

2 Comparison of nitrifier activity versus growth in the Scheldt estuary - a turbid, tidal estuary in northern Europe

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2.1 Introduction

Nitrification, the microbial oxidation of ammonium (NH_4^+), is one of the key nitrogen transformation processes. It is performed under oxic conditions by 2 groups of bacteria, one group that oxidizes ammonium to nitrite (NO_2^-) and a second group that further oxidizes nitrite to nitrate (NO_3^-). Nitrification links organic matter mineralization (ammonification) and nitrogen removal through denitrification (conversion of nitrate to nitrogen gas). Nitrifying bacteria are chemolithoautotrophic organisms which use energy from the oxidation of NH_4^+ or NO_2^- to fix inorganic carbon in their biomass. Accordingly, nitrification links the flows of nitrogen with those of oxygen and carbon. In ammonium-rich systems, nitrification may make significant contributions to, or even dominate, total oxygen consumption and carbon fixation. Nitrifiers were responsible for up to 60 % of the total O_2 consumption in the Seine estuary, France (Garnier et al., 2001). Nitrification is the most important carbon fixation process in the upper part of the Scheldt estuary (Soetaert and Herman, 1995), where it accounts for 12 to 78 % of oxygen consumption (Gazeau et al., 2005). One by-product formed during nitrification is nitrous oxide gas (N_2O), which contributes to global warming and ozone depletion. de Wilde and de Bie

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(2000) showed that a major portion of N_2O production in the Scheldt estuary results from nitrification, and that almost all of it is lost to the atmosphere within the estuary and is not transported out to sea. In recognition of the pivotal role of nitrification in estuarine biogeochemistry, research has been initiated to identify the organisms and governing factors of this process so that it may be accurately quantified (Bollmann and Laanbroek, 2002; de Bie et al., 2002b; Caffrey et al., 2003). One of the most common methods to measure nitrification rates is the N-serve sensitive ^{14}C -bicarbonate incorporation technique (Somville, 1978; Brion and Billen, 1998), henceforth referred to as the ^{14}C method. It is based on the incorporation of inorganic carbon by growing nitrifiers. Nitrification rates measured with the ^{14}C method are expressed as the amount of carbon incorporated over time. However, many researchers are interested in nitrogen transformations (rather than nitrifier growth) and use a conversion factor to express these carbon incorporation rates into nitrogen oxidation rates. This approach requires (1) a tight coupling between nitrifier activity (NH_4^+ oxidation to NO_2^- or NO_3^-) and growth (carbon incorporation), and (2) a fixed stoichiometry between the quantity of nitrogen transformed and carbon fixed. Although these 2 requirements are essential for the conversion of C incorporation rates into N oxidation rates, their validity has not been tested thoroughly in natural conditions.

Nitrifying bacteria are relatively slow growing, growth rates between 0.04 and 0.06 h^{-1} (Helder and de Vries, 1983), implying that nitrifying communities need time to adapt to environmental changes. Organisms in estuarine systems are submitted to variable biogeochemical conditions. Seasonal factors like temperature, nutrient availability and oxygen concentration can affect nitrifier activity and growth (Carlucci and McNally, 1969; Goreau et al., 1980; Berounsky and Nixon, 1993; Bodelier et al., 1996; Bollmann et al., 2002). Estuarine nitrifier populations are subject to large environmental variations because of tides and strong salinity gradients across estuaries. Different populations of ammonium oxidizing bacteria occur at estuarine sites with different salinities (de Bie et al., 2001; Bollmann and Laanbroek, 2002). In turbid estuaries, nitrifying bacteria are attached to suspended particulate matter in a manner similar to that in a “fluidized bed reactor” (Owens, 1986; Brion and Billen, 2000). Particle-attached nitrifiers experience a longer residence time in the estuary, which allows them to develop their population (Brion et al., 2000).

However, these particles with associated nitrifiers are subject to repeated cycles of settling and resuspension. Given these multiple causes of environmental variability, one might question whether nitrifier growth (as measured with the ^{14}C method) and activity are coupled. The main aim of the present study was to assess whether nitrifier activity and growth are so tightly coupled that nitrogen oxidation can be estimated from carbon incorporation in combination with conversion factors. In parallel with the ^{14}C method, nitrification rates in the Scheldt estuary were measured with a stable isotope technique where ^{15}N labeled NH_4^+ was added and the appearance of ^{15}N in NO_3^- after incubation was measured.

2.2 Materials and methods

2.2.1 Study Area

The Scheldt estuary is located in the southwest of the Netherlands and Belgium. The estuary is fed by the Scheldt river which originates in northern France (St. Quentin) and flows into the North Sea near Vlissingen (Netherlands). The Scheldt estuary constitutes a dynamic environment: it is turbid and well mixed, with a water residence time of about 2 months (Heip, 1988). The tidal amplitude is high, ranging from 3.8 m in the western to 5.2 m in the eastern part, and the estuary is about 100 km long covering an area of $\sim 300 \text{ km}^2$. The total catchment area of the Scheldt river is 22000 km^2 , contains several large industrial areas, and supports a population of approximately 10 million (Soetaert et al., 2006). Compared to other tidal estuaries, the maximum turbidity values in the Scheldt are low and the zone of maximum turbidity less pronounced. The river and its tributaries are a major drain for industrial and domestic waste discharges, which are not all treated in waste water treatment plants. Water quality is poor in the greater part of the river and the eastern part of the estuary (Baeyens et al., 1998). Until the mid 1970s discharges caused an increase in nutrient levels, but after the end of the 1970s nutrient loading decreased and the oxic conditions of the Scheldt estuary improved (Soetaert et al., 2006). In the early 1970s denitrification occurred in the water column, but since 1980 water column denitrification has been reduced and a nitrification front has progressed toward the freshwater section. Water

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column denitrification has not occurred since 1990, and since 2000 most nitrification has occurred in the upstream region (Soetaert et al., 2006). Nevertheless, nitrification is the most pervasive process of the nitrogen cycle in the Scheldt estuary.

