplankton, zooplankton and ichthyoplankton) in the waters off the Adelaide Desalination Plant to species level where possible
2. To assess the abundance, composition and temporal variability of plankton species in the water column off the coast of the Adelaide Desalination Plant
3. To provide a comparison of plankton abundance and composition found at Port Stanvac to other regions in South Australia.
4. To examine seasonal variation in rates of primary and secondary productivity in the waters off the Adelaide Desalination Plant
5. To assess potential impact of entrainment of plankton species, and the presence of brine discharge on local marine scale-fish, lobster, abalone, prawn and blue crab fisheries and protected fish taxa
6. To assess how the entrainment of plankton species, and the presence of brine discharge may impact on the local food web in the Port Stanvac area
7. To provide Adelaide Aqua with detailed reports of the findings of the above surveys sufficient for a decision to be made on the likely environmental impacts to the plankton community of a desalination plant

3. Methods

SA Water provided baseline data for this study in the form of extracted chl *a* concentrations from samples collected monthly (without replication) during 2008 from stations SAW_D2 and SAW_I2 (Fig. 1) as part of a study into potential sites for intake and discharge structures for a pilot desalination plant off Port Stanvac, prior to the commencement of the ADP (Kildea et al., 2008).

Sampling for the ADP plankton characterisation study took place from June 2009 to April 2010 at two sites, the intake pipe (station IRP) and outlet pipe 4 (station ORP4) (Fig. 1). While sampling was intended to be monthly, adverse weather prevented sampling in July and September 2009. Samples for seasonal studies of primary productivity were collected from station PP, midway between station IRP and station ORP4, in August and October 2009, and January and April 2010.

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 ichthyoplankton 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; ORP4 = 138.47129 E 35.09255 S; PP = 138.46961 E 35.09158 S; SAW_I2 = 138.469452 E 35.09633 S; SAW_D2 = 138.465826 E 35.09237 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), to provide an assessment of the biomass distribution between major phytoplankton groups. Three litre samples were collected from 3 m depth, without replication, at each station with a Niskin bottle and kept cool in the dark until returned to the laboratory within 3 hours. Samples were filtered through stacked mesh (to retain cells $> 5 \mu\text{m}$) and Whatman GF/F filters (nominal pore size $0.4 \mu\text{m}$, to retain cells $< 5 \mu\text{m}$), allowing the examination of size fractionated phytoplankton biomass. Filters were frozen at -80°C prior to HPLC analysis at CSIRO Marine and Atmospheric Research, Hobart, Tasmania.

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 (= 2 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 $50 \mu\text{m}$ mesh net (30 cm net mouth diameter) lowered to within 1 m of the bottom and towed vertically to the surface. 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). 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). The five samples from each station were pooled during rinsing through a $35 \mu\text{m}$ mesh sieve. The contents of the sieves 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 50x). 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 group 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, (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 $< 1 \text{ cm}^3$, and a factor of 41 for samples with displacement volumes $1\text{-}10\text{cm}^3$ (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 $350 \mu\text{m}$ mesh bongo net (30 cm net mouth diameter). The net was lowered to ~ mid-

water depth, towed 50 m horizontally and then brought to the surface. 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 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 (recorded by flow meters suspended in the mouth of each net) multiplied by the area of the net mouth. Identifications used the guides of 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 in the dark until return to the laboratory within 3 hours. 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 ¹⁴C stock solution with an activity of 200 µCi ml⁻¹ was prepared by adding 1ml of sodium bicarbonate (GE Life Sciences NaH¹⁴CO₃, 2 mCi ml⁻¹, 55mCi mmol⁻¹) to 9ml of Na₂CO₃ solution (concentration 0.12 g L⁻¹). A known quantity of NaH¹⁴CO₃ (20 µCi) was mixed into each replicate bottle via the addition of 0.1 ml of ¹⁴C stock solution. Bottles were then incubated in a flow-through water bath for 24 hours at *in-situ* water temperatures in natural 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 ¹⁴CO₂. 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 CO₂ 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):

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

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):

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

Where $\sum P$ is the integral productivity, P_{\max}^b 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 ($P_{\max} = P_{\max}^b * \text{chl } a$). 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 SA Water baseline data

There was some variation in chlorophyll concentrations between stations sampled by SA Water in 2008 (Fig. 2). Concentrations were generally similar between the two stations from January to April, after which concentrations at station SAW_I2 were 0.05-0.6 $\mu\text{g L}^{-1}$ higher than concentrations at station SAW_D2 (with the exception of July). An initial spike in chl *a* concentration to $> 0.4 \mu\text{g L}^{-1}$ occurred in February 08, followed by a peak in May 08 of $\sim 0.8\text{-}1.1 \mu\text{g L}^{-1}$. There was another peak at station SAW_I2 in August ($\sim 0.7 \mu\text{g L}^{-1}$).

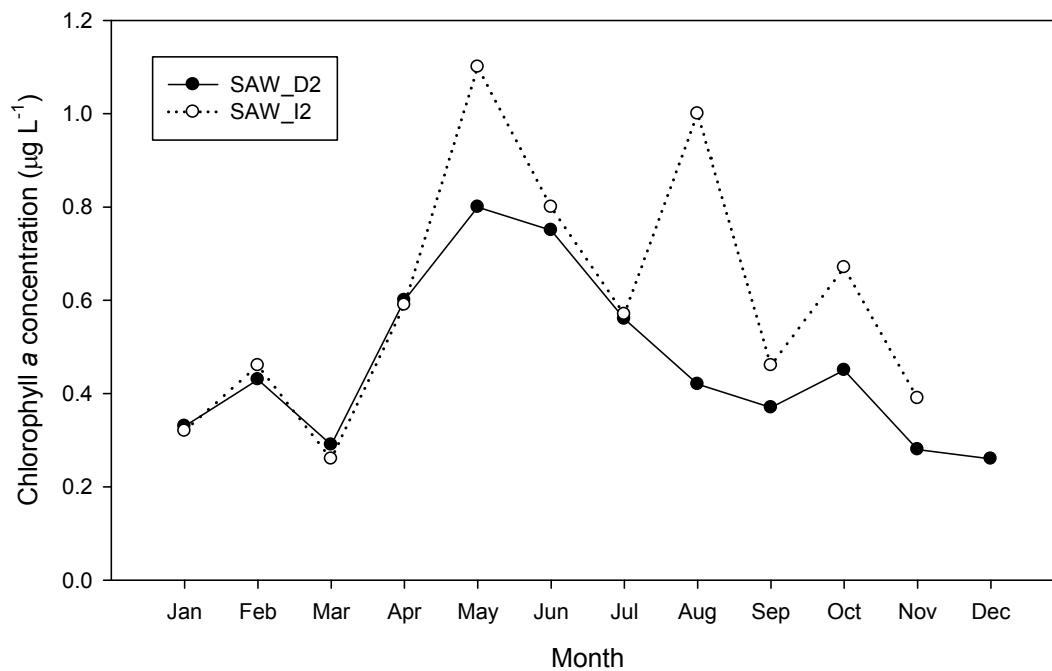


Figure 2. Monthly variation in chlorophyll *a* concentration in samples collected by SA Water off Port Stanvac in 2008. See Fig. 1 for station locations.

4.2 Phytoplankton biomass, abundance and community composition

There was little difference in total chl *a* concentrations between stations sampled as part of the ADP plankton characterisation study (Fig. 3), aside from the low concentration measured at IRP in January 2010 ($0.03 \mu\text{g L}^{-1}$). Chl *a* concentrations were generally $\sim 0.2 \mu\text{g L}^{-1}$, with a peak of $\sim 0.8 \mu\text{g L}^{-1}$ in June 2009, a small increase to $\sim 0.3 \mu\text{g L}^{-1}$ in October 2009, and another small peak of $\sim 0.4 \mu\text{g L}^{-1}$ in April 2010. Most of the phytoplankton biomass occurred in the small size fraction ($< 5 \mu\text{m}$, Fig. 4). Chl *a* concentrations in the large size fraction ($> 5 \mu\text{m}$) were generally $< 0.1 \mu\text{g L}^{-1}$, contributing little to the peaks in chl *a* mentioned above. No chl *a* was detected in the small size fraction in January, and given that it is highly unlikely that this size fraction was missing at this time, it is assumed that there was a problem with the analysis, so this value is not plotted.

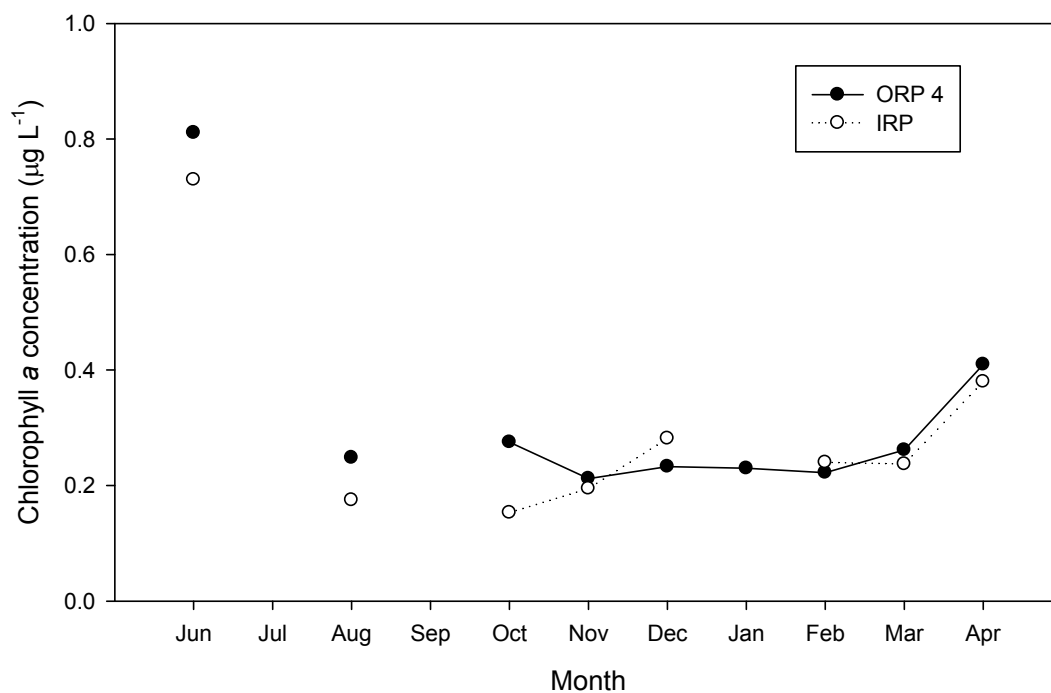


Figure 3. Monthly variation in chlorophyll *a* concentration during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. See Fig. 1 for station locations.

