CHAPTER 8-4
NUTRIENT RELATIONS: UPTAKE AND LOCATION

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Figure 1. Drops of "steam" from geothermal vents resting on the wire-like leaves of *Campylopus holomitrius* and providing a source of nutrients and a means of trapping airborne nutrients. Photo by Janice Glime.

**Uptake**

The role of bryophytes in nutrient uptake within ecosystems is generally ignored because of their small stature. Weetman and Timmer (1967) showed that the common feather moss *Pleurozium schreberi* (Figure 2) in a black spruce (*Picea mariana*; Figure 3) forest took up only 23-53% of N, P, K, and Mg taken up by trees. Nevertheless, that is a non-trivial figure. But bryophyte contributions to sequestering nutrients can be substantial. Yet we have little concept of whether their net contribution is beneficial or detrimental in those ecosystems where they abound.

Figure 2. *Pleurozium schreberi*, a common feather moss in black spruce forests. Photo by Michael Lüth, with permission.

Figure 3. *Picea mariana* forest in Northern Alberta, Canada, with *Pleurozium schreberi* and *Hylocomium splendens*. Photo by Richard Caners, with permission.

**General Considerations**

**Sources**

First we need to recall that the sources of nutrients for bryophytes include precipitation, dust, and to a limited extent, substrate. Brown (1982) explains that bryophytes absorb mineral nutrients over their entire surface (Figure 1). This ability is promoted by two characteristics of bryophytes: a large surface area to volume ratio, and a low surface resistance, relative to *tracheophytes* (lignified
vascular plants), due to the limited development of cuticle. This is further enhanced within the bryophyte by typically having leaves of only one cell layer in thickness, hence exposing every leaf cell directly to the nutrient supply immediately. Based on what we know thus far, actual entry into the cell is most likely similar to that of tracheophytes.

**Site of Uptake**

Their typical differences in site of uptake would seemingly remove the bryophytes from competition with tracheophytes for soil nutrients. Due to lack of vessels and tracheids, we have assumed that uptake of nutrients by bryophytes is primarily through their leaves. Even in the endohydric *Polytrichum* (Figure 4), water entry is gained primarily at the tips of the plants by water that has travelled upward through external capillary spaces (Trachtenberg & Zamski 1979). Hence, we can expect that most nutrient entry is not through rhizoids, but through leaves, and at least in some mosses may be greater at the tips than in lower parts of the plant. Brown and Wells (1990) remind us that despite their small size, the bryophytes have intricate surface areas that are effective at trapping both dust and moisture that can subsequently enter the bryophyte (Figure 1). The ratio of surface area to volume in bryophytes is enormous compared to that of trees and other tracheophytes.

**Rhizoids**

Even if bryophytes were to use their rhizoids to gather some nutrients, the soil penetration by these structures is generally shallow and well above the zone occupied by most fine roots of tracheophytes, especially trees. Instead, we have assumed that bryophytes typically rely largely on dust on their surfaces and on nutrients dissolved in rainfall. In forests, these arrive primarily through leachates acquired in canopy throughfall. This is a quite different strategy from that of tracheophytes, although in *Polytrichum commune* (Figure 4) it does appear that some nutrients might enter through the rhizoids (Chapin *et al*. 1987). On the other hand, *P. commune* and other forest floor mosses in the black spruce forest (Figure 3) lose nutrients to the black spruce fine roots through mycorrhizae (fungal associates).

**Growth Form**

Growth form affects nutrient trapping and subsequent uptake. Taylor and Witherspoon (1972) found that *Dicranum* (Figure 5), which grows in a relatively tight clump, retains more particles than do open lichens such as *Cladonia/Cladina* (Figure 6), even though these lichens display considerable surface area. Hence, we should expect such tight cushions to be more effective at trapping than more open bryophytes like *Brachythecium* (Figure 7) or *Mnium* (Figure 8). On the other hand, Shacklette (1965) found that bryophytes were significantly contaminated with soil particles, including insoluble ones such as Al, Be, Fe, Si, and Zr. But, it would appear that even deeper soil is not immune to moss nutrient scavenging, perhaps through a combination of capillary action and concentration gradient.
Nutrient Relations: Uptake and Location

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Age

Bryophyte uptake can relate to age. In studying the Alaskan black spruce (*Picea mariana*) forest (Figure 3), Chapin *et al.* (1987) found that in three of the moss taxa studied, the phosphate absorption capacity increases with age of green tissue, but decreases with age of brown tissue.

In the aquatic moss *Warnstorfia fluitans* (Figure 9), an acidophile (preferring acid habitats), iron (Fe) accumulates in the cell wall (Satake 2000). The highest concentrations are in the base, increasing toward the tip. In addition to the biological accumulation within the walls, iron is held on the mosses in crystal form.

Nutrient Concentration

Lou *et al.* (2013) found that the content of the heavy metals Pb, Cr, and Cu in the moss *Haplocladium microphyllum* (Figure 10) correlated with the concentrations in the medium. Iron (Fe), on the other hand, increased in a similar manner until the concentration in the medium reached 400 mg L⁻¹ as Fe⁺⁺. Below that level the iron facilitated uptake of other nutrient ions. The absorption capacity for these metals follows the order Fe>Cr>Cu>Pb. Like most things in nutrient relationships, the amount matters. At low concentrations, both lead and copper ions, as with iron, promote the absorptive capacity of other nutrient elements. At high concentrations the same metals decrease uptake of other nutrient elements. Chromium is an exception, inhibiting absorption capacity of the nutrients P, K, Ca, S, Fe, and Cu even when the Cr concentrations are low. Lou and coworkers found that Pb and Cr are stored primarily in the peripheral cortex of the moss stem in *Haplocladium microphyllum*. It is not clear how this affects uptake of other ions.