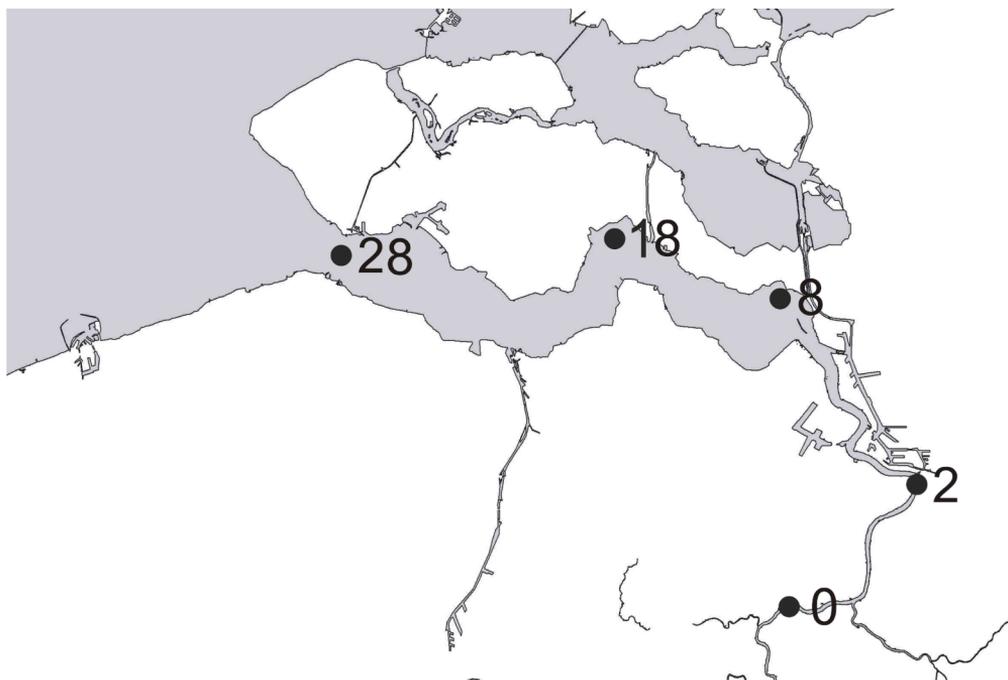


Figure 2.1: Scheldt estuary. Numbers indicate salinity and approximate location of stations

2.2.2 Sampling

During 2003, 4 cruises were conducted with RV 'Luctor' in January, April, July and October, 1 in each of 4 seasons. Five stations were sampled during every cruise. The stations were of fixed salinity but their exact location varied depending on tide and discharge. The salinities of the 5 stations were 0, 2, 8, 18 and 28, and these numbers are used as station names from this point onwards. Station 0 was located close to Dendermonde, Belgium, 122 km from the mouth of the estuary, whereas Station 28 was located at the mouth of the estuary, close to Vlissingen, the Netherlands

(Figure 2.1). Since the stations were situated along a salinity gradient, nutrient and oxygen concentrations and bacterial communities differed among the 5. Water was sampled in 20 L Niskin bottles from approximately 2 m depth and subsampled immediately after retrieval of each bottle.

2.2.3 Concentration measurements

Samples were taken from 17 fixed monitoring stations along a transect through a salinity gradient that ranged from 0 to marine. The water was filtered through preweighed, precombusted Whatman GF/F filters (47 mm), stored frozen, and then analyzed for ammonium, nitrate and nitrite using automated colorimetric techniques. The filters were weighed and analyzed for concentrations of suspended particulate matter (SPM) and particulate organic carbon and nitrogen using a Carlo Erba NA 1500 elemental analyzer (Nieuwenhuize et al., 1994). Salinity, temperature and oxygen were measured at all 17 stations.

2.2.4 Nitrification measurements

¹⁵N method

Water samples were spiked with 98 % ¹⁵N labeled NH₄⁺, at levels that approximated 2.5 % of ambient NH₄⁺ concentration. Samples were incubated at *in situ* temperature in dark bottles in a tank with running estuarine water. Incubations were terminated after 3, 6, 9, 12 and 24 hours by filtration through precombusted Whatman GF/F filters (20 mm). MgO (3 g L⁻¹) and (depending on salinity of the sample) NaCl were added to the water immediately after filtration, to a final salinity of 35. Measurements of δ¹⁵NH₄ and δ¹⁵NNO₃ were based on the diffusion method of Sigman et al. (1997) and Holmes et al. (1998), as modified by Middelburg and Nieuwenhuize (2001) in order to extract the NH₄⁺ from the water. The NH₄⁺ dissolved in the water sample was converted to ammonia gas (NH₃) under alkaline conditions. The NH₃ was then trapped as ammonium sulfate on an acidified precombusted Whatman GF/D 10 mm filter, sandwiched between two 2.5 cm, 10 μm pore size Teflon membranes. After trapping NH₄⁺ in the sample, Devarda's alloy was added to convert all

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the NO_3^- to NH_4^+ , which was then extracted as described above. Since all the original NH_4^+ was removed during the first step, NH_4^+ trapped on the second filter was completely derived from NO_3^- . The ^{15}N content of the GF/D filters was determined using a Fisons NA 1500 elemental analyzer coupled to a Finnigan Delta S massspectrometer via a Conflo II interface. During incubation, the ^{15}N label added as NH_4^+ was oxidized through nitrification and appeared in NO_3^- , see Figure 2.3. Control experiments (to which the nitrification inhibitors N-serve and chlorate [see below] were added) showed no transfer of ^{15}N from the NH_4^+ to NO_3^- pool during 24 h.

