Sampling Scale to assess soil fertility Processes –a brief Review

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Abstract
Soil processes that affect the conditions for plant growth vary in both horizontal and vertical directions. The scale of sampling might make it difficult to quantify the processes.

For mobile macronutrients such as N in the form of NO_3^-, sampling at 0.1- 0.3 m intervals to depths of metres or the base of the root zone is appropriate. However in soils that are undisturbed by vigorous cultivation, rapid variations in soil pH, concentrations of organic C and N, rates of mineralization and nitrification occur with depth through the surface 100 mm. Thus intervals of centimetres are required for studies of the top soil. At the extreme, assessment of environmental changes around plant roots or fertilizer granules, for example urea, requires sampling in millimeters to relate pH changes to processes such as NH_3 volatilization and acid or base excretion by the plant.

Key Words
Nitrate leaching, ammonia volatilization, pH variation, carbon and nitrogen mineralisation

Introduction
Most sampling for the investigation of processes influencing the supply of nutrients to plants involves collection of soil in depth intervals of 100 mm or more. However, many of the soil processes that influence soil chemical fertility, especially the supply of nitrogen (N), are the result of microbial activity that occurs at a scale of micrometres. Despite this realisation there have been few attempts to relate these processes to sampling scales commensurate to variations in soil environmental conditions such as moisture, aeration and pH.

The following discussion provides several examples that reveal the benefits of applying appropriate sampling intervals depending on the processes being investigated.

Carbon and nitrogen mineralisation
Figure 1 shows a rapid decrease in the organic carbon concentration through the surface 200 mm of soil in a Kandosol under cropping when sampled in 25 mm intervals through the surface 100 mm (Purnomo *et al.* 2000b). This trend is consistent with the return and accumulation of organic residues at the soil surface when minimal soil disturbance was used for crop establishment. In addition, the proportion of the organic carbon that is decomposed as measured by CO_2 release also decreased with depth but only to 80 mm below which the proportion increased. The pattern of decomposition mirrored the changes in pH with depth suggesting that pH changes within the surface soil strongly influenced heterotrophic activity. Thus CO_2 evolution was related to variations in organic carbon concentration and pH (Equation 1).

CO_2 evolution (mg C/kg) = 0.036 Organic (%) + 0.212 pH – 1.16 ($r^2 = 0.89$, $P<0.05$) (Equation 1)

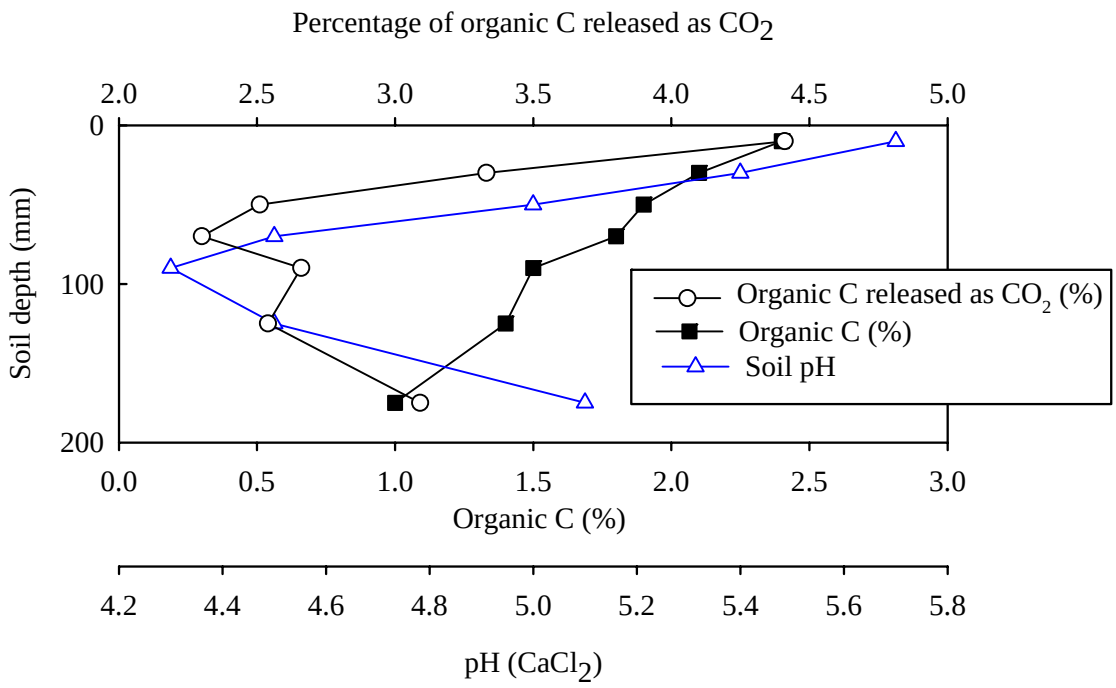

Figure 1. The vertical stratification of carbon mineralisation, organic carbon and pH in a Kandosol at Wagga Wagga, NSW. Adapted from Purnomo *et al.* (2000b).