Water Source

The standing or flowing water habitat of *Sphagnum* fen (Figure 11) species contrasts sharply with the rainfall source of many other bryophytes. Although species occupying raised bogs with no ground water input may rely almost entirely on rainfall, those mosses in fen situations undoubtedly get nutrients from the ground water as well. In a study of 21 species of *Sphagnum* (Figure 21-Figure 29) in Poland, this genus demonstrated its ability to accumulate N, P, and K in the upper parts of the plant through active uptake, whereas Ca, Mg, and Na accumulated through passive cation exchange (Wojtun 1994; see below), suggesting an arrangement of nutrients within the plant similar to that of the tracheophytes.
A number of researchers have concluded that cryptogamic crusts (soil crusts of algae, Cyanobacteria, fungi, lichens, and bryophytes) that live on the soil in areas with low rainfall increase the availability of essential elements, such as N, Cu, K, Mg, and Zn, thus benefitting seeds, seedlings, and mature tracheophyte plants (Harper & Pendleton 1993; Belnap & Harper 1995; Harper & Belnap 2001). This is most likely due to a combination of trapping airborne nutrients and preventing loss due to erosion and leaching from the soil. We are only beginning to understand the extent and role of bryophytes in nutrient trapping, sequestration, and release in various habitats.

Cation Exchange

Once we understand external transport, we must examine how the nutrients actually enter the moss. Are all nutrients equally capable of entry? Most likely not, but how is that controlled? And can these bryophyte leaves function as well as roots of tracheophytes in the absorption of nutrients?

Brown and Buck (1985) considered the cation exchange capacities (CEC; see below) of bryophyte cell walls to be important in their uptake and sequestering ability. Potassium (K) can be held on exchange sites, then remain in solution once it enters the plant. These researchers warned that it was important to know the locations of minerals within and on the bryophytes because ions such as those of Ca and Pb can remain on exterior exchange sites whereas Mg and Zn can be both internal and external.

Dainty and Richter (1993) identified two classes of weak-acid binding sites. One had a low pK (2-4) and the other a high one (>5). pK is the pH at which equal concentrations of acidic and basic forms of a substance are present; it is the negative log₁₀ of the dissociation constant of the electrolyte. The binding sites are related to the uronic, amino, and phenolic acid contents of the cell walls. Dainty and Richter concluded that "valence-dependent reductions in cation activities in the wall phase are an important contributor to the differences in the pK estimates."

The ability of bryophytes to take up nutrients from weak solutions (Babb & Whitfield 1977) permits them to grow in situations that may be limiting to tracheophytes. We know that many (perhaps all) bryophyte sequiter nutrients on exchange sites (Clymo 1964; Craigie & Maass 1966; Wells & Brown 1990; Bates 1997), but that the exchange capacity varies among species (Büscher et al. 1983).

Polyuronic Acids and CEC

In bryophytes, cation exchange is the process in which positively charged ions in the environment are able to replace H⁺ ions at the surface of the cell walls, particularly those of leaves. Cation exchange capacity (CEC) is due to high concentrations of non-esterified pectates, mostly polyuronic acids, within the cell walls (Clymo, 1963; Craigie & Maass, 1966) and seems to be the first step in uptake of nutrient cations (Koedam & Büscher 1983). Fine roots of tracheophytes use this method as the first step in obtaining cationic nutrients from their surroundings. Koedam and Büscher (1983) demonstrated that CEC in mosses, typically much higher than in tracheophyte roots (Table 1; Knight et al. 1961), was related to soil preference and carbonate content of the bryophytes.

<table>
<thead>
<tr>
<th>Cation Exchange Capacity (CEC)</th>
<th>µg g⁻¹ dry mass</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Calcicolous bryophytes</strong></td>
<td></td>
</tr>
<tr>
<td>Ctenidium molluscum</td>
<td>15,510</td>
</tr>
<tr>
<td>Tortella tortuosa</td>
<td>15,160</td>
</tr>
<tr>
<td>Schistidium apocarpum</td>
<td>12,940</td>
</tr>
<tr>
<td>Homalotheicum sericeum</td>
<td>12,460</td>
</tr>
<tr>
<td>Orthotrichum cupulatum</td>
<td>12,250</td>
</tr>
<tr>
<td>Syntrichia ruralis</td>
<td>10,160</td>
</tr>
<tr>
<td><strong>Calcifugous bryophytes</strong></td>
<td></td>
</tr>
<tr>
<td>Ptychomitrium polypodium</td>
<td>6,690</td>
</tr>
<tr>
<td>Racomitrium fasciculare</td>
<td>3,330</td>
</tr>
<tr>
<td>Dicranoweisia cirrata</td>
<td>3,200</td>
</tr>
<tr>
<td>Andreaea rothii</td>
<td>2,660</td>
</tr>
<tr>
<td>Grimma donniana</td>
<td>2,610</td>
</tr>
<tr>
<td>Racomitrium lanuginosum</td>
<td>2,330</td>
</tr>
<tr>
<td><strong>Tracheophytes</strong></td>
<td></td>
</tr>
<tr>
<td>0-5 mm</td>
<td></td>
</tr>
<tr>
<td>field bean</td>
<td>491.0</td>
</tr>
<tr>
<td>yellow lupine</td>
<td>422.0</td>
</tr>
<tr>
<td>barley</td>
<td>106.8</td>
</tr>
<tr>
<td>rye</td>
<td>63.1</td>
</tr>
<tr>
<td>5-20 mm</td>
<td></td>
</tr>
<tr>
<td>field bean</td>
<td>543.7</td>
</tr>
<tr>
<td>yellow lupine</td>
<td>527.4</td>
</tr>
<tr>
<td>barley</td>
<td>59.1</td>
</tr>
<tr>
<td>rye</td>
<td>65.5</td>
</tr>
</tbody>
</table>

The uronic acids are important in creating cation exchange sites. Popper and Fry (2003) have demonstrated that bryophytes (including hornworts, thalloid and leafy liverworts, and basal mosses) have higher concentrations of glucuronic acid in their primary cell walls than any of the other land plants. Basal mosses have higher concentrations than more advanced mosses, and the highest occurs in Sphagnum (Figure 21-Figure 29). Anthoceros (Figure 102-Figure 103) was unique in having a repeat-unit of glucuronic acid-α(1→3)-galactose, a substance nearly lacking in other kinds of plants in the study. Galacturonic acid is known as a subunit in some xyloligulans, a group of hemicellulose cell wall compounds (Peña et al. 2012). In particular, Peña et al. (2008) found that mosses and liverworts have xyloligulans that contain galacturonic acid, making them distinctly different from those xyloligulans demonstrated in both hornworts and tracheophytes. Popper and Fry (2003) considered that the cell wall xyloligulans may have been pre-adaptive substances that permitted early colonization of land, permitting rapid acquisition of nutrients during periods of short-lived surface water availability.