When the increase of ^{15}N in NO_3^- was linear (see Figure 2.3A) a linear regression was fitted, but in some cases a curve gave a better fit (See Figure 2.3B, ‘Results’):

$$Y = a_0 - a_1 e^{-kt} \quad (2.1)$$

Results from these regressions were used to calculate nitrification rates (R_N) using the following equation (Dugdale and Goering, 1967):

$$R_N = \frac{b}{(\alpha_{\text{NH}_4^+} - \alpha_n)} [\text{NO}_3^-] \quad (2.2)$$

where b is the slope obtained from the linear regression or the initial slope obtained with the fitted curve (atom % ^{15}N h^{-1}), $\alpha_{\text{NH}_4^+}$ is the initial ^{15}N content in NH_4^+ after addition of ^{15}N , α_n is the natural abundance of ^{15}N in NH_4^+ , and $[\text{NO}_3^-]$ is the average *in situ* concentration of NO_3^- during incubation, which was measured on the filtered water sample as described above. The estimated errors of R_N were obtained from the variance/covariance matrix of the fitted parameters and error propagation. Eq. 2.2 does not take other processes such as uptake or regeneration into account. Uptake of NH_4^+ should not affect the fraction of ^{15}N in NH_4^+ and consequently not influence the nitrification rate. If a high uptake of NO_3^- occurred, the calculated nitrification rates would be underestimated. Parallel measurements of the uptake of NH_4^+ and NO_3^- demonstrated that NH_4^+ is taken up to a much larger extent than NO_3^- . Regeneration of NH_4^+ would dilute the ^{15}N content of the NH_4^+ and cause nitrification rates to be underestimated at high regeneration rates.

¹⁴C method

Carbon incorporation by autotrophic nitrifiers was measured with the inhibitor based ¹⁴C-bicarbonate incorporation technique as described by Brion and Billen (1998). Water samples, taken from the same Niskin bottle as the samples used for the ¹⁵N method, were spiked with ¹⁴C-bicarbonate (50 mCi mmol⁻¹, Amersham) to a final concentration of 5 μmol L⁻¹ and incubated in dark bottles in the tank mentioned above. Carbon incorporation was measured at 3 time intervals over a 9 to 24 hours period. Incorporation due to nitrifier growth was determined using the difference between the amount of H¹⁴CO₃⁻ incorporated in samples with and without nitrification specific inhibitors. N-serve (2-chloro-6-trichloromethyl pyridine; 5 mg mL⁻¹) and sodium chlorate (NaClO₃; 10 mM) were used to inhibit the ammonium and nitrite oxidation, respectively. Nitrification rates (expressed in nMC h⁻¹) were calculated as the slope difference of linear regression lines of C incorporated versus time between samples with and without the inhibitor.

Pilot study

Prior to initiation of the field experiments, a pilot experiment was conducted. In November 2002, water from a fresh water station in the Scheldt estuary was sampled and nitrification rates were measured using the ¹⁵N and ¹⁴C method (as described above) in a laboratory where more extensive sampling was possible than during a cruise. Moreover, incubations with ¹⁵N method were performed under dark and light (approximately 200 μmol photons m⁻² s⁻¹) conditions to assess the light dependence of nitrification.

2.3 Results

2.3.1 Concentration measurements

Concentrations of NH₄⁺ and NO₃⁻ + NO₂⁻ showed a similar pattern in all seasons (Figure 2.2). NH₄⁺ concentrations were highest in the fresh water section of the estuary (≤ 150 μM) and decreased with increasing salinity.

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The decrease was faster during July and October. $\text{NO}_3^- + \text{NO}_2^-$ concentrations in the fresh water section were around $300 \mu\text{M}$, either increased or were uniform until a salinity of ~ 10 , and from there decreased throughout the estuary. The decrease of NH_4^+ and increase of NO_3^- in the upper estuary reflected intensive nitrification. The O_2 concentration varied among seasons and also along the estuary transect. Oxygen concentrations were generally low in the upper part of the estuary, even hypoxic during July and October. Oxygen concentrations increased toward the mouth of the estuary and reached saturation levels and even supersaturation ($415 \mu\text{M}$) in April. The SPM varied from 250 mg L^{-1} in freshwater in October to 10 mg L^{-1} in marine water in January. Water temperature varied with season and was recorded at 4, 10, 20, 14 °C in January, April, July and October, respectively.

2.3.2 Nitrification rates

In most samples, the ^{15}N in NO_3^- showed a linear increase with time (Figure 2.3B), but for some (Figure 2.3A) the increase was non-linear. Non linearity could be explained by substrate limitation, as shown in the comparison of NO_3^- production at Stations 0 and 2. At Station 2 the NH_4^+ concentration at the start of the incubation was $14 \mu\text{M}$ and the nitrification rate was 124 nM h^{-1} , which meant that only 16 % of the initial NH_4^+ was consumed during the 24 h incubation. At Station 0 the initial NH_4^+ concentration was $4 \mu\text{M}$ and the nitrification rate was 621 nM h^{-1} . At this rate, the initial stock of NH_4^+ was consumed within 6 h of the incubation. Regeneration of NH_4^+ during incubation may also have contributed to the non-linear increase of ^{15}N in NO_3^- : in this case, ^{15}N label in the NH_4^+ pool would have become diluted. Although further transfer of ^{15}N from NH_4^+ to NO_3^- could not be detected after 6 h (Figure 2.3A), nitrifier growth and likely nitrification of regenerated NH_4^+ still continued for at least several hours at the same rate (Figure 2.3C). Highest nitrification rates were recorded with both methods in the fresh water region of the estuary and decreased toward the North Sea, similar to the observed pattern of NH_4^+ concentrations (Figure 2.4 and Figure 2.2). With the ^{15}N method, nitrification rates ranged from 700 nM N h^{-1} in October to 150 nM N h^{-1} in April in fresh water, and from 20 nM N h^{-1} to not detectable in the marine water. With the ^{14}C method, rates at Station 0