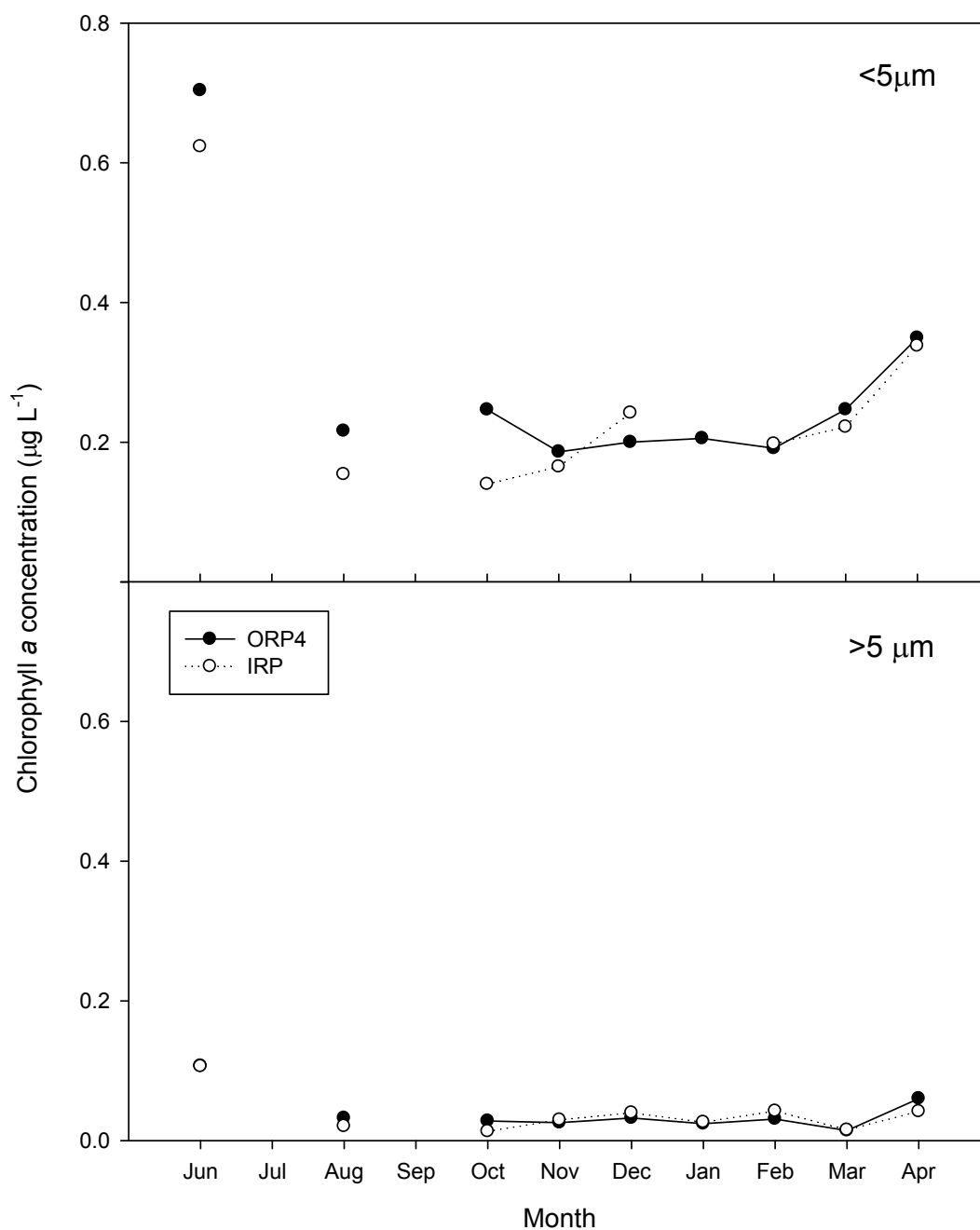


Figure 4. Monthly variation in size fractionated chlorophyll *a* during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. See Fig. 1 for station locations.

An examination of selected marker pigments in the small size fraction, normalised to chl *a*, suggests a strong seasonality in the types of phytoplankton found in the study area (Fig. 5). There was a spike in zeaxanthin (an indicator of the presence of cyanobacteria) in August 2009, with a sharp increase to a larger peak in February 2010. Chlorophyll *b* in the small size fraction peaked in June and October 2009, and again in April 2010, which, in the absence of lutein and prasinoxanthin, suggests an increase in the presence of prasinophytes in the community (Jeffrey and Wright, 2006).

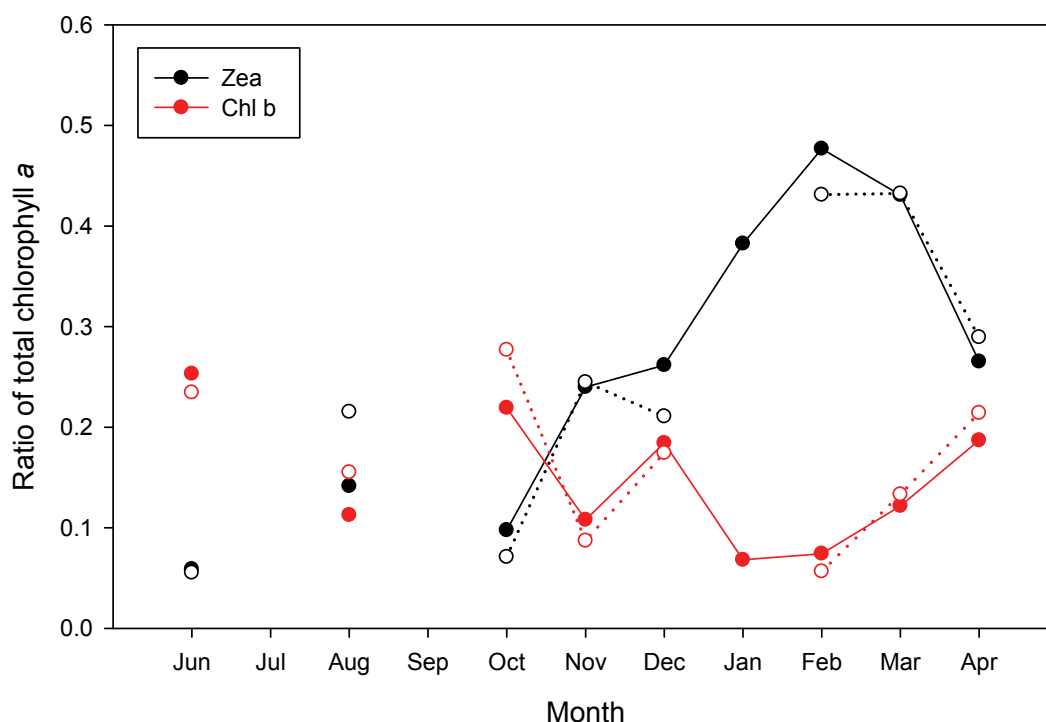


Figure 5. Monthly variation in the concentration of selected marker pigments normalised to chlorophyll *a* during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. Closed circles and solid lines represent samples from station ORP 4, open circles and dotted lines represent samples from station IRP. See Fig. 1 for station locations.

Total phytoplankton abundances showed similar patterns to pigment concentrations (Fig. 6). Abundances were high ($\sim 450,000$ cells L^{-1}) in June 2009, decreasing to $\sim 100,000$ cells L^{-1} in October before another spike to $\sim 400,000$ cells L^{-1} in November. Abundance decreased again in December, before gradually increasing through to a peak in abundances of $\sim 600,000$ cells L^{-1} in April 2010. Diatom abundances were low for most of this study ($< 50,000$ cells L^{-1} , Fig. 7), with a spike in January 2010 to $\sim 100,000$ cells L^{-1} due to an increase in abundance of *Leptocylindrus danicus* to $> 50,000$ cells L^{-1} , and peak of $\sim 400,000$ cells L^{-1} in April 2010 driven by increases in abundance of *L. danicus* (up to 130,000 cells L^{-1}), *L. minimus* ($\sim 18,000$ cells L^{-1}) and *Cylindrotheca closterium* ($\sim 10,000$ cells L^{-1}). Dinoflagellate abundances remained $< 50,000$ cells L^{-1} throughout the study period, with a small spike in abundances to $\sim 100,000$ cells L^{-1} in November 2009 due to increased abundance of Gymnodinoid spp. and *Heterocapsa rotundata* ($> 25,000$ cells L^{-1}). There were large variations in flagellate abundances, with a peak at $\sim 350,000$ cells L^{-1} in June 2009 attributed to high numbers of the Cryptophytes *Hemiselms* spp. ($> 65,000$ cells L^{-1}) and *Plagioselmis prolunga* ($> 130,000$ cells L^{-1}), decreasing to $\sim 50,000$ cells L^{-1} in October, before another peak at $\sim 300,000$ cells L^{-1} in November again driven by *Hemiselms* spp. (up to 126,000 cells L^{-1}) and *Plagioselmis prolunga* (up to 44,000 cells L^{-1}). From January to April 2010 flagellate abundances were $> 100,000$ cells L^{-1} , rising to another *Hemiselms* spp./*Plagioselmis prolunga* driven peak at $> 150,000$ cells L^{-1} in April (Fig. 7)

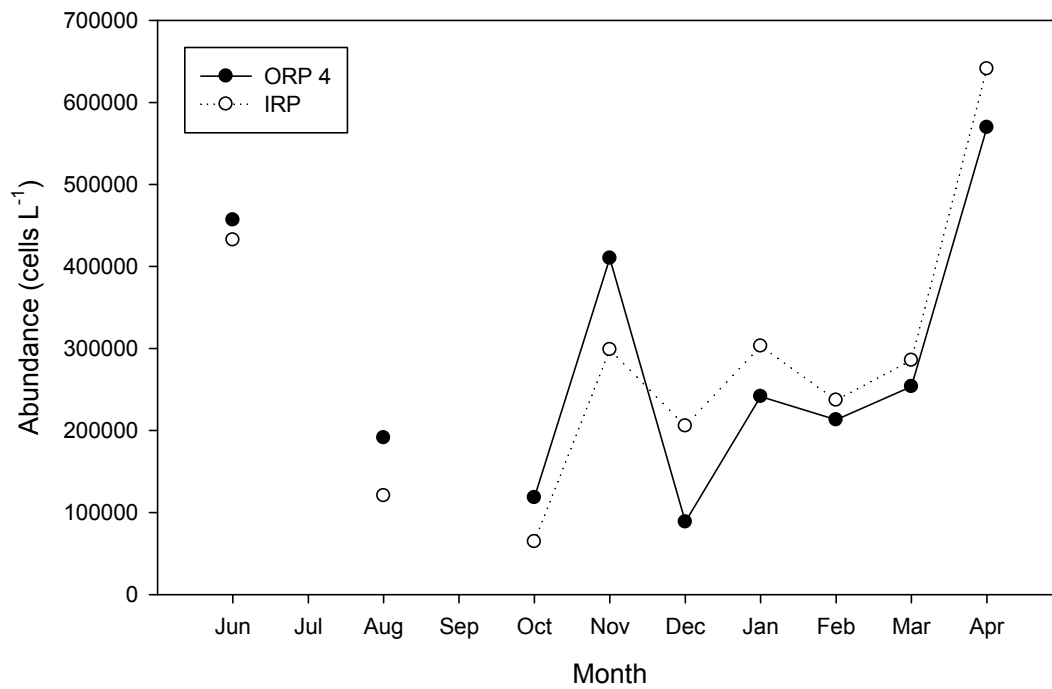


Figure 6. Monthly variation in total phytoplankton abundance during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. See Fig. 1 for station locations.