The role of cation exchange in nutrient uptake in poor nutrient habitats is further supported by the greater ability of Sphagnum (Figure 21-Figure 29) to exchange Ca⁺⁺ and Mg⁺⁺ ions for H⁺ ions, providing them with a mechanism to obtain the very limited nutrients in their habitats. For example, Temple et al. (1981) reported the exchange capacity of Sphagnum to range 0.9 to 1.5 meq per gram dry biomass, whereas that of other mosses generally ranges 0.6-1.1. Figures in meq on tracheophytes were hard to
find; I was able to find that wheat (Triticum vulgare) has a low CEC of 0.02 meq per gram dry biomass of roots, with the highest in that study of 0.2 meq in cress (Lepidium sativum) (Wiersum & Bakuma 1959).

On the other hand, if the Ca ++ content of the habitat is too high, Sphagnum will bind so much Ca ++ to its leaf surfaces that it will eventually kill the moss (personal observation). Although this cation exchange process is beneficial in obtaining nutrients, it can also result in accumulation of high levels of heavy metal pollutants (Brown 1984) such as Cd because the moss lacks sufficient selectivity in either binding or uptake of these non-nutrients (Brown & Bates 1990).

**The Mechanism**

As early as 1961, Knight et al. found a correlation between uronic acid contents and cation exchange capacity. Sphagnum (Figure 21–Figure 29), in particular, has extensive binding sites through its use of the polyuronic acid known as galacturonic acid (Clymo 1963). Through this capability, Sphagnum is able to outcompete tracheophytes. By creating an "intense nutrient impoverishment" for other plants, Sphagnum gains a competitive edge (Van Breemen 1995). It can impede growth of peatland shrubs such as leatherleaf (Chamaedaphne calyculata; Figure 12) (Bartsch 1994) by sequestering nutrients the shrubs need for growth.

Polyuronic acids such as galacturonic acid have a carboxyl group (COOH+) protruding on the outer surface of the cell wall. This carboxyl group freely exchanges its H + for other cations in its surroundings (Figure 13). Hence, when cations such as K +, Mg ++, and Ca ++ filter through the bryophyte layer, these ions are often bound on these bryophyte cell wall exchange sites.

Seemingly all bryophytes have a large number of exposed exchange sites, compared to those even of roots of tracheophytes (Knight et al. 1961). These exchange sites are essential to the uptake of nutrients in non-Sphagnum bryophyte taxa as well. For example, Pseudoscleropodium purum (Figure 14) ceased absorbing Mg ++ and lost intracellular Mg when the exchange sites were saturated with CaCl₂, suggesting adherence to exchange sites may be a necessary prerequisite to Mg ++ uptake (Bates 1989). Addition of both K + and Ca ++ greatly increased their concentrations in the exchangeable fraction of the cell but significantly reduced the concentration of Mg ++. Malmer et al. (1992) found that the concentrations of Mg ++ and Ca ++ in Canadian mire species [three Sphagnum species and Tomentypnum nitens (Figure 15), all from hummocks] correlated with the surface water concentrations. It is interesting that when Ca ++ is increased, the brown mosses are more competitive than are Sphagnum species. And there is evidence that brown mosses as well as Sphagnum can lower the pH, but that they typically do it at a higher level of pH (Figure 16) (Glime et al. 1982).
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Figure 16. Comparison of pH-lowering ability of three mosses from an alkaline fen, Lawrence Lake, Barry County, Michigan, USA, following 48-hour incubation. Co = control lake water, Dr = *Drepanoclados revolvens* (Scorpidium revolvens; Figure 17), Cs = *Campylium stellatum* (Figure 18), Sr = *Sphagnum russowii* (Figure 41). 12 hours dark and light indicate last cycle completed. Vertical lines represent 95% confidence intervals. Horizontal lines indicate no significant differences among green (active) (——) and brown (senescent) (• • •) moss species (distribution-free multiple comparisons test, α= 0.05; n = 10). Starting pH = 8.25. From Glime et al. 1982.

Figure 17. *Drepanoclados revolvens*, an alkaline fen moss that lowers the pH of its medium. Photo by Michael Lüth, with permission.

Figure 18. *Campylium stellatum*, alkaline fen moss that lowers pH of surroundings. Photo by Michael Lüth, with permission.

Cation Competition

So how does competition between cations happen? Divalent cations such as Ca ++ and Mg ++ require two binding sites. When there are many of these ions in the environment, they compete for binding sites, occupying two of them in paired sites. Other ions that require two sites then have more difficulty finding the pair of sites they need to bind.

Because plants have a finite number of exchange sites, ions must compete with each other for those locations. Thus, if one cation is in excess, it can cause cellular deficiency of other cations that are unable to gain access to these exchange sites. Based on their experiments with *Hylocomium splendens* (Figure 19) and *Sphagnum*, using artificial precipitation, Gjengedal and Steinnes (1990) considered that cations such as Na + and Mg ++ in the precipitation may occupy exchange sites and affect the uptake of other ions by this competition. They found that uptake of Zn and Cd were pH dependent and that increasing temperatures increased the uptake for all four of the metals tested (Ca, Cu, Pb, Zn).

Figure 19. *Hylocomium splendens*, a species in which cations such as Na + and Mg ++ in the precipitation may occupy exchange sites and affect uptake of nutrient cations. Photo by Michael Lüth, with permission.

Figure 20. *Calliergonella cuspidata*, a species whose growth is inhibited at high Ca concentrations. Photo by Michael Lüth, with permission.