In this soil net N mineralisation measurement during the growth of a wheat crop showed that 32 and 72 % of mineralisation occurred in the 0 to 20 and 0 to 60 mm depth intervals respectively (Purnomo *et al.* 2000a). Since moisture and temperature regimes vary markedly through the surface 100 mm of soil, it is proposed that to model biological processes involving C and N, sampling at intervals of approximately 20-25 mm is likely to provide improved predictions. However fine scales of 1 – 30 µm, as described by Strong *et al.* (1998, 1999a,b) are not possible in field based research.

Ammonia volatilisation from broadcast urea or urine patches

In other situations, a finer sampling scale is certainly warranted. In urine patches, Ball *et al.* (1979) sampled the soil in depth intervals of 25 mm and the maximum pH observed was 6.5. Since the pK for the NH_3/NH_4^+ equilibrium is 9.2, the observed pH was insufficient for significant volatilisation to occur. However, losses averaged 16% of urine N applied. Subsequently, Sherlock *et al.* (1986) used a sampling device which enabled soil to be collected in 1 mm intervals at distances of 0, 8, 16 and 24 mm from the site of the urea granule placed on a soil. The soil moisture content was near field capacity. Figure 2 shows the resultant pattern of pH. This shows that within a radius of 8 to 10 mm from the granule, the pH was greater than 8 and reached 9 within 3-4 mm, which would favour the formation of NH_3 and hence loss by volatilisation. The measured pH enabled Sherlock *et al.* (1986) to model volatilisation vindicating the measurement of pH in such small intervals. Other studies with urine (Vallis *et al.* 1982) and urea application (Rachhpal-Singh and Nye 1986; Fan and MacKenzie 1993) also showed that pH was between 8 and 8.5 within 10 mm of the soil surface. These examples justify the sampling at the millimetre scale to quantify the pH resulting from urea hydrolysis and enable NH_3 volatilisation to be modelled.

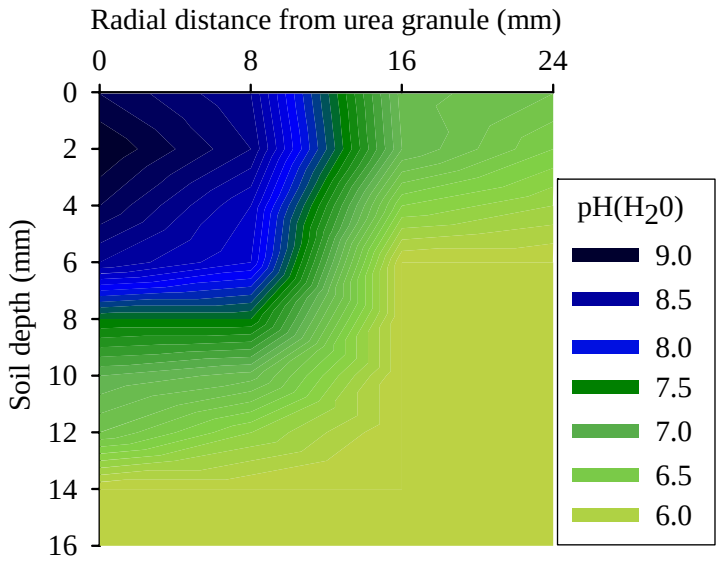

Figure 2. The spatial distribution of pH around a urea granule site 24 hours after application. Redrawn from Sherlock *et al.* (1986).

Development of Acidic Subsurface Layers under Urine Patches

The above example was directed to the scale of a urea granule. Condon *et al.* (2004) sampled at larger depth intervals of 20 mm, to investigate the contribution of various N processes to the formation of acidic subsurface layers. This was fine enough to assess the magnitude, site and rate of N processes such as urea hydrolysis, NH_3 volatilisation, immobilisation, and nitrification. Figure 3 shows the pH change in urine treated, relative to untreated, soil over the experimental period. At the final sampling at about 6 weeks, an acidic subsurface layer had developed confirming that the layers can develop rapidly in these systems. Condon (2002) demonstrated that acidic subsurface layers also formed in urine patches under actively growing plants. This is consistent with observations that these layers could develop after about a month in spring under cereal crops and ungrazed subterranean clover pastures (Paul *et al.* 2003).