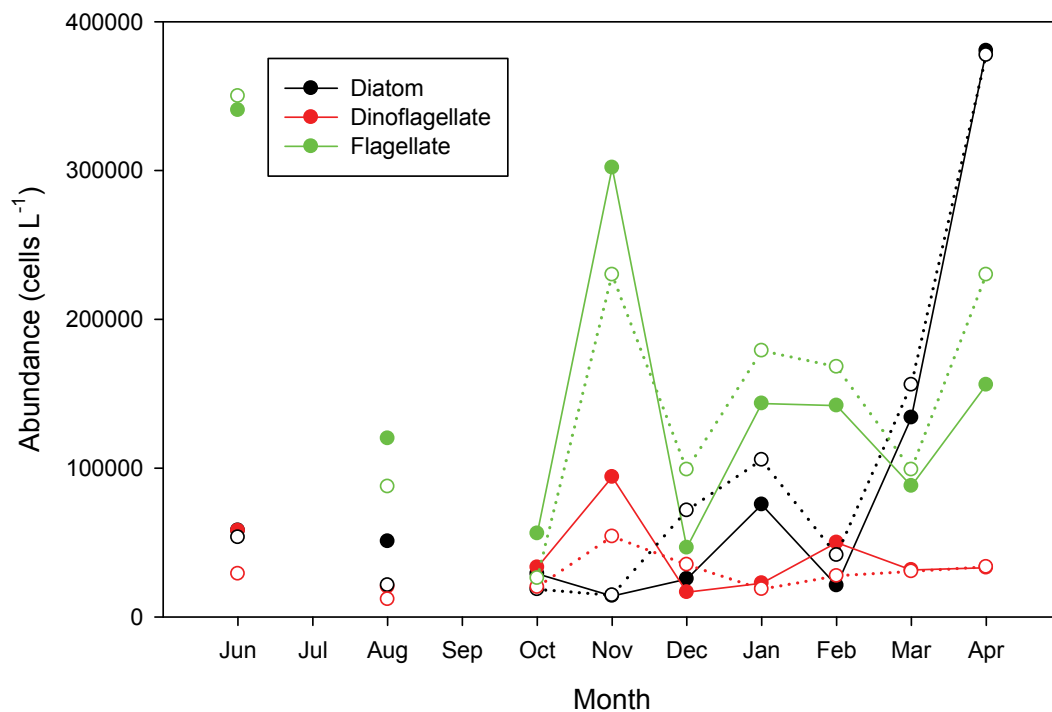


Figure 7. Monthly variation in phytoplankton abundance during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. Closed circles and solid lines represent samples from station ORP 4, open circles and dotted lines represent samples from station IRP. See Fig. 1 for station locations.

There were no clear patterns in species richness evident between the two stations, but the general temporal trend was for S to peak in June 2009 at ~40 species, then decline through spring to 24 species at station IRP in October 2009 and 25 species at station ORP4 in January 2010 (Fig. 8). At station IRP, S then had a general upward trend to a peak of 38 species in April 2010, while at ORP4, a peak of 34 species was observed in February 2010, before a further decline to 25 species through March and April. A full list of phytoplankton species identified during the study is provided in Appendix 1. Temporally persistent diatoms included *Chaetoceros* spp., *Cylindrotheca closterium*, *Nitzschia* spp., and *Thalassiosira cf. mala*, which were present in all months of the study, but rarely in abundances $> 6,000$ cells L^{-1} . *Minidiscus trilocatus* was found in June and August 2009 and again from January to April 2010, in abundances $< 6,000$ cells L^{-1} . The rest of the diatoms listed in Appendix 1 occurred infrequently, and at abundances $< 6,000$ cells L^{-1} , with the exception of *Skeletonema costatum/pseudocostatum*, which numbered 30,000 cells L^{-1} at station IRP in April 2010. The dinoflagellates Gymnodinoid spp., *Gyrodinium* spp., *Heterocapsa rotundata*, and *Scropsiella* spp. were present in all months of the study, and dominated the dinoflagellate community. Other dinoflagellates listed in Appendix 1 were found on few occasions, and only in abundances < 5000 cells L^{-1} . Flagellates present year round included the potentially ichthyotoxic haptophyte *Chrysochromulina* spp., which reached abundances of $\sim 30,000$ cells L^{-1} in June and August 2009 and again in February and April 2010. The haptophyte *Emeliana huxleyi* was also found year round in abundances $> 10,000$ cells L^{-1} . The cryptophytes *Hemiselmis* spp., *Leucocryptos marina*, *Plagioselmis prolunga*, and *Teleaulax acuta*, prasinophytes *Pyramimonas* spp. and *Tetraselmis* spp., and the euglenophyte *Eutreptiella* spp. were also present in relatively high abundances through the study period.

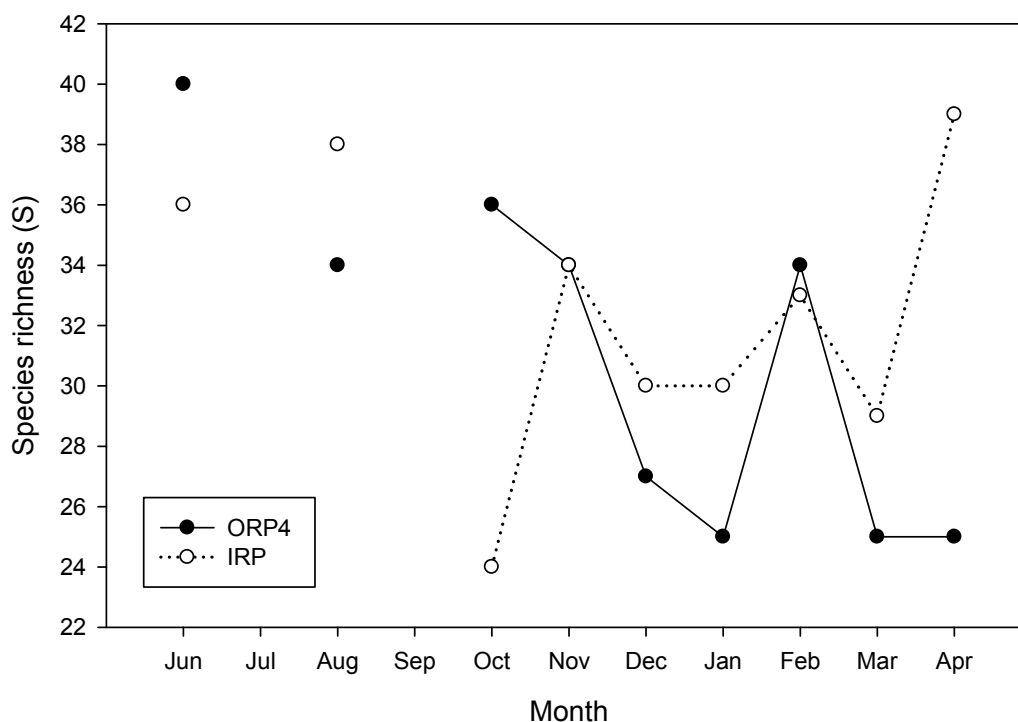


Figure 8. Monthly variation in species richness (S) during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. See Fig. 1 for station locations.

4.3 Zooplankton biomass, abundance and community composition

There were no clear patterns in zooplankton biomass between stations or times of day (Fig. 9). Biomass decreased from June to November 2009 before increasing to peak in night samples in April 2010. Day samples peaked again in February/March 2010 before decreasing in April 2010. There were similar patterns in zooplankton abundance (Fig. 10), with a decline from a peak in June through to October 2009, and a peak in abundance in February/March 2010 followed by a decline in April.

Zooplankton species richness showed no clear patterns between stations or times of day. Species richness (S) increased from June 2009 to peak at 14-17 species in August, before falling to 3-6 species in October (Fig. 11). A general trend of increasing species richness then occurred throughout to January/February 2010 (10-12 species), before a small drop to ~8-10 species in March/April. Copepoda and Copepoda nauplii (see Appendix 2 for common names of all zooplankton taxa) dominated the zooplankton community during the ADP plankton characterisation study (Fig. 12). Veliger larvae, and Cnidaria were also prominent. In Fig. 12, zooplankton taxa which contributed < 3% to the total community were grouped together as “other”. These taxa included Amphipoda, cypris larvae, barnacle nauplii, Chaetognatha, Cladocera, Decapoda, Gastropoda, pluteus larvae, Thaliacea and trocophore larvae. Copepoda and Copepoda nauplii were present in high abundances in all months of the study. Veliger larvae were prominent in the region from June to August 2009 and again from February to April 2010. Cnidaria were abundant through March/April 2010, while Polychaeta, Appendicularia and Bivalvia were present throughout the study. Bryozoa were present June through August 2009, then from February to April 2010, and Ostracoda were abundant from June to August 2009.

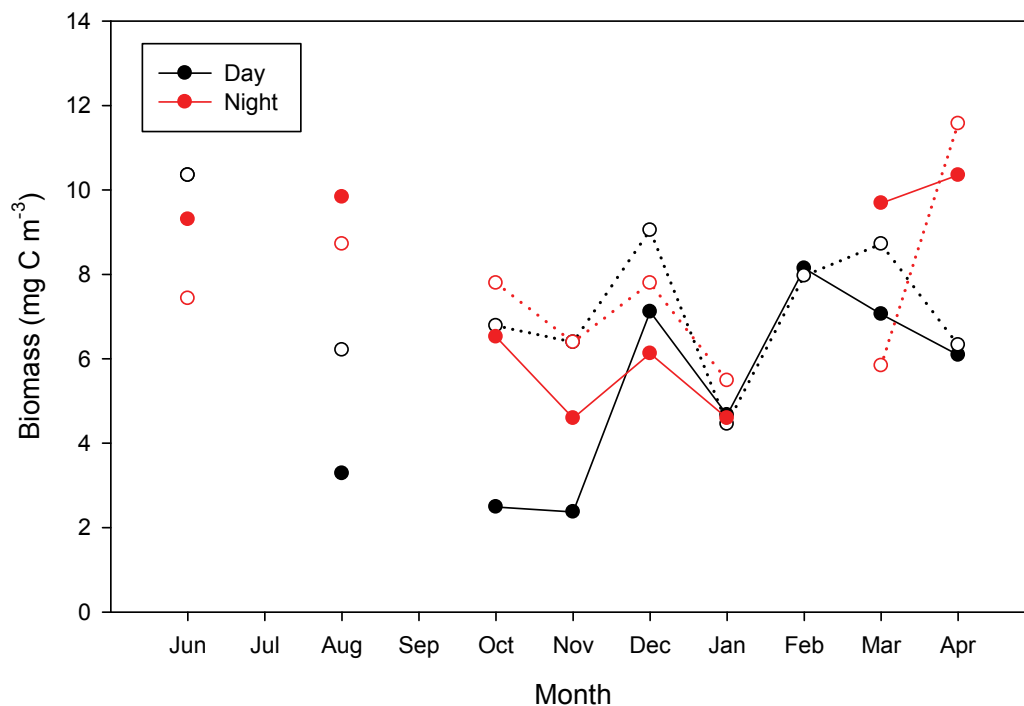


Figure 9. Temporal variation in zooplankton biomass during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. Solid lines represent biomass at station ORP4, dotted lines represent biomass at station IRP. See Fig. 1 for station locations.