Complexing reactions with anions such as Cl − may also interfere with uptake. When Bates and Farmer (1990) applied CaCl2 to three bryophytes, their responses varied by habitat. *Pseudoscleropodium purum* (Figure 14) and...
Calliergonella cuspidatum (Figure 20) from chalk soil exhibited significantly reduced growth at high Ca concentrations (5 mol CaCl₂ m⁻³), whereas P. purum and Pleurozium schreberi (Figure 2) from acidic clay were unaffected by the additions. The mosses from the chalk soil had lower initial tissue levels of K and Mg, suggesting that the additional CaCl₂ caused deficiencies in these nutrients through exchange site competition.

Ions in the external solution will first establish equilibrium with the exchange sites (Brown 1982). This physical process is completed very rapidly in the lab, but may require days in the field (Brown & Bates 1990). Once that is established, the remaining ions are available for uptake to the interior of cells (Pickering & Puia 1969). Hence, high concentrations of minerals will ultimately increase the uptake.

The number of exchange sites seems to be adaptive, at least in Sphagnum. Sphagnum section Acutifolia (Figure 21), which inhabits drier locations, has more exchange sites per unit of biomass than do members of section Cuspidatum (Figure 22), which are wet hollow species (Brown 1982). Both Clymo (1963) and Spearing (1972) showed that the number of exchange sites correlated positively with height above water of the optimum habitat for Sphagnum species. This permits hummock species to hold nutrients on their cell surfaces until they are needed without having to wait for rainfall to provide a new source.

Monovalent ions have little effect on CEC for divalent ions (Brehm 1968). But CEC of monovalent cations drops to 0.025 - 0.14 times capacity when in company of divalent cations, presumably due to double binding of divalent ions, much like doubling the strength of a magnet.

Brehm found that dead and living material have the same CEC on a dry weight basis. Nevertheless, living Sphagnum (Figure 21-Figure 29) cells contain most of the K⁺ and Na⁺, Ca⁺⁺ is mostly on the external exchange sites, and Mg⁺⁺ is on both locations. On the other hand, branches and stems of Sphagnum have very different CEC. The living Sphagnum is able to maintain a relatively constant cellular content of cations, even when the concentrations of the medium varies widely.

The ability of an exchange site to hold a given positively charged ion depends not only on the valence (charge) of the ion, but also on concentration. When there is a flood of H⁺ ions, these will replace the other, more rare and higher mass cations. Again, this is like a magnet; it is harder for a magnet to hold something heavy than something light (like H⁺). Hence, basic cations from the bryophyte surface are released into the soil (Foth & Ellis 1997). A striking example of this phenomenon is the case of acid rain making a Sphagnum (Figure 21-Figure 29) peatland alkaline and causing the Sphagnum to die! (Kilham 1982). The acid rain caused the release of alkaline positive ions from the surrounding hillside, which ultimately washed into the peatland. Although Sphagnum is equipped to bind such ions and make its surroundings more acid, it was not equipped to handle the large concentration that resulted from the uphill release. Instead, cations such as Mg⁺⁺ and Ca⁺⁺ accumulated on the surface of Sphagnum and eventually killed it. In forested ecosystems, cations released from soil exchange sites become available to roots, may be leached from the organic layer into deeper layers, or may be lost through runoff.

Heavy Metal Relationships

Several bits of information suggest that heavy metals like cadmium use cation exchange of low valence ions to aid their uptake. For example, in the thallose liverwort Dumortiera hirsuta (Figure 23), pretreatment with 80 mM KNO₃ causes higher Cd⁺⁺ uptake, suggesting that potassium is able to strip exchange sites of competing cations, allowing higher valence cadmium to then strip some of those sites and enter cells (Mautsoe & Beckett 1996).
The heavy metals, in turn, influence uptake of potassium and magnesium – two essential nutrients (Carballeira et al. 1999). The researchers suggested that loss of K, a very soluble nutrient, from the cells might be due to the effect of the heavy metals on the cell membranes, changing their permeability. The cation Mg ++ was most likely displaced from the cation binding sites by the heavy metals.

In one experiment, three aquatic bryophytes were exposed for 60 minutes to solutions of 0, 1, 10, 50, 100, and 200 ppm each of Cd, Co, Cu, Ni, Pb, and Zn (Carballeira et al. 1999). Locations of these metals plus K and Mg were determined. Most metals remained on extracellular locations, rather than intracellular. On the other hand, only negligible amounts appeared in the particulate fraction. The relationship between water concentration and extracellular concentration could be modelled with a Michaelis-Menten equation:

\[
\frac{d[P]}{dt} = \frac{V_{max} [S]}{K_M + [S]}
\]

\(V\) = reaction rate  
\([S]\) = concentration of substrate S  
\(P\) = product  
\(t\) = time  
\(V_{max}\) = max rate achieved at saturating substrate concentration  
\(K_M\) = substrate concentration at which reaction rate is half of \(V_{max}\)  
\(d [P]/dt\) = change in product per change in time

But the three aquatic species in this study by Carballeira et al. (1999) differed markedly. In *Scapania undulata* (Figure 24-Figure 25), the extracellular cation-binding sites demonstrated high metal affinity. On the other hand, *Fissidens polyphyllus* (Figure 26) has relatively low affinity. Nevertheless, *F. polyphyllus* had the highest internal concentrations of these metals at the end of the experiment. The uptake priorities were the same for all three species.

In these aquatic bryophytes, *Fontinalis antipyretica* (Figure 27), *Scapania undulata* (Figure 24-Figure 25), and *Fissidens polyphyllus* (Figure 26), the extracellular compartment held more metals than did the intracellular compartment (Vázquez Castro et al. 1999). The extracellular cation-binding sites of *S. undulata* had a high metal affinity, whereas it was relatively low in *F. polyphyllus*. On the other hand, *F. polyphyllus* after the incubation in the metal solutions had the highest intracellular metal contents. All three species had the same ranking of metal uptake.