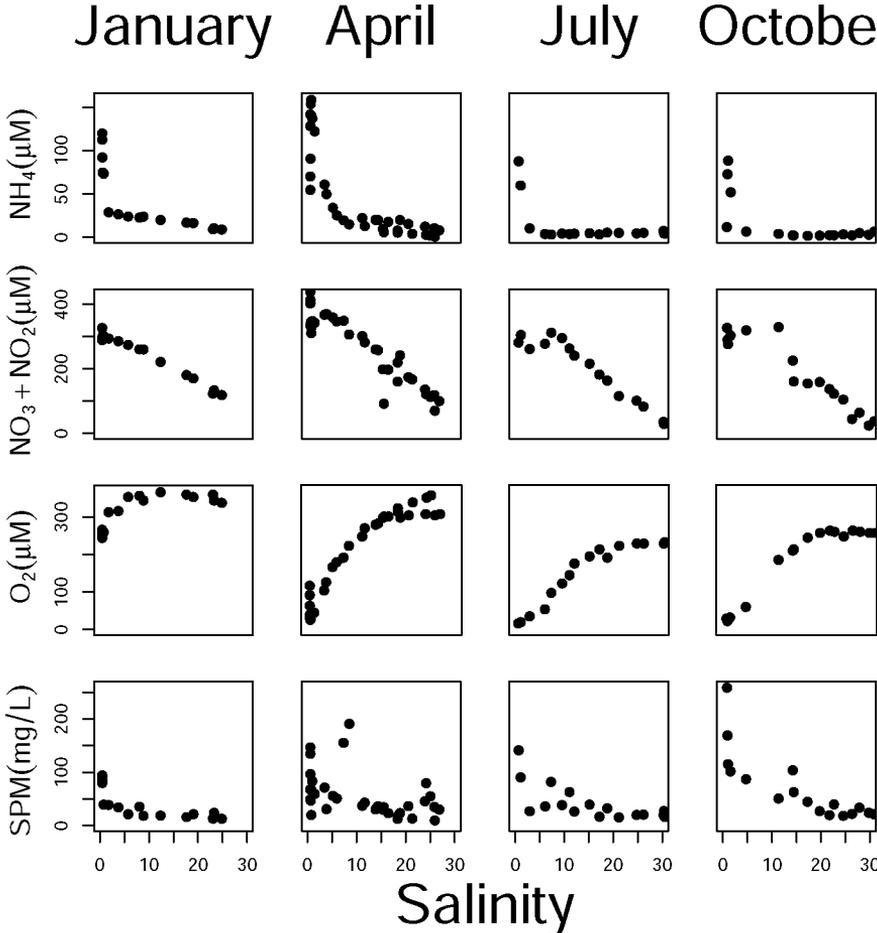


Figure 2.2: Concentrations of NH_4^+ , $\text{NO}_3^- + \text{NO}_2^-$, O_2 and suspended particulate matter (SPM) in the pelagic zone of the Scheldt estuary during 2003.

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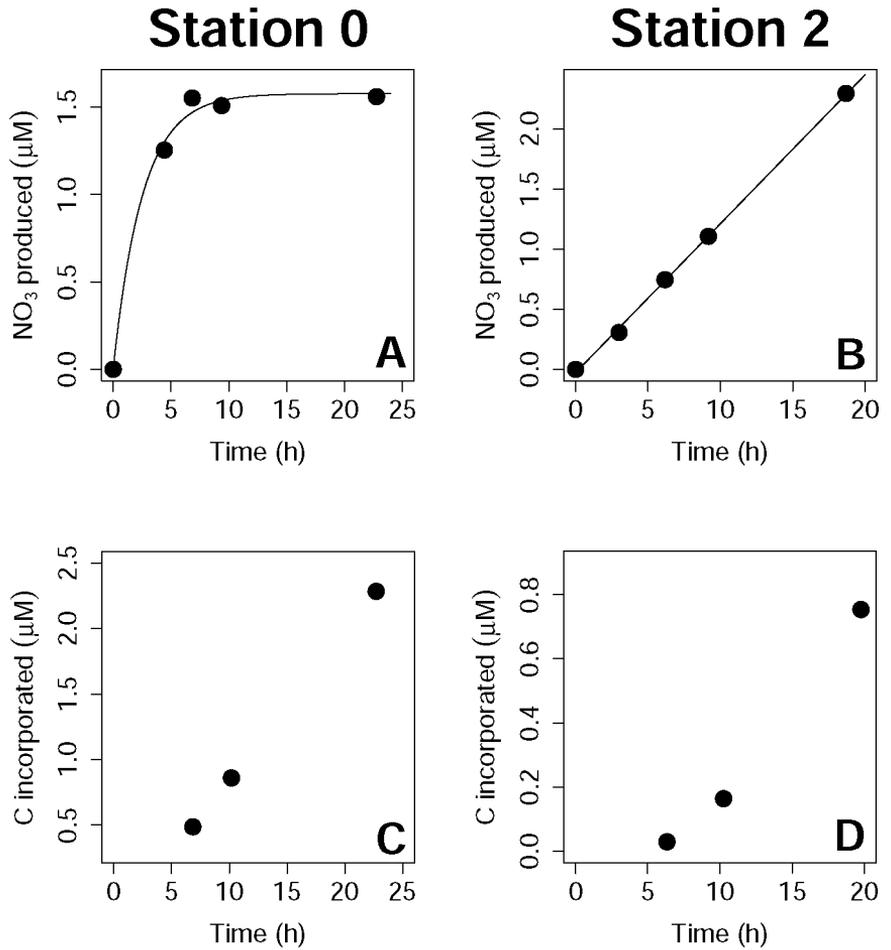


Figure 2.3: (A, B) NO₃ produced and (C, D) carbon incorporated at Stations 0 and 2 in July.

ranged from 150 nM C h^{-1} in October to not detectable in January. Nitrification rates decreased toward Station 28 where values from 1 nM C h^{-1} in January to 10 nM C h^{-1} in October were measured (Table 2.1).