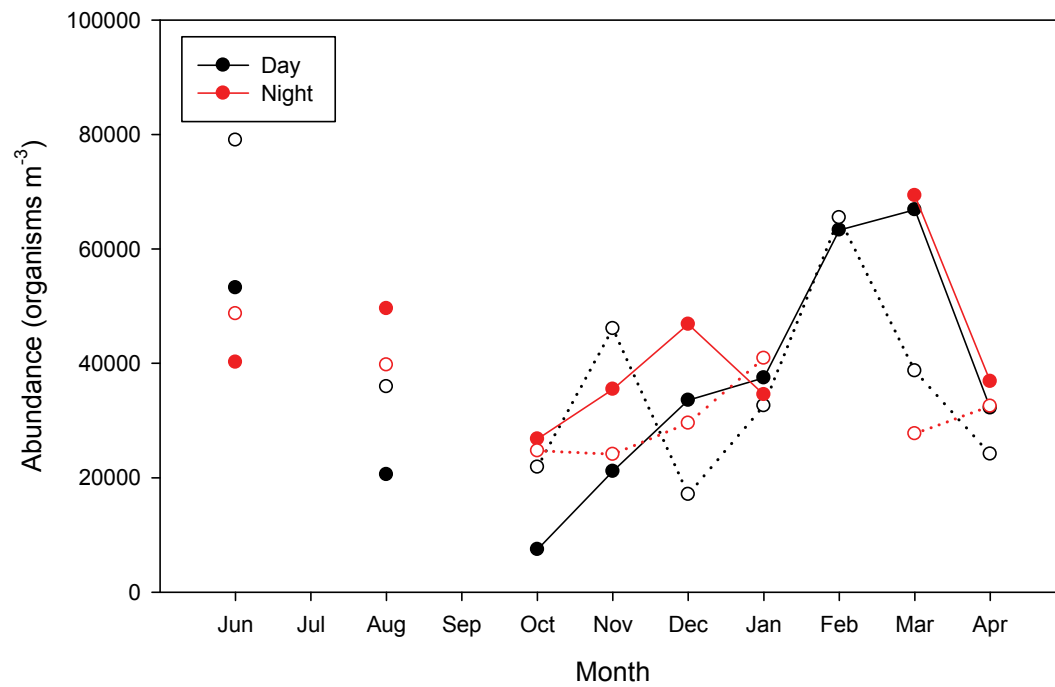


Figure 10. Temporal variation in zooplankton abundance during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. Solid lines represent biomass at station ORP4, dotted lines represent biomass at station IRP. See Fig. 1 for station locations.

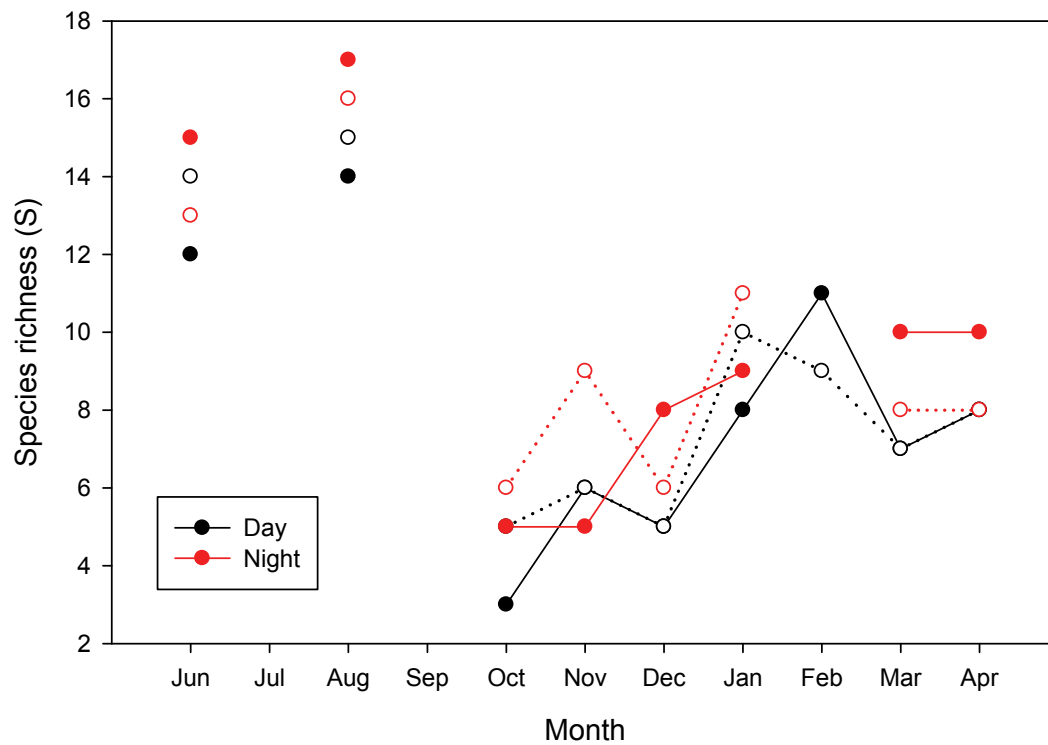


Figure 11. Temporal variation in zooplankton species richness (S) during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. Solid lines represent S at station ORP4, dotted lines represent S at station IRP. See Fig. 1 for station locations.

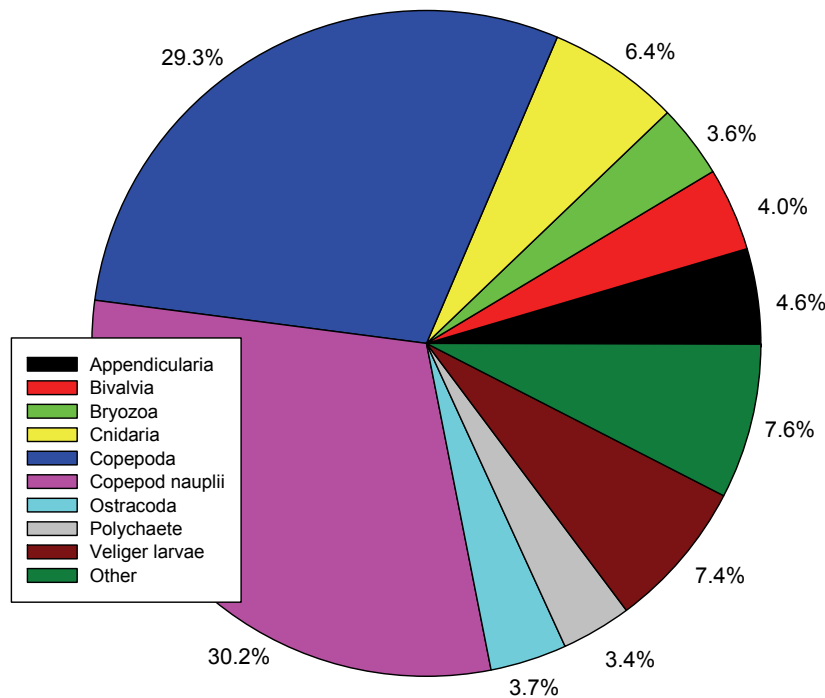


Figure 12. Proportional composition, based on abundance, of the zooplankton community during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010.

4.4 Ichthyoplankton abundance and community composition

No clear patterns in egg abundance were evident between stations or times of day. For most of the study abundances were < 3 eggs m^{-3} (Fig. 13). Both stations had similar egg numbers in midday samples, with highest egg numbers in October 2009 (~ 4 eggs m^{-3}) decreasing to \sim zero eggs m^{-3} by February 2010. There was considerable variation between midnight samples, however, with a peak in egg numbers at ~ 6 eggs m^{-3} in November 2009 at IRP and in January 10 at ORP4, before a decline through February/March 2010. Highest midnight egg numbers at both stations occurred in April 2010.

There was a large proportion of unidentified eggs in the ichthyoplankton community over the course of this study, and a large proportion of sardine (*Sardinops sagax*) eggs. Anchovy (*Engraulis australis*) and leatherjacket eggs were also present (Fig. 14). Unidentified eggs were found year round, with highest abundances in October and November 2009. Anchovy eggs were present between August 2009 and January 2010, with highest abundances at ORP4 in January ($0.5-1.76$ eggs m^{-3}). Sardine eggs were present throughout the study, and were in highest numbers in April 2010 (up to 10 eggs m^{-3}). Leatherjacket eggs were only identified in midnight samples in August 2009, at low abundance (0.75 eggs m^{-3}).

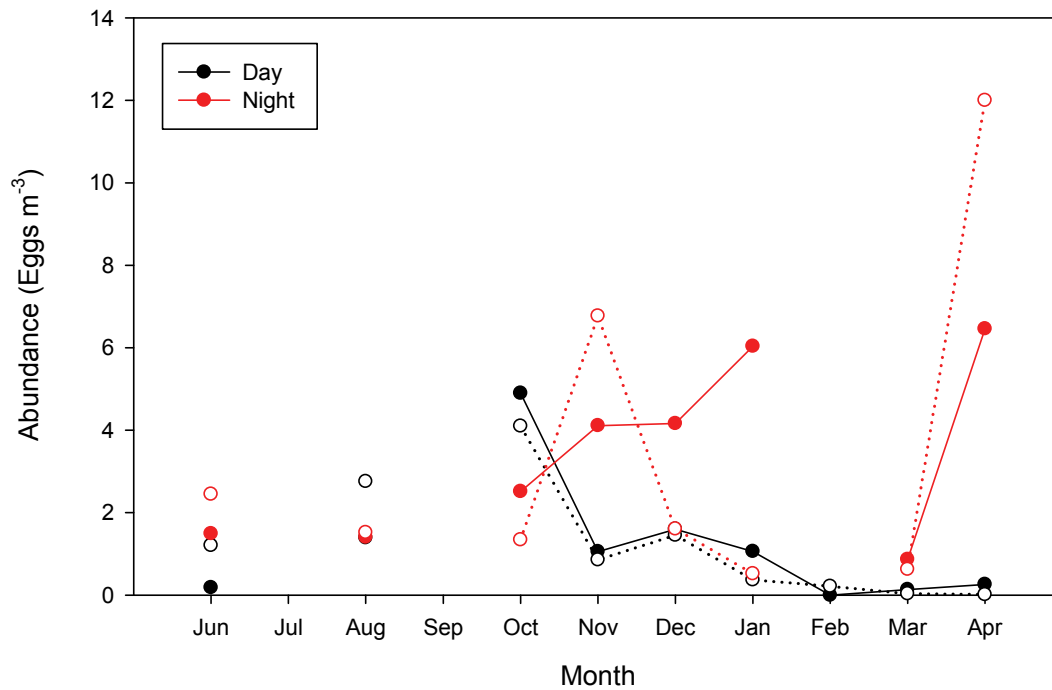


Figure 13. Temporal variation in egg numbers during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. Solid lines represent egg number at station ORP4, dotted lines represent egg numbers at station IRP. See Fig. 1 for station locations.