Uptake of heavy metals in these aquatic bryophytes led to considerable losses of intracellular K (probably due to effects on plasma membrane properties) (Carballeira et al. 1999). Similarly, Mg ++ cellular contents decreased, but it was apparently due to competition by the metals on the binding sites, limiting uptake. Species differences were again interesting. *Scapania undulata* (Figure 24-Figure 25) exhibited the highest losses of K from internal cell sites, followed by *Fontinalis antipyretica* (Figure 27). On the other hand, *S. undulata* had the lowest losses of Mg from its extracellular exchange sites. These experiments help to explain competition among nutrients and locations in the short term, but long-term effects could be different, as seen in Chapter 8-3 on nitrogen.
Much of what we know about uptake of minerals into plants comes from studies on these heavy metal pollutants. Cadmium, a common pollutant in areas with agricultural fertilizers and other human uses, moves from extracellular sites of the bryophytes to intracellular sites. In *Rhytidiadelphus squarrosus* (Figure 28) cadmium altered photosynthetic rates (Wells & Brown 1987). Its activity at the plasmalemma (cell membrane) may exercise control over other ions, affecting their accumulation within the cell, and *vice versa*.

**Differing Affinities**

Breuer and Melzer (1990a) contributed to the explanation of ion competition using *Sphagnum* (Figure 21-Figure 29) from a high moor. They found that when two or more ions are present, there is an order to the binding success: Pb++ > Cd++ > Ca++ > Mg++ > K+ > Na+ ≥ NH4+. Hence, those with higher binding affinities were able to suppress the binding of the lower affinity ions.

Breuer and Melzer (1990b) commented that *Sphagnum* (Figure 21-Figure 29) "shows behaviour of a relatively ideal ion exchanger." And, while species differ in their capacity, the coefficients of selectivity are independent of species. These bound cations can readily be displaced if another cation is present at a higher concentration, has a larger hydrated atomic radius, or has a higher valency (Bates 2000).

In *Sphagnum* (Figure 21-Figure 29) Hájek and Adamec (2009) found the exchangeable cation content decreased in the order of Ca++ ≥ K+, Na+, Mg++ > Al+++ > NH4+, whereas the intracellular element content demonstrated the order of N > K > Na, Mg, P, Ca, Al. While Ca occurred primarily on exchange sites, Mg, Na, and especially K, Al, and N occurred inside the cells. Vertical position in the bog influenced the nutrient uptake and location. Hummock species have a higher cation exchange capacity (CEC) and accumulate more exchangeable Ca++. By contrast, the hollow species have a lower CEC and accumulate more exchangeable Na+, especially among the lower dead shoot segments. Intracellular N and P were consistently lower in the dead portions, indicating their translocation to growing upper portions. *Sphagnum magellanicum* (Figure 29) has about 40% lower N content in its cells compared to other species, suggesting its inability to compete for N. This can cause it to lose competition to other species (Hájek & Adamec 2009), but its drought tolerance aids it in occupying tops of hummocks (Li et al. 1992). This leaves us wondering why it has such a low N content.

**Figure 27.** *Fontinalis antipyretica*, a species that loses potassium and magnesium when exposed to heavy metals. Photo by Bernd Haynold, through Wikimedia Commons.

**Figure 28.** *Rhytidiadelphus squarrosus*, a species that has an altered photosynthetic rate in the presence of cadmium. Photo by Michael Lüth, with permission.

**Figure 29.** *Sphagnum magellanicum*, a species with low N content. Photo by Michael Lüth, with permission.

Rühling and Tyler (1970) demonstrated the order of binding affinity of several heavy metal cations using *Hylocomium splendens* (Figure 19): Cu ≈ Pb > Ni > Co > Zn ≈ Mn. In a two-hour experiment, these heavier cations preferentially bound to the exchange sites even when lighter cations of Ca++, K+, Mg++, and Na+ were present in high concentrations. However, this sequence changed when the elements were supplied individually. Other researchers also demonstrated differences from this sequence in *Rhytidiadelphus squarrosus* (Figure 28) (Brown & Beckett 1985), *Brachythecium rutabulum* (Figure 7) (Brown & Buck 1978), and *Pohlia nutans* (Figure 30) (Webster 1985). These differences may relate to damage by some of the elements, such as cell membrane damage by mercury in some of the experiments,
concentration differences, and possible contamination from soil particles (Brown & Wells 1990).

In the moss *Rhytidiadelphus squarrosus* (Figure 28), competition occurs in the order of Cd ≅ H > Ca > Mg ≫ K on the extracellular sites (Wells & Brown 1990), suggesting that the low weight of H compensates for the higher valency of Cd. However, affinity of the intracellular Cd transport site occurred in the order of Ca > Cd > Mg ≫ K. Thus, internally calcium was a competitor for cadmium, but magnesium was not. Cadmium experienced maximum uptake at pH 5.6 and was very sensitive to pH. Potassium had no competitive ability against these ions. This presents another interesting question. How do internal differences in pH affect uptake, transport, and storage of ions? And what are the extent and locations of those pH differences?

Nieboer and Richardson (1980) found a divalent metal ion selectivity binding order of Pb > Cu > Cd > Co ≅ Fe > Ni > Zn > Mn, although Rühling and Tyler (1970) found a slightly different order for *Hylocomium splendens* (Figure 19): Cu ≅ Pb > Ni > Co > Zn ≅ Mn, an order that seems to be widespread in bryophytes (Bates 2000). However, once the sites are nearly fully occupied, this preferential binding is no longer the strongest force, possibly accounting for differences illustrated here. Isolated binding sites are only able to bind one position on the cation, hence eliminating the advantage for higher valency ions. In fact, at this stage, the isolated sites are more likely to bind univalent ions than divalent ones and more likely to bind divalent ones than trivalent ones (Richter & Dainty 1990). This is because divalent and trivalent ions require adjacent binding sites, whereas monovalent ions can utilize isolated sites. It is also likely that in systems with lower pH, more sites are occupied by H⁺ ions, creating more isolated sites. This would favor the binding of lower valency ions such as K⁺ and account for the high selectivity at a low pH.