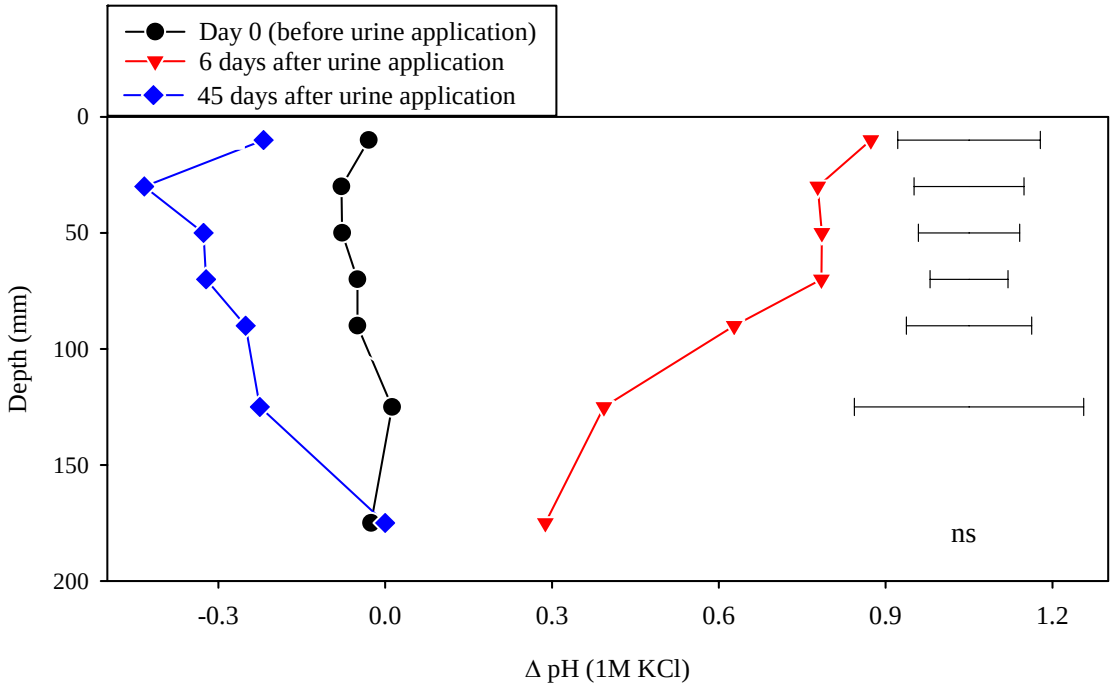

Figure 3. The vertical distribution of difference in pH between a control and a simulated urine solution at various times following application. LSD bars (p<0.05%). Adapted from Condon (2002)

Soil pH variation under and between perennial plants

The examples shown in Figures 1 and 3 demonstrate that acidic subsurface layers can be identified providing that soils are sampled in depth intervals of 20-50 mm. The mechanisms involved in the formation of these layers were reviewed by Paul *et al.* (2003). The presence of plants influences the soil environment, especially pH due to the influence of the balance of cation/anion uptake and the return of organic anions. To study the pH effects, Swan (2000) sampled a vertical soil cross section 150 mm deep from the centre of a cocksfoot plant to the centre of the interplant space. A grid with cell dimensions of 12.5 mm^2 was inserted onto the vertical face of the soil wall. From each cell, an 8 mm diameter core was collected horizontally to 7 mm. The pattern of pH is shown in Figure 4.