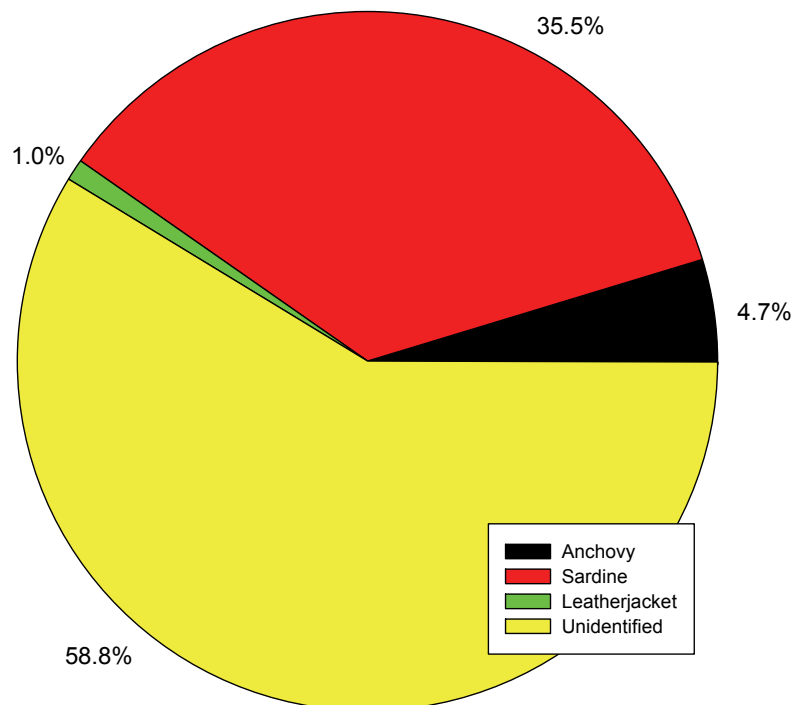


Figure 14. Proportional composition of egg types in the community during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010.

There was considerable temporal variation in larval abundance, and much variation between stations and times of day (Fig. 15). Overall, however, larval abundance was low, < 0.9 larvae m^{-3} throughout the study. Peaks in midnight larval abundance occurred at station ORP4 in August 2009 and January 2010, with peaks at IRP in December 2009 and April 2010. Midday peaks in larval abundance were recorded at ORP4 in November 2009 and at IRP in February 2010. Abundance at both stations at both midday and midnight increased in April 2010 from lows of < 0.1 larvae m^{-3} in March.

Unidentified larvae dominated the community throughout the study, with a large proportion of anchovy larvae also present (Fig. 16). Leatherjacket, sardine and goby larvae made up smaller proportions of the total larval community. Unidentified larvae were present year round, with highest abundances in midnight samples in December 2009 and January 2010, and at station ORP4 at midnight in April 2010. Anchovy larvae were found at night in August, October and December 2009 in low abundance ($0.1-0.3$ larvae m^{-3}), then from January to April 2010, with highest abundances in April 2010 (peak 0.74 larvae m^{-3}). Leatherjacket larvae were most abundant in June and August 2009 ($< 0.1-0.3$ larvae m^{-3}), with sardine larvae present in June, August and October 2009 and again in January/February 2010. Abundances in these months were generally very low (< 0.1 larvae m^{-3}), with a peak at midnight at station ORP4 of 0.24 larvae m^{-3} . Goby larvae were only identified in midnight samples in August and October 2009, and March 2010, at abundances < 0.1 larvae m^{-3} .

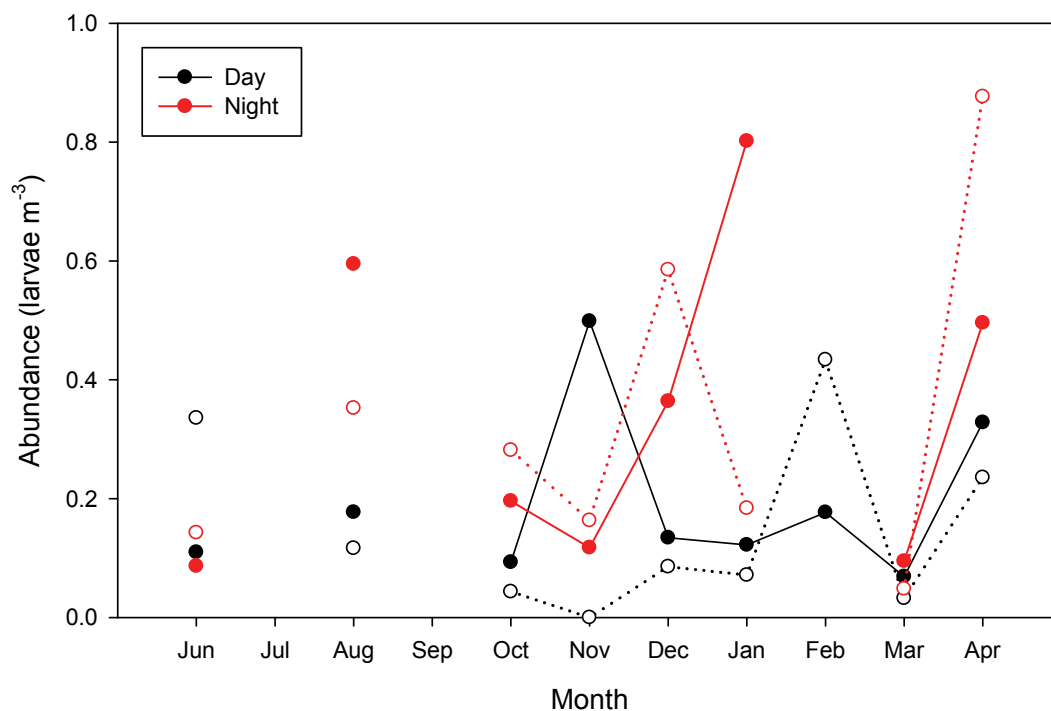


Figure 15. Temporal variation in larval abundance during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. Solid lines represent larval abundance at station ORP4, dotted lines represent larval abundance at station IRP. See Fig. 1 for station locations.

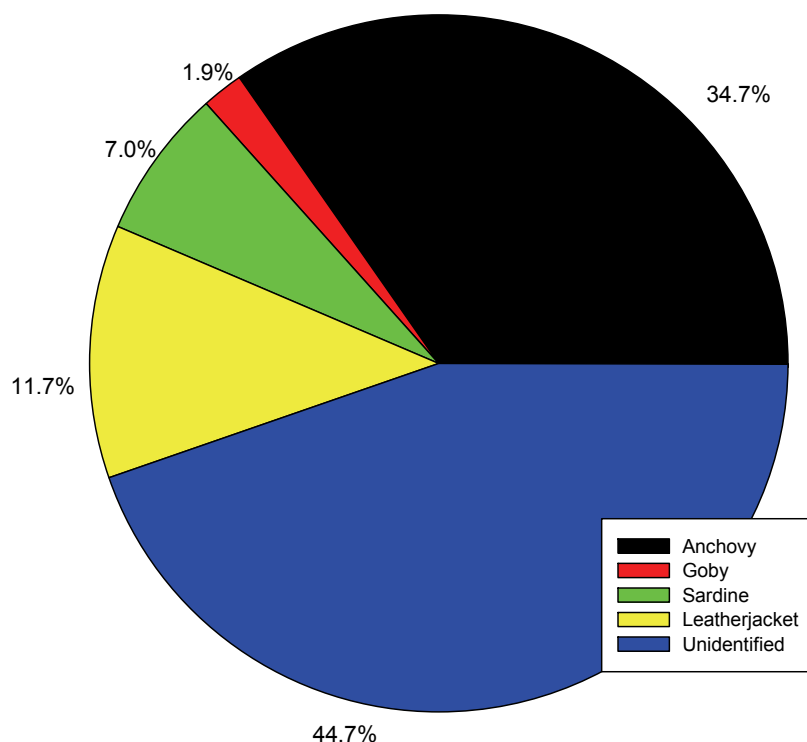


Figure 16. Average proportional composition of ichthyoplankton larval community during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010.

4.5 Primary productivity

Modelled rates of primary productivity agreed well with measured results (Fig. 17). There was high temporal variation in photosynthetic parameters (Table 1). P_{\max}^b ranged between 1.20 and 3.91 mg C (mg chl)⁻¹ hr⁻¹, with highest values in January and lowest values in April 2010. Photosynthetic efficiency ranged between 0.025 and 0.047 mg C (mg chl)⁻¹ hr⁻¹ (μmol m⁻² s⁻¹)⁻¹, with highest α in August 2009 and lowest in April 2010. Daily integral productivity ranged from 95.9 to 227.1 mg C m⁻² d⁻¹, with the highest productivity in January 2010 > twice the value if the lowest productivity which occurred in April 2010 (Table 1, Fig. 18). Phytoplankton turnover times decreased from August into October 2009 before peaking in January 2010. Highest gross phytoplankton growth rate occurred in April 2010 (Fig. 18).