**Habitat Differences**

Cation exchange sites can serve two conflicting purposes: bind the cations against further uptake, or concentrate them for absorption sites (Büscher et al. 1990). These roles have rarely been discussed for terrestrial bryophytes. This affects the bryophyte tolerance of various substrates. Using the acidiphile (preferring soils with pH <5) species *Atrichum undulatum* (Figure 31), *Leucobryum glaucum* (Figure 32, *Mnium hornum* (Figure 33), and *Polytrichium formosum* (Figure 34, and the neutrocline (preferring pH close to neutral, i.e. >5) species *Homalothecium sericeum* (Figure 35) and *Plagiomnium undulatum* (Figure 36), Büscher et al. found that acidophilous and acidiline taxa generally have lower CEC and are more able to tolerate the toxic aluminium (Al) levels, but not high levels of Ca. Neutrocline taxa instead avoid habitats with aluminium in the substrate and thrive on high calcium levels. (But what is the mechanism causing the avoidance?) They concluded that cation exchange properties do not protect mosses against potentially toxic ions, including aluminium, by sequestering them. But they did conclude that the exchange sites could increase the availability of cations. High CEC favored fixation of Al ions over Ca ions, indication that a low CEC is needed for taxa to tolerate acid soils.
Chapter 8-4: Nutrient Relations: Uptake and Location

Bates (1992) found that in epilithic and woodland soils the cation exchange capacity decreases with decreasing Ca content, and likewise with decreasing pH of the substrate. This change might help to protect the bryophytes against the toxic aluminium that increases in concentration in acidic solutions, e.g. soils polluted by acid rain.

Calcicole rocks, inhabited by calcicoles (Ca-preferring species), typically have Ca\(^{2+}\) concentrations 16-17 times that found in species from non-calcicole rocks (calcifuges – species avoiding Ca) (Bates 1982a). The calcicoles exhibit 3-4 times as many cation exchange sites as the calcifuges. Bates suggested that the calcicole mosses may require greater Ca\(^{2+}\) concentrations to maintain cell membrane integrity.

Uptake Rate

Uptake of these nutrients is very rapid when concentrations are high. Half the maximum extracellular uptake can be achieved in \(4.45\pm1.03\) minutes in 100 \(\mu\)mol L\(^{-1}\) Cd (Brown & Beckett 1985). This rate is concentration dependent and at lower (more natural) concentration levels it can take several days to reach equilibrium (Mouvet 1987). Release of the cations from the exchange sites when the element is removed from the medium takes even longer, as shown in the aquatic liverwort Chiloscyphus polyanthos (Figure 37-Figure 38) (Maurel-Kermarrec et al. 1985). The uptake ability varies between clones that grow within meters of each other (Wells & Brown 1987; Wells 1988). This can result from differences in light/moisture availability in the open vs under shrubs, as demonstrated in Rhytidiadelphus squarrosus (Figure 28) (Wells & Brown 1987). This difference could have been caused by thicker cell walls in the higher light population of R. squarrosus.
ammonium source of N (Rudolph et al. 1982). This preference likewise contradicts the results of Wanek and Pörtl (2008) who concluded that Sphagnum (Figure 21-Figure 29) prefers ammonium because of its numerous cation exchange sites. But it does coincide with the inhibition of nitrogen reductase by ammonium (Syrett & Morris 1963; Orebanjo & Stewart 1975). Furthermore, Wanek and Pörtl (2008) found that amino acids contributed a significant fraction of the N used by Sphagnum from the lowland rainforest in Puerto Rico. Brown (1982) suggested that anion adsorption is probably especially low in mosses because they have low iron and aluminium content and high cation exchange capacity Clymo 1963; Chapin et al. 1987). Phosphorus, as the phosphate anion, is taken up primarily from the mineral substrate (Bates 1992). Chapin et al. (1987) concluded that accumulation of the phosphate anion, as they observed in mosses of the Alaskan black spruce (Picea mariana, Figure 3) forest, was therefore by active absorption similar to that of higher plants. In these forests, mosses hold 17% of the phosphorus pool, despite accounting for 75% of the annual P accumulation. The mosses have a greater ability to absorb phosphate than do fine roots of the black spruce.

Proton Pumps

After ions have reached the surface of the cell, they require energy to enter the cell. In tracheophytes, the proton pump is well known in such activities as bringing nutrients into root hairs, opening and closing guard cells, closure of the Venus flytrap, and growth, to name only a few. In bryophytes, the proton pump has likewise been demonstrated, and like that of tracheophytes, it uses ATP to "pump" H+ ions out of a cell (Figure 39). This leaves the cell with a negative charge that attracts cations into the cell (Raven et al. 1998). The resulting negative charge provides the force needed to bring in K+, NH4+, Mg++, Ca++, sugars, and amino acids, and probably other cations that have not yet been confirmed experimentally.

Cotransport

As a positively charged ion enters the cell, it typically brings along an associated anion by cotransport. The pump, at the same time, regulates the pH within the cell to about 7.3-7.6. In bryophytes, the leaf cell surface and interstitial spaces between the cells provide sites where adhering cations are able to enter the cell through the proton pump mechanism.
As a result of ion movement through CEC and the proton pump, the bryophyte most likely has an influence on the rhizoidosphere (soil space immediately surrounding rhizoids) similar to that of tracheophytes on the rhizosphere (Raven et al 1998), although in the case of bryophytes, leaves may contribute to the alteration of conditions even more than the rhizoids. The rhizoidosphere is acidified in the process of cation exchange and proton pumping to bring nutrient cations into the cells, creating positive charges within the cells and accumulating organic anions in the cell vacuoles.

Pinocytosis

Pinocytosis results when a cell ingests a liquid by budding small vesicles inward from the cell membrane, thus containing the droplet. The droplet of liquid then is incorporated into the cell cytoplasm. Gullvåg et al. (1974) observed this mechanism in Rhytidadelphus squarrosus (Figure 28) when it was treated with lead-rich particles in the lab. When they tested mosses that were exposed to lead pollution in the field, they found that the lead was bound within the nucleus. The importance of pinocytosis for incorporating nutrients into the cells of bryophytes seems to lack study.