In January, high rates were measured with the ^{15}N method and not detectable or very low rates with the ^{14}C method (Figure 2.4, Table 2.1). In April, the ^{15}N method also showed relatively high activity compared to growth when measured using the ^{14}C method at Station 8. This corresponded to an increase in SPM content at this station during this season (Figure 2.2). The error was larger when a fitted curve was used to calculate nitrification rates (Figure 2.4 and Table 2.1). This observation should be taken into account when interpreting the results.

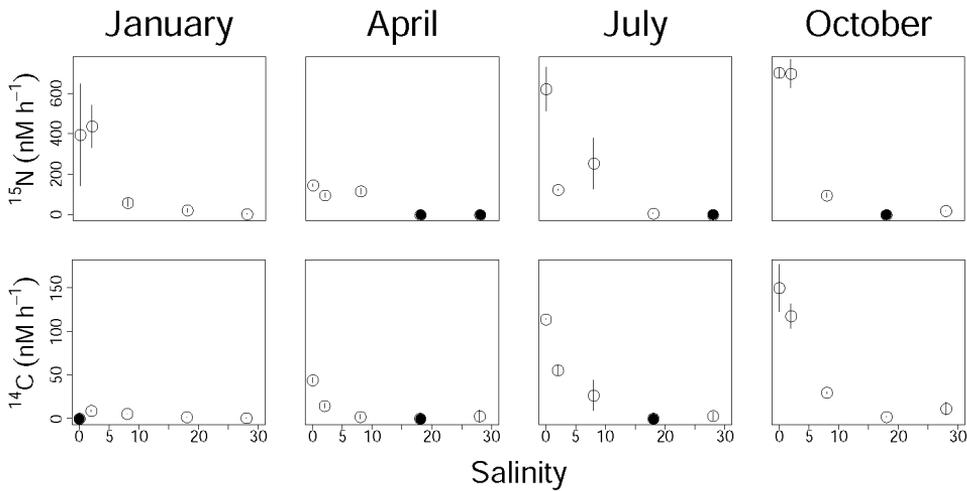


Figure 2.4: Nitrification rates measured with the (top row) ^{15}N method or (bottom row) ^{14}C method during 4 seasons over a salinity gradient. For details of the 2 methods, see ‘Material and methods’. (●) Not detectable; errors are SE calculated from fitted parameters

2.3.3 Pilot study

NO_3^- produced during the light and dark incubations is shown in Figure 2.5A. It appears that nitrification was linear up to 24 hours and is a

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Table 2.1: Comparison of nitrification rates measured with ^{15}N and ^{14}C methods (see ‘Materials and methods’) at 5 salinities (Sal.) during 4 seasons. Curve (C): nitrification rate for ^{15}N method calculated using fitted curve, Line (L): nitrification rate for ^{15}N method calculated using fitted line, nd: not detectable, errors are SE calculated from fitted parameters.

Sal.	Month	^{15}N nM N h $^{-1}$	^{14}C nM C h $^{-1}$	N/C ratio	Curve/ Line
0	January	395.2 \pm 251.0	nd	nd	C
	April	146.6 \pm 5.0	44.1 \pm 3.0	3.3 \pm 0.3	L
	July	620.9 \pm 106.8	114.0 \pm 0.3	5.4 \pm 0.9	C
	October	701.5 \pm 22.1	150.0 \pm 27.0	4.7 \pm 0.9	L
2	January	437.3 \pm 105.2	8.9 \pm 1.2	49.2 \pm 13.5	L
	April	97.2 \pm 10.4	14.6 \pm 2.5	6.7 \pm 1.4	L
	July	124.3 \pm 1.9	55.5 \pm 6.3	2.2 \pm 0.3	L
	October	696.8 \pm 68.7	117.8 \pm 14.2	5.9 \pm 0.9	L
8	January	59.5 \pm 18.8	5.5 \pm 0.1	10.8 \pm 3.4	C
	April	116.7 \pm 14.0	1.9 \pm 2.5	60.2 \pm 77.2	L
	July	254.0 \pm 125.0	26.5 \pm 17.3	9.6 \pm 7.8	C
	October	96.5 \pm 10.5	29.8 \pm 1.0	3.2 \pm 0.4	L
18	January	23.5 \pm 9.3	1.7 \pm 0.2	13.7 \pm 5.8	C
	April	nd	nd	nd	
	July	6.9 \pm 1.8	nd	nd	L
	October	nd	2.0 \pm 0.6	nd	
28	January	4.0 \pm 1.1	0.6 \pm 0.2	6.8 \pm 2.7	L
	April	nd	2.6 \pm 7.2	nd	
	July	nd	2.9 \pm 5.0	nd	
	October	19.7 \pm 0.7	11.3 \pm 7.6	1.7 \pm 1.2	L

light sensitive process. NH_4^+ concentration at the start of the incubation was $102 \mu\text{M}$ and nitrification rates from light and dark incubations were 443 ± 9 and $792 \pm 17 \text{ nM N h}^{-1}$, respectively. Accordingly, 44 % of the nitrification activity was inhibited by the light. Parallel incubations in the dark with the ^{14}C method revealed a rate of $197 \pm 16 \text{ nM C h}^{-1}$ (Figure 2.5B). When we combined ^{14}C and ^{15}N methods for dark incubations we observed that 4.0 ± 0.3 moles of nitrogen were transformed for each mole of carbon fixed.