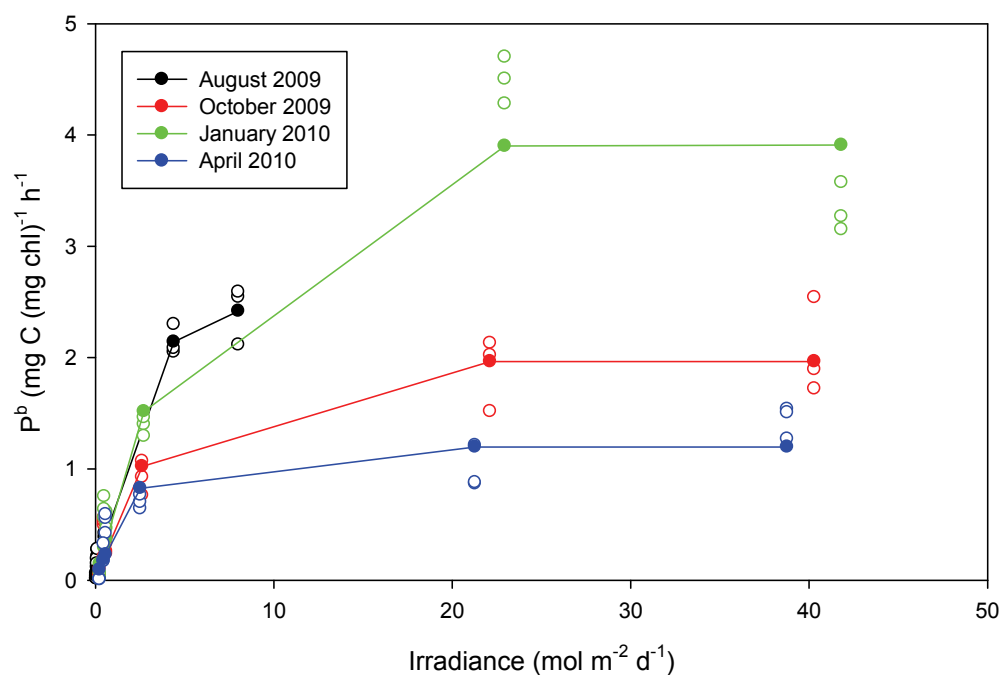


Figure 17. Seasonal variation in photosynthesis – irradiance curves from ^{14}C experiments conducted during the Adelaide Desalination Project plankton characterisation study on water collected from station PP. Open circles indicate measured rates of primary productivity, closed circles indicate rates 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 in decimal hours, I'_o is the irradiance just below the sea surface ($\mu\text{mol m}^{-2} \text{s}^{-1}$), K_d is the attenuation coefficient of downwelled irradiance (m^{-1}), Chl a is surface extracted chlorophyll a concentration ($\mu\text{g L}^{-1}$), I_k is the irradiance corresponding to the onset of light saturation of photosynthesis ($\mu\text{mol m}^{-2} \text{s}^{-1}$), α is the photosynthetic efficiency ($\text{mg C (mg chl)}^{-1} \text{hr}^{-1} (\mu\text{mol m}^{-2} \text{s}^{-1})^{-1}$), P^b_{max} is the biomass specific maximum photosynthetic rate ($\text{mg C (mg chl)}^{-1} \text{hr}^{-1}$), and DIP is the daily integral productivity ($\text{mg C m}^{-2} \text{d}^{-1}$).

Month	D_{irr}	I'_o	K_d	R^2	Chl a	I_k	Alpha	P^b_{max}	DIP
Aug-09	10.95	990	0.16	0.92	0.25	52.4	0.05	2.45	137.0
Oct-09	12.13	1018	0.17	0.89	0.28	72.8	0.03	1.97	118.0
Jan-10	14.13	1945	0.18	0.95	0.23	106.5	0.04	3.91	227.1
Apr-10	11.25	1360	0.21	0.95	0.41	47.7	0.03	1.20	95.9

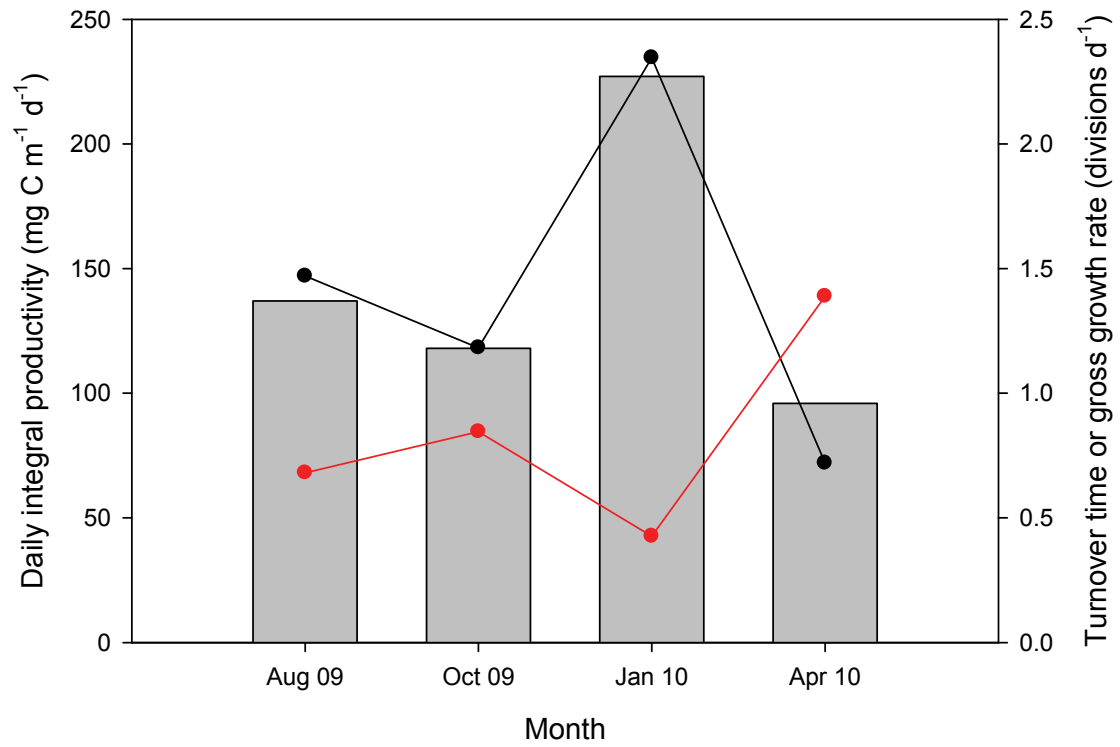


Figure 18. Seasonal variation in daily integral productivity during the Adelaide Desalination Project plankton characterisation study. Black line represents turnover time, red line represents gross phytoplankton growth rate.

4.6 Secondary productivity

G_{\max} increased with increasing SST during this study to peak in December 2009/January 2010 before declining as SST decreased with the coming of autumn (Table 2). There were no clear patterns in zooplankton grazing rate between stations or times of day (Table 2, Fig. 19). Grazing rate peaked at ~ 6 to $8 \text{ mg C m}^{-3} \text{ d}^{-1}$ in December 2009, then declined in January 2010 before a midday peak at $\sim 7 \text{ mg C m}^{-3} \text{ d}^{-1}$ in February/March 2010, and a decline in midday grazing in April. There was a peak in midnight grazing in April 2010 at $\sim 8 \text{ mg C m}^{-3} \text{ d}^{-1}$ (Fig. 19). Zooplankton grazed between 18.8 and 43.2% of phytoplankton standing stock per day during this study. 25.4% of standing stock was grazed in August 2009, declining to 18.8% of standing stock in October. The greatest grazing impact occurred in the latter parts of the study, with 43.2% of standing stock grazed in October 2010, and 38.2% grazed in April.

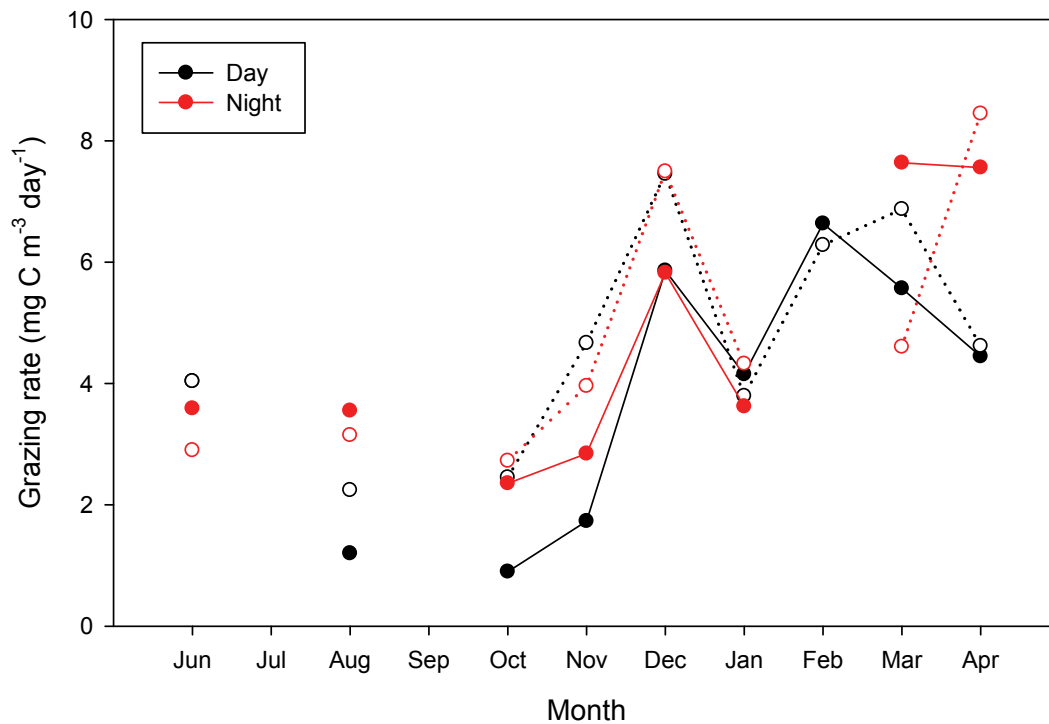


Figure 19. Temporal variation in zooplankton grazing rate during the Adelaide Desalination Project plankton characterisation study between June 2009 and April 2010. Solid lines represent biomass at station ORP4, dotted lines represent biomass at station IRP. See Fig. 1 for station locations.