Nanoparticles

The concept of nanoparticles is a relatively new idea in bryophyte ecology. Canivet et al. (2014) found, for the first time, that nanoparticles of iron in a mineral water suspension could penetrate the leaves of the moss Physcomitrella patens (Figure 40). In follow-up experiments Canivet et al. (2015) further demonstrated the penetration of iron nanoparticles into the moss Physcomitrella patens. Using concentrations of 5 ng, 50 ng, 500 ng, 5 µg, and 50 µg per plant, they found no effect on ATP concentrations, reactive oxygen species, malondialdehyde, or glutathione, suggesting that the plants had not been physiologically harmed at any of these concentrations. The role of nanoparticles in providing essential nutrients or harming the plants seems to thus far lack exploration.

Influence of Cellular Structures

Many studies have treated cellular influences as if the cells were homogeneous (Brown & Wells 1990). First, the nutrient must cross the cell membrane. But in fact, once inside the cell, the nutrient may be held in solution, like K, or bound into amino acids and proteins, like some of the N and P. The chlorophyll molecule can take Mg out of play. When these binding compounds take the nutrient out of solution, they affect the concentration gradient from outside to inside the cell, affecting the concentration gradient used for the nutrient to cross the cell membrane and enter the cell. Others are bound to intracellular binding sites, again altering uptake rate.

Pickering and Puia (1969) described three phases of element uptake against time, an "unusual" process compared to that in algae and tracheophytes (Brown & Wells 1990). The first phase is the initial rapid uptake as the ions diffuse into interstitial spaces in the tissues (Pickering & Puia 1969). Then the uptake is controlled by equilibration with cell wall exchange sites. The final phase is a slow, linear increase of intracellular uptake. This third phase does not occur in dead material. As demonstrated in Rhytidadelphus squarrosus (Figure 28), carriers can be used to transport the element across the membrane (Brown & Beckett 1985; Wells & Brown 1987). Specificity of these carriers determines how much inter-element competition there is for the intracellular uptake. This in turn affects the rate of uptake.

Location Is Important

As already noted, location of nutrients on and in the bryophytes is important (Brown and Buck (1985). The method used can present a bias that is misleading regarding normal nutrient concentrations. A nutrient adhering to the cell wall is not immediately available to the cells and may not be representative of the needs of the cells. Others may be held in the spaces within the walls. To fully understand the nutrient physiology, we must understand where these nutrients are located on and in the bryophyte plants (see Table 2).

Table 2. Element locations in bryophytes. Based on Brown 1982.

<table>
<thead>
<tr>
<th>Location</th>
<th>Nutrient</th>
</tr>
</thead>
<tbody>
<tr>
<td>in particles trapped by leaves</td>
<td></td>
</tr>
<tr>
<td>in solution on exterior &amp; in matrix of cell wall</td>
<td></td>
</tr>
<tr>
<td>as ions bound to external exchange or chelating sites &amp; on plasma membranes</td>
<td></td>
</tr>
<tr>
<td>in solution in cytoplasm &amp; vacuoles</td>
<td></td>
</tr>
<tr>
<td>as insoluble substances in cytoplasm &amp; vacuoles</td>
<td></td>
</tr>
<tr>
<td>in leptome (especially Polytrichaceae)</td>
<td></td>
</tr>
</tbody>
</table>

New Growth

When new branches are formed and expand in the absence of additional nutrients, these nutrients must be obtained from existing tissues. In some cases, this is through acropetal (base to tip) transfer, as seen in Rhytidadelphus squarrosus (Figure 28) (Wells 1988). Potassium, a very soluble nutrient, declines in lower portions as the apex grows. Calcium, on the other hand, is
not soluble and is taken from the initial apical segment, not transported from older tissues. Magnesium exhibited a somewhat similar response, but all segments lost Mg from intracellular sites as the apex grew.

**Specificity**

Some nutrients are taken up more easily than others. Leblond (2004) examined the uptake of heavy metals in the moss *Pseudoscleropodium purum* (Figure 14). The nutrient elements manganese and potassium had the highest retention. Non-nutrient ions of sodium, aluminium, and silica had the least retention. Youngest tissues accumulated the most nutrients, but internal redistribution occurred. Leblond found that soluble materials were taken in more easily than those deposited as particulates.

We know that cation exchange sites selectively bind higher valency cations (Richter & Dainty 1990). But at least in *Sphagnum russowii* (Figure 41-Figure 43) there are two classes of exchange sites. The well-known one is associated with polygalacturonic acids and accounts for more than 50% of the cation exchange capacity (Richter & Dainty 1989). In addition to that, phenolic acids account for about 25%, whereas amino acid, sulfate ester, and silicate deposits in the cell wall contribute to a lesser degree.

Figure 41. *Sphagnum russowii*, a species with both polygalacturonic acid and phenolic acid exchange sites. Photo by Ralf Wagner <www.dr-ralf-wagner.de>, with permission.

Figure 42. *Sphagnum russowii* leaf cells showing the exposed surface area of the much larger hyaline cells where cation exchange can occur on both inside and outside of the cell. Photo by Ralf Wagner <www.dr-ralf-wagner.de>, with permission.

Figure 43. *Sphagnum russowii* leaf cells in cross section showing the exposed surface area of the much larger hyaline cells where cation exchange can occur on both inside and outside of the cell. Photo by Ralf Wagner <www.dr-ralf-wagner.de>, with permission.

When studying aquatic bryophytes, Burton and Peterson (1979) found that 33% of the cell-wall-bound Zn could be removed by the enzyme pronase (mix of enzymes that break down proteins), suggesting that a considerable portion of its binding might be due to protein binding. Richter and Dainty (1989) found a small number of binding sites that are more specific to small valency cations such as potassium. If these sites include phenolic compounds, one can presume that such binding sites might be widespread in bryophytes, wherein phenolic compounds are common (Mues & Zinmeister 1988; Liao 1993; Basile *et al*. 1999). Is this yet another use for these presumed "secondary" compounds? If so, what does it mean for cycling of potassium if it can be bound to the cell walls? Does this help the plant to retain its valuable potassium when cell membranes, damaged by desiccation, permit potassium to leak from the cell? Such a mechanism could contribute to the survival of bryophytes after desiccation and permit them to become a long-term sink for this and other ions.