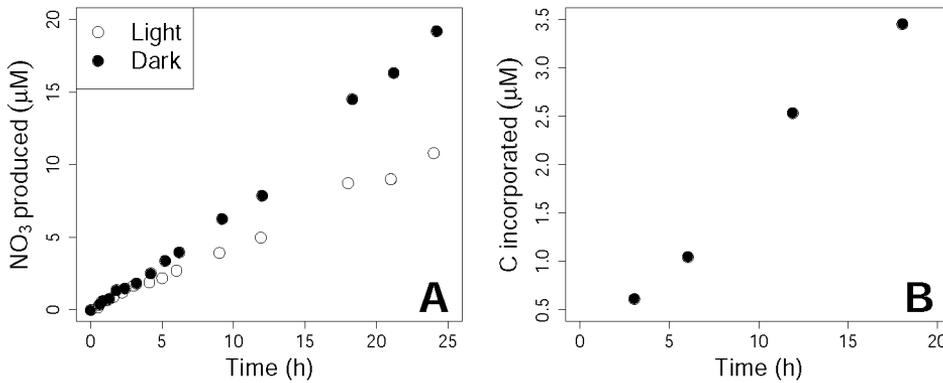


Figure 2.5: (A) NO_3 produced during light and dark incubations, ^{15}N method. (B) Carbon incorporated during dark incubations, ^{14}C method. For details of the 2 methods, see ‘Materials and methods’

2.4 Discussion

Given that nitrification plays a pivotal role in estuarine biogeochemistry, it is essential to have a reliable and accurate technique to quantify the actual activity of ammonium and nitrite oxidizing bacteria. This is especially true when the results are to be used to calculate nitrogen budgets. The ^{14}C method (Somville, 1978) is probably the most common way to measure nitrification and is based on quantifying presence and growth of nitrifying bacteria (Brion and Billen, 1998). This approach is an excellent way to determine whether nitrifying bacteria are growing, but it can not be

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used directly as a quantitative measure of nitrification. Nitrification rates obtained from the ^{14}C method (in carbon units) need to be converted into nitrogen units to be relevant in a nitrogen context. This conversion requires knowledge of how much carbon is fixed by the nitrifier community in order to oxidize a known amount of ammonium to nitrate, and was the subject of extensive research during the 1970 and 1980s (Table 2.2).

Literature values for conversion ratios have often been determined under optimal laboratory growth conditions on pure strains of oxidizing bacteria, e.g. *Nitrosomonas* or *Nitrospira* strains for ammonium oxidation and *Nitrobacter* strains for nitrite oxidation. These studies (Table 2.2) revealed highly variable results, with N:C ratios ranging from 6.0 (Owens, 1986) to 18.9 (Helder and de Vries, 1983). It is questionable whether these ratios are applicable in nature where environmental conditions as well as bacterial population and composition are variable. Factors such as temperature (Bianchi et al., 1997), salinity (Feliatra and Bianchi, 1993), oxygen concentrations (Carlucci and McNally, 1969; Goreau et al., 1980), and substrate availability (Belser, 1984) can affect the N:C ratio. The assumption that slow-growing nitrifiers (Helder and de Vries, 1983) have a conversion factor similar to nitrifiers under optimal growth conditions, in such a heterogeneous and highly variable system as an estuary, is quite possibly erroneous. Considering that even optimal growing populations have variable N:C ratios (Table 2.2), the applicability of a single, constant conversion factor is even more dubious.

Several studies have expressed strong concerns over this issue. For example, one study conducted under optimal growth conditions produced a low N:C ratio, because the bacteria needed little energy for growth under optimal conditions. Therefore, use of this factor provides a minimum estimate of nitrification activity *in situ* (Owens, 1986). The ^{14}C method can be used as a relative index of nitrification, but accurate estimation of the rate of nitrogen oxidation can only be deduced if a constant ratio exists for natural populations of nitrifying bacteria (Billen, 1976). As stated by Hall (1982), "It is unlikely that one ratio could possibly cover a range of environmental conditions and that the absence of reliable data casts doubt on the general applicability of the ^{14}C method as a quantitative measure of nitrification".

Some attempts have been made to quantify the *in situ* N:C ratio through parallel measurements with the ^{14}C method and alternative methods for

Table 2.2: Comparison of N:C ratio values

N:C	Bacterial community	Studied area	Source
8.3	Enrichment cultures	North Sea	Billen (1976)
18.9	Cultures of pure <i>Nitrosomonas</i> and <i>Nitrobacter winogradskyi</i> strains	Ems-Dollard estuary	Helder and de Vries (1983)
9.1	Cultures of <i>Nitrosomonas europaea</i> and <i>Nitrospira</i> sp. strains and <i>Nitrobacter winogradskyi</i> strains	Cultured bacteria	Belser (1984)
6.0	Enrichment cultures	Tamar estuary	Owens (1986)
8.6-9.8	Natural samples	Rhone estuary	Feliatra and Bianchi (1993)
2-60	Natural samples	Scheldt estuary	Present study