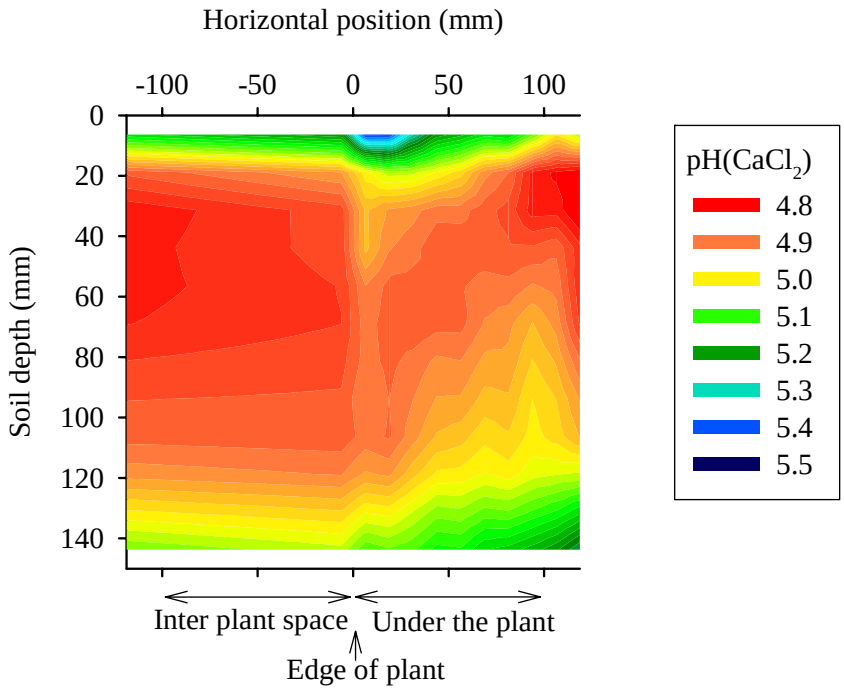

Figure 4. The spatial distribution of pH with depth from the centre of a cocksfoot (*Dactylis glomerata*) plant into the interplant space. Redrawn from Swan (2000).

In the inter plant space, an acidic subsurface layer as seen in Figure 3 was evident between 20 and 100 mm. Immediately beneath the plant, an acidic zone extended from the surface to a depth of about 6 cm which may be attributed to the uptake of an excess of cations over anions that is known to result in the excretion of acidic products into the soil. The remainder of the root zone under the plant was generally more alkaline than in the inter plant space with a more alkaline tongue extending down through the soil at the plant edge. These higher pH values are likely to result from the return of plant residues containing organic anions that will raise the pH following association and oxidation. Sampling at this scale showed the effect of the plant on the soil environment.

The influence of plant root exudates on soil pH requires sampling on a very small scale. Soil pH gradients within a millimetre away from roots have been measured using microelectrodes. The relative magnitude of each of the processes that change pH along roots vary on a very fine scale along the length of growing roots and thus have different influences on rhizosphere pH (Figure 5). If studying the influence roots in the rhizosphere, sampling at the scale of a few millimetres is necessary.

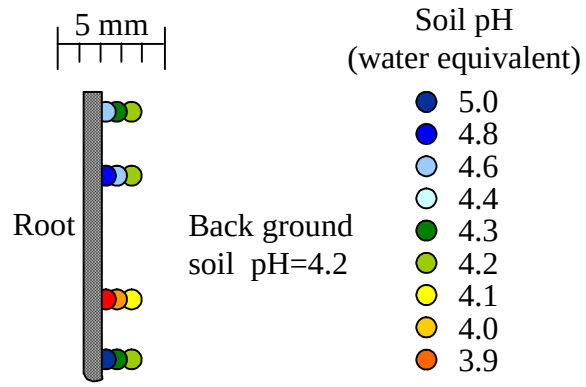

Figure 5. Soil pH along a root and into the rhizosphere of a Norway Spruce tree as measured by microelectrodes. Redrawn from Häussling *et al.* (1985).

Lateral variations in surface soil pH

In Figures 2 and 4, it is evident that there is horizontal variation in soil pH attributable to either the presence of a urea granule or a plant. Figure 6 reports pH changes observed in an ungrazed subterranean clover pasture on a Red Kandosol at Wagga Wagga during March 1993. One centimetre cubes of surface soil from a 100 mm square plot were collected and pH determined. The Figure shows that the pH within the area of 100 by 100 mm ranged from 4.8 to 6.4. This variation could not be related to current or prior processes such as plant location or other factors. In addition, a variation of similar magnitude is observed at a whole paddock scale.