Table 2. Temporal variation in zooplankton settling volume (ml m^{-3}), biomass (mg m^{-3}), and grazing rate ($\text{mg C m}^{-3} \text{d}^{-1}$) during the Adelaide Desalination Project plankton characterisation study. SST = CTD measured sea surface temperature ($^{\circ}\text{C}$), 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	June	Day	1.41	10.35	15	0.27	4.04
IRP	June	Night	1.01	7.43	15	0.27	2.90
ORP4	June	Day	1.41	10.35	15	0.27	4.04
ORP4	June	Night	1.27	9.31	15	0.27	3.59
IRP	August	Day	0.85	6.21	14	0.25	2.24
IRP	August	Night	1.19	8.72	14	0.25	3.15
ORP4	August	Day	0.45	3.29	14	0.26	1.20
ORP4	August	Night	1.34	9.83	14	0.25	3.55
IRP	October	Day	0.92	6.78	14	0.25	2.45
IRP	October	Night	1.06	7.80	14	0.24	2.72
ORP4	October	Day	0.34	2.49	14	0.25	0.90
ORP4	October	Night	0.89	6.52	14	0.25	2.35
IRP	November	Day	0.87	6.39	20	0.51	4.67
IRP	November	Night	0.87	6.39	19	0.43	3.96
ORP4	November	Day	0.32	2.37	20	0.51	1.73
ORP4	November	Night	0.63	4.59	19	0.43	2.84
IRP	December	Day	1.23	9.05	22	0.58	7.46
IRP	December	Night	1.06	7.80	23	0.67	7.49
ORP4	December	Day	0.97	7.11	22	0.58	5.86
ORP4	December	Night	0.83	6.13	23	0.67	5.82
IRP	January	Day	0.61	4.45	22	0.60	3.79
IRP	January	Night	0.75	5.49	21	0.55	4.33
ORP4	January	Day	0.63	4.67	22	0.62	4.15
ORP4	January	Night	0.63	4.59	21	0.55	3.62
IRP	February	Day	1.08	7.96	21	0.55	6.28
ORP4	February	Day	1.11	8.14	21	0.57	6.64
IRP	March	Day	8.00	8.72	21	0.55	6.88
IRP	March	Night	5.50	5.84	21	0.55	4.61
ORP4	March	Day	6.00	7.06	21	0.55	5.57
ORP4	March	Night	8.00	9.69	21	0.55	7.64
IRP	April	Day	5.50	6.33	20	0.51	4.62
IRP	April	Night	9.50	11.57	20	0.51	8.45
ORP4	April	Day	5.00	6.09	20	0.51	4.45
ORP4	April	Night	8.00	10.35	20	0.51	7.56

5. Discussion

ADP plankton characterisation study pigment data corresponded well with baseline pigment data provided by SA Water. This suggests that the April/May/June peak in phytoplankton biomass and abundance is a regular pattern in the region. A similar pattern was identified in phytoplankton biomass and abundance in south western Spencer Gulf (SWSG) (van Ruth et al, 2009a), which also found that the majority of phytoplankton biomass in SWSG was in the small size fraction (< 5 μm). The May chl *a* peak in SWSG, however, was double the June peak observed off Port Stanvac during this study. There were also differences in total phytoplankton abundance between SWSG and Port Stanvac communities, with abundances in SWSG generally 50% less than total abundances recorded off Port Stanvac during this study. This difference, despite the higher chl *a* concentrations in SWSG, is due to differences in dominant phytoplankton between the regions. Chl *a* concentration provides an indication of phytoplankton biomass only, and can be influenced by many factors, including phytoplankton cell size, with larger cells containing more chlorophyll than smaller cells. It is, therefore, clearly possible to have a phytoplankton community dominated by fewer large cells (such as diatoms, as found in SWSG) that has a higher chl *a* concentration than a community of many smaller cells (such as flagellates, as found off Port Stanvac in this study). This also explains why the November peak in abundance off Port Stanvac, which was driven by an increase in flagellate abundance, did not result in an increase in chl *a*. Flagellate abundances off Port Stanvac in this study were always at least 10,000 cells L^{-1} greater than abundances in SWSG, and up to $\sim 250,000$ cells L^{-1} greater, while diatom abundances off Port Stanvac were generally similar to abundances in SWSG, although the April peak was $\sim 100,000$ cells L^{-1} lower than the peak in SWSG (van Ruth et al., 2009a). The March/April pattern of phytoplankton abundance observed in this study (diatoms > flagellates > dinoflagellates) is similar to results reported for February/March in lower GSV (van Ruth, 2008), where chl *a* concentrations ranged between < 0.1 and 0.5 $\mu\text{g L}^{-1}$. Nayar et al. (2009) reported total phytoplankton abundances off Tennyson of $\sim 5,000$ to 29,000 cells L^{-1} in a diatom dominated community, with highest abundance in August, and low abundance in October. In a similar result to this study, the February diatom community was dominated by *Leptocylindrus sp.* (which became dominant in the diatoms off Port Stanvac in January-April 2010). Abundances off Tennyson were an order of magnitude lower than abundances measured during this study, however, possibly due to the absence of flagellates in the samples of Nayar et al. (2009) (these may not have been sampled/identified/counted by that study due to their small size).

We found several potentially harmful/toxic harmful algal bloom (HAB) phytoplankton species in the community off Port Stanvac. These 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. All *Chaetoceros* species possess setae, but it is only the species that have setae with barbs (*C. concavicornis* and *C. convolutus*) that are potentially harmful to fish (Hallegraeff, 2002). While *Chaetoceros* spp. were present in all months of this study, potentially harmful species were not identified. Potentially toxic diatoms found in the waters off Port Stanvac include *Pseudonitzschia delicatissima*, *P. fraudulenta/australis* and *P. pungens/multiseriis*, which are the most likely cause of amnesic shellfish poisoning (Hallegraeff, 2002).

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 during this study. *Alexandrium catenella* is a producer of toxins that cause paralytic shellfish poisoning in humans, 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* can cause diarrhetic shellfish poisoning in humans (Hallegraeff, 2002). Ichthyotoxic flagellates identified during this study include the haptophytes *Chrysochromulina* spp., a dominant flagellate throughout the study period, and *Heterosigma* sp. With the exception of *Chaetoceros* spp. and *Chrysochromulina* spp., these harmful/toxic species of phytoplankton were found infrequently at very low abundances ($< 2,000$ cells L⁻¹). Despite this, their presence warrants ongoing monitoring should changes to ecological conditions in the region resulting from ADP operations provide conditions suitable for these harmful/toxic phytoplankton to bloom. Such changes may include variations in nutrient concentrations due to increased terrestrial run-off/wastewater outflow or resuspension of sediments due to dredging during construction work, changes to irradiances from increased turbidity due to terrestrial run-off and dredging during construction, and variations in temperature, salinity and levels of stratification due to brine discharge. These changes will cause fluctuations in the dominant species in the phytoplankton community, as species with different photosynthetic requirements and different growth and resource uptake rates compete with each other for available resources. Reduced zooplankton grazing and microbial infection of phytoplankton due to entrainment of these organisms in the intake pipe may also promote phytoplankton blooms.

Analysis of marker pigments by HPLC provided data about the phytoplankton community composition that would have been missed if this study had relied only on microscopic identification. Marker pigments are particularly useful for providing information about phytoplankton $< 5\mu\text{m}$ in size, which are generally too small to be identified by light microscopy. These results indicate an increase in cyanobacteria in the waters off Port Stanvac through summer. A similar trend was observed in SWSG during the Risk and Response project, although the peak in SWSG was about half the size of the peak measured during this study (van Ruth et al., 2009a). Cyanobacteria are more tolerant to environmental extremes than phytoplankton, and are often the sole phototrophic organisms found in extreme environments, such as highly saline 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 using HPLC would be wise given that increased salinity in the vicinity of the ADP outflow structures has the potential to promote cyanobacterial blooms.

Peaks in zooplankton biomass and abundance roughly corresponded to peaks in phytoplankton biomass and abundance. Monthly variation in zooplankton biomass was similar to patterns observed in SWSG which had increases in March, May and July 2007, and low biomass in December 2007 (van Ruth et al., 2009b). The peaks in zooplankton biomass recorded during this study were, however, an order of magnitude lower than peaks observed in SWSG (van Ruth et al., 2009b), and were comparable to biomass measured in the more oligotrophic (low nutrient) offshore regions of the eastern Great Australian Bight (van Ruth, 2009). Despite the variation in biomass between Port Stanvac and SWSG, zooplankton abundances measured

during this study were similar to those measured by van Ruth et al. (2009b). This is most likely because the smaller mesh net used in this study (50 μm compared with 150 μm in the Risk and Response project) was able to capture more of the smaller zooplankton that would flow through coarser mesh nets. Indeed, many small zooplankton were identified in Port Stanvac samples, such as copepod nauplii, which made up a large proportion of the community. These organisms were not identified in large numbers in SWSG, but may have been had a finer mesh net been used. With this in mind, it is likely that with identical sampling effort, differences in zooplankton abundances between SWSG and Port Stanvac would be larger.

The dominance of smaller zooplankton in the Pt Stanvac community when compared to the community of SWSG may also be explained by the variation in dominant phytoplankton groups between the regions. Greater abundances of small flagellates in the waters off Port Stanvac would provide an ample food source for smaller zooplankton. The dominant diatoms in SWSG, on the other hand, are too large to be consumed by copepod nauplii, which are only present as a small proportion of the total SWSG community (van Ruth et al., 2009b). Indeed, the March/April increases in diatom abundances recorded in this study corresponded to decreases in copepod nauplii abundance, and increases in abundance of larger zooplankton such as Cnidaria. No evidence of the presence of lobster, abalone, prawn or blue crab larvae was found in the waters off Port Stanvac during this study. Although Decapoda and Gastropoda were found in zooplankton samples, they were a very small proportion of the total zooplankton community during this study, contributing < 0.4% and < 2% to the total community respectively.

Ichthyoplankton egg and larval abundance were low during this study, corresponding with previous studies in GSV. When converted to eggs m^{-2} (by multiplying by depth) to allow comparisons with previous studies, sardine egg and larval numbers off Port Stanvac during this study (generally 0-50, but up to 180 eggs m^{-2} at the April 2010 peak, and 0.2 to 4.5 larvae m^{-2}) were comparable to ranges reported for lower GSV during the sardine spawning biomass seasons (January to March) of 1995-1999 (0-500 eggs m^{-2} , Ward et al., 2001) and 2009 (0-500 eggs m^{-2} , 0-10 larvae m^{-2} , Ward et al., 2009). Higher egg and larval abundances, however, were found in the main spawning regions of the eastern Great Australian Bight (up to 5000 eggs m^{-2} and 5000 larvae m^{-2}), supporting the presumption made in the ADP EIS that sardines are likely to be transient in the Port Stanvac area. Anchovy densities in GSV ranged between 0 and 100 eggs m^{-3} and 0-50 larvae m^{-3} in February/March 2000 and 2001 (Dimmlich et al., 2004), with the highest abundances in upper GSV. Abundances in lower GSV were between 0 and 10 eggs and larvae m^{-3} , comparable to values measured in this study. Unidentified eggs and larvae made up a large proportion of the ichthyoplankton 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 ichthyoplankton further by next-generation DNA analysis, to confirm identifications and provide data on the identity of unidentified samples. It is possible, however, that some of the unidentified species recorded in this study are the clupeids blue sprat (*Spratelloides robustus*) and sandy sprat (*Hyperlophus vittatus*). Blue sprat have been recorded throughout GSV in abundances of up to 0.2 larvae m^{-3} (Rogers et al., 2003), with sandy sprat measured in abundances of up to 0.2 eggs m^{-3} (Rogers and Ward, 2007). No other commercially important marine scale-fish larvae or protected fish taxa were identified during this project.