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nitrification (Enoksson, 1986; Feliatra and Bianchi, 1993; Dore and Karl, 1996; Bianchi et al., 1997). A ^{15}N labeling approach similar to the one used in this paper was used by Enoksson (1986), but the addition of labeled NH_4^+ increased the concentration far above ambient concentrations. Consequently, potential rates were obtained and this complicated a direct comparison between methods. Nevertheless, the main conclusion was that the ^{15}N method and ^{14}C method should be used in parallel in future studies to determine whether the N:C ratio is constant. When comparing the ^{14}C method with direct measurements of changes in NO_2^- and NO_3^- concentrations in incubated samples, and using an N:C ratio of 8.3 (Billen, 1976), Dore and Karl (1996) found that the 2 independent methods agreed reasonably well in open ocean waters despite exhibiting a high degree of variability. Nitrification has also been assessed with the ^{14}C method and via the change in NO_2^- concentration using specific inhibitors that block the first and the second step of nitrification, respectively (Feliatra and Bianchi, 1993; Bianchi et al., 1997). These studies demonstrated that the N:C ratio varied with salinity, and decreased from the river to the sea (Feliatra and Bianchi, 1993). In addition, a strong negative correlation between the N:C ratio (values ranging between 3 to 9) and temperature was observed in the Indian sector of the Southern Ocean, confirming that the ratio fluctuates with environmental conditions (Bianchi et al., 1997).

Although these studies report variable N:C ratios and concerns about the validity of using 1 fixed ratio in a fluctuating environment, many other studies continue to report nitrification rates in nitrogen units are based on the ^{14}C method and converted with a ratio provided by those studies referred to above: e.g. Indrebo et al. (1979); Joye et al. (1999); Brion et al. (2000), and de Bie et al. (2002b) used the N:C ratio of Billen (1976); Berounsky and Nixon (1990, 1993); Iriarte et al. (1997) and Iriarte et al. (1998) used the N:C ratio of Owens (1986); and Jiang and Bakken (1999b) used the ratio from Belser (1984).

The relationship between nitrifier activity and nitrifier growth observed in our study is shown in Figure 2.6. A constant conversion factor imposes a straight line with a slope corresponding to the quantity of nitrogen transformed per unit carbon fixed. In the Scheldt estuary a conversion factor of 8.3 is often used (Billen, 1976), which is shown as a dashed line in Figure 2.6. We observed large, systematic deviations from the N:C ratio of 8.3 for January and October samples taken in the upper estuary (Fig-

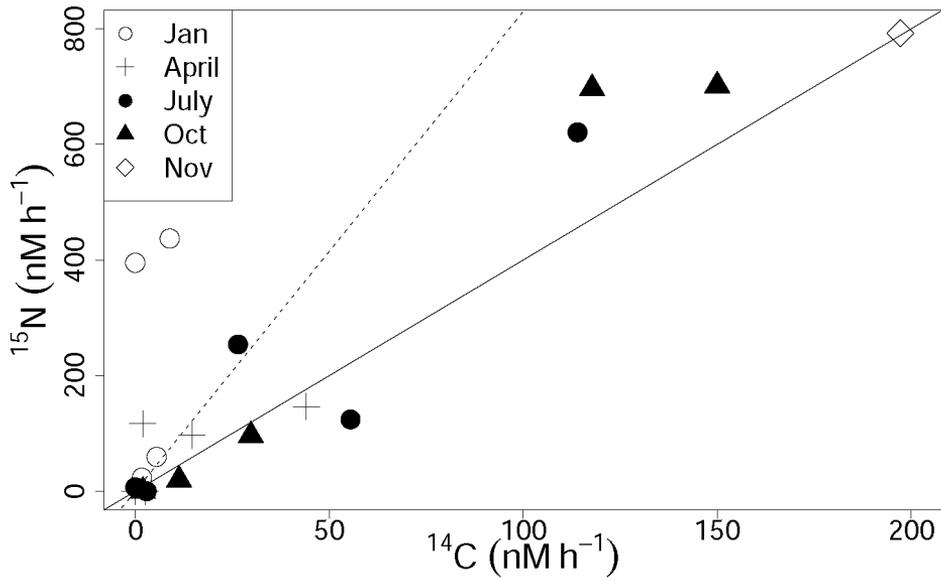


Figure 2.6: ^{15}N method versus ^{14}C method (for details of methods, see ‘Materials and methods’). Dashed line: conversion factor 8.3 (Billen, 1976); solid line: conversion factor of 4.0 (our pilot study), November data: result from pilot study.

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ure 2.6). Rates measured in January showed high nitrification activity and no or very low growth. In January, the water temperature was 4 °C and a high N:C ratio agreed with the strong negative correlation between N:C ratio and temperature observed by Bianchi et al. (1997). Temperature decreases have been shown to induce bacteria to increase their substrate requirement for optimal growth (Wiebe et al., 1992). Accordingly, it may very well be that N:C ratios of nitrifying communities are seasonally variable in temperate systems. Nitrification can efficiently proceed at low temperatures (even < 0 °C) despite the general impression that nitrifiers are inactive at temperatures below 4 to 5 °C (Collos et al., 1988). High activity without growth might also be due to heterotrophic nitrification, which can constitute a significant fraction of ammonium oxidation under favorable conditions (Zhao et al., 1999). This process would not be detected using the ^{14}C method. A number of common denitrifying bacteria have the ability to carry out heterotrophic nitrification (Castignetti and Hollocher, 1984). Heterotrophic nitrifiers occur among algae, fungi and bacteria and, in comparison to autotrophic nitrifiers, rates of heterotrophic nitrification are low, and occur preferentially under conditions not favorable for autotrophic nitrification (Schmidt et al., 2003). Low water temperatures in January could constitute such a condition. Complete heterotrophic growth has been demonstrated for both *Nitrosomonas* spp. and *Nitrobacter* spp. (Hall, 1982, and references therein).