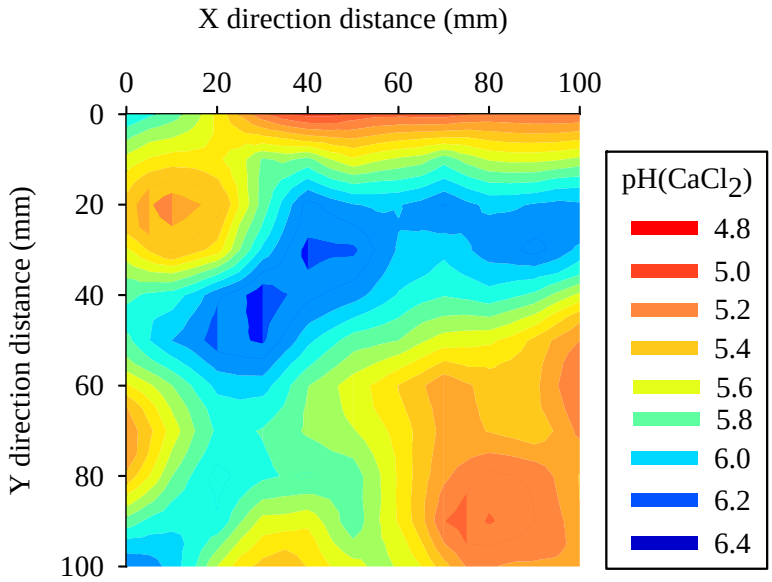

Figure 6. The spatial distribution of pH in 1 cm cubes of top soil of a Kandosol from Wagga Wagga under subterranean clover during the autumn (Poile, Evans and Conyers, unpublished)

Nitrate leaching

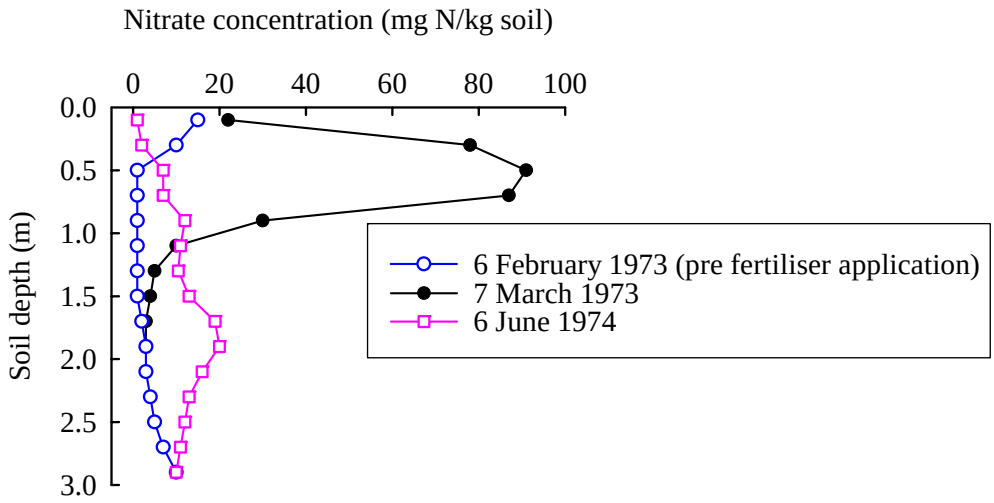

Figure 7. The distribution of nitrate-N in a Ferrosol at various times following the application of ammonium nitrate fertiliser at 160 kg N/ha to French beans (adapted from Black and Waring 1976)

Not all processes require sampling at a scale of millimetres. Where the available nutrient form is highly soluble and not commonly retained by the soil clays, sampling in 0.1 to 0.3 m intervals to the depth of the root zone is appropriate. In these circumstances, where environmental consequences of nutrient loss by leaching need to be considered, sampling well below the root zone of the vegetation is necessary. The nutrient most commonly meeting these criteria is N in the form of nitrate (NO_3^-) and to a lesser extent, S in the form of sulphate (SO_4^{2-}). Figure 7 shows an example of deep sampling to quantify the distribution of NO_3^- within and below the root zone in a highly permeable Ferrosol in south eastern Queensland.

Figure 7 shows that under the summer rainfall conditions, nitrate leached rapidly within one month of application to a French bean crop. Where a fallow treatment was imposed following the bean crop, some of the residual nitrate was leached to depths below 2.5 m with the maximum accumulation at a depth of 1.8 m. At these depths the nitrate was well beyond the depth of rooting of most crops and was a pollution hazard to underground water reserves. In this example, the sampling increment was 0.2 m and was appropriate to assess the distribution of nitrate.

Conclusions

The variation in a range of soil properties and processes can only be shown using suitable scales of sampling. The traditional sampling regime of 0.1 – 0.2 m is appropriate for certain studies especially those involving mobile nutrients. However to be able to quantitatively relate microbial processes to soil properties sampling in smaller intervals is more likely to be successful. While these processes occur at the µm scale, under field conditions the use of sampling intervals of 1 to 25 mm has been shown to relate soil properties to the processes.

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