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 ($<400 \text{ mg C m}^{-2} \text{ d}^{-1}$, van Ruth, 2009), the Leeuwin current off south west Western Australia ($110\text{-}530 \text{ mg C m}^{-2} \text{ d}^{-1}$, Hanson et al., 2005), and the north and south Atlantic sub-tropical gyres ($18\text{-}362 \text{ mg C m}^{-2} \text{ d}^{-1}$, Maranon et al., 2003). Rates were comparable to the lowest rate of primary productivity measured in SWSG ($\sim 200 \text{ mg C m}^{-2} \text{ d}^{-1}$ in October 2007) by van Ruth et al. (2009b). Rates of primary productivity alone, however, 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 January 2010, but this did not translate into a peak in phytoplankton biomass or abundance. There was an increase in abundance from a December 2009 low which was driven by a spike in flagellate and diatom numbers, but the phytoplankton growth rate was lowest in January 2010, and grazing impact was highest, which together prevented the accumulation of significant phytoplankton biomass despite the high rate of primary productivity. Lowest primary productivity was measured in April 2010, but phytoplankton growth rate was at its highest in this month, more than three times the growth rate recorded for January. This high growth rate allowed the accumulation of significant phytoplankton biomass during April, despite low rates of primary productivity and a high grazing impact. G'_{max} calculated for the waters off Port Stanvac were comparable to values recorded for SWSG during the Risk and Response project (van Ruth et al., 2009b), 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. As a result, the grazing impact of zooplankton on phytoplankton biomass was also lower, $> 40\%$ less impact in Port Stanvac waters in comparison with the waters of SWSG.

6. Summary and conclusions

There was high phytoplankton abundance in the waters off Port Stanvac, with an April/May/June peak in abundance that appears to occur there seasonally (albeit based on only two years data). This pattern may be regional (occurring in both gulfs) given the similarities to data from SWSG. There was a difference between SWSG and the Port Stanvac region, however, in the high abundances of flagellates off Port Stanvac, which result in low chl *a* concentrations and low primary productivity. Zooplankton biomass and grazing impact was also low, although abundances were relatively high due to a large number of smaller zooplankton in the community. Ichthyoplankton abundances were also low, in agreement with previous studies in the region.

This study should be viewed as a preliminary study, effectively providing baseline data before the commencement of ADP operations. Monitoring the plankton community should be continued to assess the impact of the working desalination plant. It is possible that activities associated with the working plant, such as brine discharge and entrainment in the intake structure, will change the ecology of the plankton in the region, and may promote blooms of harmful/toxic algal species which have been identified as components of the plankton community off Port Stanvac. Further monitoring is required before the impact of the Adelaide desalination plant on the plankton community off Port Stanvac can be adequately assessed.

7. References

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Appendix 1: Phytoplankton species detected at Port Stanvac between June 2009 and April 2010.

Genus	Species	Genus	Species	Genus	Species
Diatoms		Diatoms		Chrysophytes	
<i>Amphora</i>	<i>sp.</i>	<i>Rhizosolenia</i>	<i>setigera</i>	<i>Calycomonas</i>	<i>sp.</i>
<i>Asterionellopsis</i>	<i>glacialis</i>	<i>Skeletonema</i>	<i>costatum/pseudocostatum</i>	<i>Dinobryon</i>	<i>sp.</i>
<i>Attheya</i>	<i>sp.</i>	<i>Stauroneis</i>	<i>sp.</i>	<i>Meringiosphaera</i>	<i>mediterranea</i>
<i>Bacillaria</i>	<i>paxillifera</i>	<i>Thalassionema</i>	<i>sp.</i>	<i>Ochromonas</i>	<i>spp.</i>
<i>Bacteriastrium</i>	<i>elegans</i>	<i>Thalassiosira</i>	<i>sp.</i>		
<i>Cerataulina</i>	<i>sp.</i>	<i>Thalassiosira</i>	<i>cf. mala</i>	Haptophytes	
<i>Chaetoceros</i>	<i>spp.</i>			<i>Calciopappus</i>	<i>caudatus</i>
<i>Climacodium</i>	<i>sp.</i>	Dinoflagellates		<i>Chrysochromulina</i>	<i>spp.</i>
<i>Cocconeis</i>	<i>spp.</i>	<i>Alexandrium</i>	<i>sp.</i>	<i>Emiliana</i>	<i>huxleyi</i>
<i>Coscinodiscus</i>	<i>spp.</i>	<i>Alexandrium</i>	<i>catenella/fundyense</i>	<i>Gephyrocapsa</i>	<i>oceanica</i>
<i>Cyclotella</i>	<i>spp.</i>	<i>Amphidinium</i>	<i>sp.</i>	<i>Paraphysomonas</i>	<i>sp.</i>
<i>Cymbella</i>	<i>sp.</i>	<i>Ceratium</i>	<i>furca</i>	<i>Phaeocystis</i>	<i>sp.</i>
<i>Cylindrotheca</i>	<i>closterium</i>	<i>Ceratium</i>	<i>fuscus</i>	<i>Phaeocystis</i>	<i>pouchetii cells</i>
<i>Dactyliosolen</i>	<i>fragilissimus</i>	<i>Ceratium</i>	<i>lineatum</i>		
<i>Diploneis</i>	<i>sp.</i>	<i>Ceratium</i>	<i>macroceros</i>	Cryptophytes	
<i>Encyonema</i>	<i>sp.</i>	<i>Ceratium</i>	<i>pentagonum</i>	<i>Goniomonas</i>	<i>truncata</i>
<i>Entomoneis</i>	<i>sp.</i>	<i>Ceratium</i>	<i>tripos</i>	<i>Hemiselmis</i>	<i>sp.</i>
<i>Fallacia</i>	<i>sp.</i>	<i>Dinophysis</i>	<i>acuminata</i>	<i>Leucocryptos</i>	<i>marina</i>
<i>Fragilaria</i>	<i>sp.</i>	<i>Goniodoma</i>	<i>sp.</i>	<i>Plagioselmis</i>	<i>prolonga</i>
<i>Gomphonema</i>	<i>sp.</i>	<i>Gonyaulax</i>	<i>spp.</i>	<i>Rhodomonas</i>	<i>salina</i>
<i>Grammotophora</i>	<i>marina</i>	Gymnodinioid	<i>spp.</i>	<i>Teleaulax</i>	<i>acuta</i>
<i>Guinardia</i>	<i>flaccida</i>	<i>Gyrodinium</i>	<i>spp.</i>		
<i>Guinardia</i>	<i>striata</i>	<i>Heterocapsa</i>	<i>rotundata</i>	Chlorophytes	
<i>Hemiaulus</i>	<i>sp.</i>	<i>Heterocapsa</i>	<i>triquetra</i>	<i>Chlamydomonas</i>	<i>sp.</i>
<i>Leptocylindrus</i>	<i>danicus</i>	<i>Karenia</i>	<i>papillionacea</i>	<i>Dunaliella</i>	<i>sp.</i>
<i>Leptocylindrus</i>	<i>minimus</i>	<i>Noctiluca</i>	<i>scintillans</i>	<i>Unidentified</i>	5um coccoid
<i>Licmophora</i>	<i>sp.</i>	<i>Oxyrrhis</i>	<i>marina</i>		
<i>Lioloma</i>	<i>sp.</i>	<i>Peridinium</i>	<i>sp.</i>	Prasinophytes	
<i>Minidiscus</i>	<i>tricolatus</i>	<i>Prorocentrum</i>	<i>sp.</i>	<i>cf. Dunaliella</i>	<i>sp.</i>
<i>Minutocellus</i>	<i>sp.</i>	<i>Prorocentrum</i>	<i>compressum</i>	<i>Micromonas</i>	<i>pusilla</i>
<i>Naviculoid</i>	<i>spp.</i>	<i>Prorocentrum</i>	<i>cordatum</i>	<i>Nephroselmis</i>	<i>sp.</i>
<i>Nitzschia</i>	<i>spp.</i>	<i>Prorocentrum</i>	<i>dentatum</i>	<i>Pseudoscourfieldia</i>	<i>sp.</i>
<i>Nitzschia</i>	<i>longissima</i>	<i>Prorocentrum</i>	<i>emarginatum</i>	<i>Pyramimonas</i>	<i>spp.</i>
<i>Nitzschia</i>	<i>sigmoidea</i>	<i>Prorocentrum</i>	<i>gracile</i>	<i>Tetraselmis</i>	<i>spp.</i>
<i>Odontella</i>	<i>sp.</i>	<i>Prorocentrum</i>	<i>micans</i>	<i>Unidentified</i>	<i>flagellate</i>
<i>Plagiotropis</i>	<i>sp.</i>	<i>Prorocentrum</i>	<i>rhathymum</i>		
<i>Pleurosigma</i>	<i>sp.</i>	<i>Prorocentrum</i>	<i>triestinum</i>	Euglenophyta	
<i>Proboscia</i>	<i>alata</i>	<i>Protopteridinium</i>	<i>spp.</i>	<i>Eutreptiella</i>	<i>spp.</i>
<i>Pseudo-nitzschia</i>	<i>delicatissima</i> group	<i>Protopteridinium</i>	<i>bipes</i>		
<i>Pseudo-nitzschia</i>	<i>fraudulenta/australis</i>	<i>Protopteridinium</i>	<i>minutum</i>	Cyanoprokaryota	
<i>Pseudo-nitzschia</i>	<i>heimii</i>	<i>Protopteridinium</i>	<i>steinii</i>	<i>Gloeocapsa</i>	<i>sp.</i>
<i>Pseudo-nitzschia</i>	<i>pungens/multiseries</i>	<i>Scrippsiella</i>	<i>spp.</i>	<i>Phormidium</i>	<i>sp. (filaments)</i>
<i>Pseudosolenia</i>	<i>calcar-avis</i>	<i>Takayama</i>	<i>sp.</i>		
<i>Rhizosolenia</i>	<i>spp.</i>				

Appendix 1 (cont.)

Genus	Species
Other	
<i>Apedinella</i>	<i>spinifera</i>
<i>Ebria</i>	<i>tripartita</i>
<i>Dictyocha</i>	<i>fibula</i>
<i>Heterosigma</i>	<i>sp.</i>
<i>Pseudopedinella</i>	<i>pyriforme</i>
<i>Stephanoeca</i>	<i>sp.</i>
<i>Unidentified</i>	<i>amoeba</i>
<i>Unidentified</i>	<i>bodonids</i>
<i>Mesodinium</i>	<i>rubrum</i>

Appendix 2: Common names of zooplankton taxa detected at Port Stanvac between June 2009 and April 2010.

<u>Taxa</u>	<u>Common name</u>
Amphipoda	Amphipod
Appendicularia	Larvacean
Cypris larvae	Barnacle larvae
Barnacle nauplii	Barnacle larvae
Bivalvia	Bivalve
Bryozoa	Moss animals
Chaetognatha	Arrow worm
Cladocera	Water flea
Cnidaria	Jellyfish larvae
Copepoda	Copepod
Copepoda nauplii	Copepod larvae
Decapoda	Decapod
Gastropoda	Gastropod
Ostracoda	Ostracod
Polychaeta	Polychaete worm
Pluteus larvae	Sea urchin larvae
Thaliacea	Salp
Trochophore larvae	Polychaete larvae
Veliger larvae	Gastropod larvae