In contrast, October rates based on the ^{14}C method were higher than would be predicted from an N:C ratio of 8.3. Our pilot experiment with tidal freshwater was conducted in November 2002 and produced an N:C ratio of 4.0 (solid line, Figure 2.6), and October data from 2003 appeared to follow this trend. This result implies that bacteria grow faster during these periods than the energy from nitrification at N:C ratio of 8.3 would allow. There are several possible explanations for this: (1) bacteria may use an additional energy source or stored energy reserves, (2) community composition of ammonium-oxidizing bacteria in the estuary may vary over time and/or space, and these different communities may have variable optimal N:C ratios; metabolic activity and growth rates can differ among cultures (Jiang and Bakken, 1999a) and thus also among different communities, (3) our N:C ratios may have been biased towards low values because of our neglect of ammonium regeneration that caused dilution of ^{15}N , and (4) low N:C ratios coincided with low oxygen concentrations i.e.

$< 100 \mu\text{M}$. Higher carbon assimilation per unit nitrogen oxidized at low oxygen concentrations has been observed in several studies (Carlucci and McNally, 1969; Goreau et al., 1980). Our results were consistent with these findings, which demonstrated a significant ($p = 0.007$, $r^2 = 0.64$) positive correlation between N:C ratio and oxygen concentration (Figure 2.7).

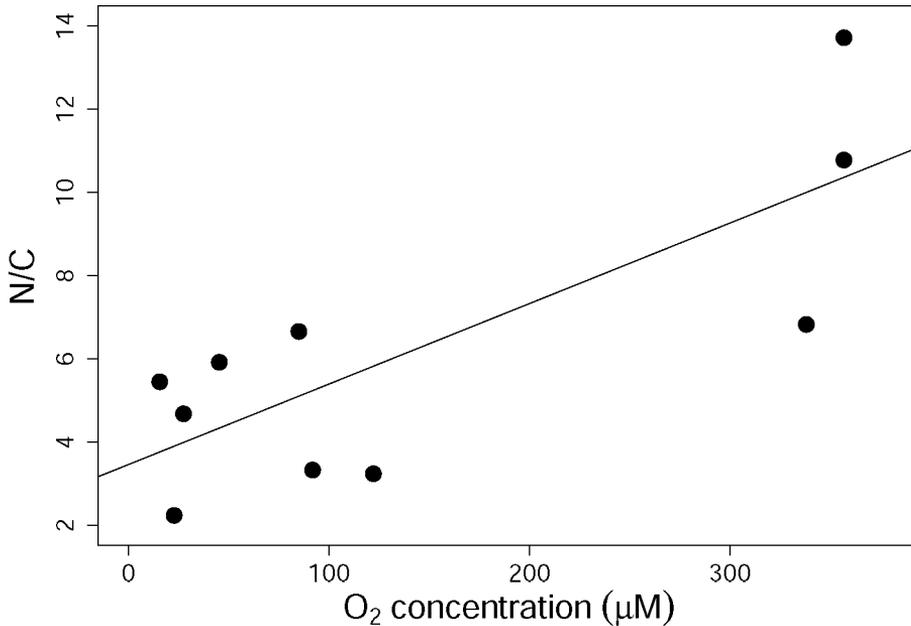


Figure 2.7: N:C ratio < 25 and relative error $< 50 \%$ versus O₂ concentrations

Based on our results and previously published studies, we propose that a clear distinction should be made between nitrification activity expressed in units of nitrogen and nitrifier growth expressed in units of carbon. It is only under optimal steady-state conditions, such as in the open ocean or in a lake with long residence time, that a uniform N:C ratio is to be expected. Accordingly, we propose that the ¹⁵N method is preferable when nitrifier activity is of interest, while the less costly and less labour intensive ¹⁴C method provides a good measure of the growth of chemoautotrophic nitrifying bacteria. In addition to providing a direct measure of

2 Comparison of nitrifier activity versus growth

nitrifying activity in nitrogen units, the ^{15}N method has 2 other advantages over the ^{14}C method: (1) the ^{15}N method is independent of specific inhibitors, and (2) it enables light dependent studies of nitrification. Use of specific inhibitors has been questioned because these may not be 100 % selective, i.e. they may inhibit other processes as well, or because they may not completely block all nitrifying activity (Oremland and Capone, 1988). Not all natural communities of nitrifying bacteria are affected to the same extent by a given inhibitor, and responses to inhibitor concentration may vary among communities (de Bie et al., 2002b). Moreover, several inhibitors are not soluble in water and require organic solvents for application, which may affect nitrification rates. One draw back of the ^{15}N method is that in waters with very low ambient NH_4^+ concentrations, the rates measured will be potential rather than actual *in situ* nitrification rates. We observed that light inhibited more than 40 % of nitrifying activity and, depending on the system studied, it might be highly relevant to measure light dependence. Earlier studies demonstrated an important relationship between nitrification rate and light intensity (Ward, 2005). In cultures of both oceanic and estuarine isolates, NH_4^+ oxidizers in oceanic communities were inhibited by light to a much greater extent than estuarine isolates (Horrigan and Springer, 1990). This result was attributed to high NH_4^+ concentrations and high nitrification rates in estuaries. Our light inhibition of 44 % agreed with the findings of Horrigan and Springer (1990) for 1 of the 3 estuarine isolates studied.

2.5 Conclusions

The present study came to the following conclusions:

(1) A comparison between the ^{14}C and ^{15}N method for nitrification revealed that growth and activity can be uncoupled and consequently that N:C ratios can vary (from 2 to 60 in our study); (2) Oxygen concentration and temperature govern N:C ratios; (3) It is preferential to use ^{15}N techniques over ^{14}C method to measure actual nitrification rates; (4) Nitrification is a light sensitive process: 44 % of nitrifying activity was observed to be inhibited by light in this nitrogen-rich, turbid estuary.