We know that cation exchange is a somewhat selective process. Higher valency ions are bound preferentially over lower ones because they occupy more than one exchange site. Cations have binding preferences (Nieboer & Richardson 1980, 1981). **Class A** includes K, Ca, Mg, and S. These elements prefer oxygen-rich ligands, such as carboxylic groups. **Class B** elements such as Ag, Cu, Hg, Pb, and Au are toxic and prefer ligands that are rich in sulfur and nitrogen. The third group is a borderline class that includes Cu ++, Ni ++, Pb ++, and Zn ++. Hence, the quantity of any element bound to the cell wall will depend on concentration in the medium (precipitation, water, soil), its affinity for type of exchange site, and the total number of suitable exchange sites.

Dead cells may actually have more exchange sites than live ones due to shrinkage of cell membranes that cover them on the inside. On the other hand, Wells (1988) found that when the cells are killed by a strong acid, the exchange capacity decreases, a phenomenon he attributed to loss of cytoplasm.

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the cells are killed by a strong acid, the exchange capacity decreases, a phenomenon he attributed to loss of cytoplasm.

Fortunately, bryophytes seem to have uptake specificity for things they need over things they do not. For example, the thallose liverwort *Dumortiera hirsuta* (Figure 23) preferentially took up Ca, Mg, and Zn over Cd (Mauts & Beckett 1996). When KNO₃ was used to pretreat the plants, Cd uptake occurred, suggesting that the high concentration of K⁺ removed the competing ions from the exchange sites and they were subsequently replaced by Cd. Light and increased temperatures also stimulated Cd uptake. Even *Sphagnum* (Figure 21-Figure 29, Figure 41-Figure 44), the champion of cation exchangers, distinguishes among ions in ways that do not seem to depend strictly on valence. It accumulates Al and Mn, but excludes Cu and Zn, accumulating much less of these than the concentrations in the surrounding fen water (Li & Glime 1990).

Shimwell and Laurie (1972) found that ectohydric and mixohydric mosses differ in their absorption, retention, and excretion of heavy metals. During droughts, **ectohydric** (having external conduction) mosses excrete such heavy metals as Zn and Pb, forming surface crusts containing up to 6% Pb and 1-5% Zn. In **mixohydric** (having both external and internal conduction) mosses, on the other hand, the metals generally are located at the base of the moss carpet in the older growth, suggesting their accumulation in older tissues and lack of internal transport.

**Seasons**

Since most bryophytes gain most of their nutrients from precipitation, we might assume that most nutrient uptake therefore occurs when it rains. Yet the relationship is most likely not so simple. Francez and Loiseau (1999) found that *Sphagnum fallax* (Figure 44) was more efficient at intercepting applied N (as NH₄NO₃) in August than in June, even though August had the lowest rainfall. Dust accumulation can benefit bryophytes that are able to absorb nutrients in early morning dew and even on humid nights when there is no benefit for tracheophytes.

Figure 44. *Sphagnum fallax*, a species that takes up more N in August than in June. Photo by Michael Lüth, with permission.

Bates (1992) considered that in rapidly growing species such as *Brachythecium rutabulum* (Figure 7) the seasonal deposition of tracheophyte litter is especially important. In the slower-growing species such as *Pseudoscleropodium purum* (Figure 14), wet deposition may be the most important.

Turner and coworkers (2003) found that rates of acid phosphatase activity in moss apices differed markedly among species, but most taxa had the most activity in winter and least in summer. Nevertheless, tissues maintained relatively constant N and P concentrations throughout the year. A negative correlation between phosphatase activity and P concentration in the tissues suggests that the enzyme may become active in response to phosphorus needs and serves to indicate nutrient stress. Núñez-Olivera *et al.* (2001) found that seasonal differences in several aquatic bryophytes [*Fontinalis antipyretica* (Figure 27), *F. squamosa* (Figure 45), *Jungermannia eucordifolia* (Figure 45), and *Pellia endivifolia* (Figure 47)] did not mimic the seasonal differences in their native streams. Rather, the concentrations depended on the interactions of internal and external factors. The elements that had the most persistent annual cycle were mostly essential nutrients: N, P, and Fe, plus the non-essential Na. The lowest concentrations occurred in spring and the highest in autumn. Concentrations were lowest during periods of growth.

Figure 45. *Fontinalis squamosa* in alpine water. Photo from <www.aphotofauna.com>, with permission.

Figure 46. *Jungermannia eucordifolia*, a species for which internal nutrient concentrations do not mimic those of its stream habitat. Photo by Jan-Peter Frahm, with permission.
Glucose Uptake

External glucose can enhance growth of at least some bryophytes (Jennings 1918). *Ceratodon purpureus* (Figure 48) grew 4-5 times as much when provided with glucose on nutrient agar compared to nutrient agar without glucose. This implies that organic sources of carbon that may be available in the substrate are suitable carbon sources for at least some mosses. Vujičić et al. (2009) found that the best conditions for axenic culture of the moss *Dicranum scoparium* (Figure 49) was in MS medium enriched with sucrose at 1.5% at 18-20°C. Adding glucose, fructose, and sucrose to the medium causes these bryophytes to down-regulate photosynthesis when the bryophytes are kept either in the dark or in the light (Marschall 2010). On the other hand, when no carbohydrates were added, darkness had little influence on total carbohydrates, suggesting that they maintain a well-regulated carbohydrate pool.
Figure 52. *Sphagnum flexuosum*, a species that stores sucrose and fructan and down-regulates photosynthesis when sugars are available in the medium. Photo by Michael Lüth, with permission.

Sugars differ in their effects on bryophyte development (Sabovljevic et al. 2005). In *Bryum argenteum* (Figure 53), added sugars have a positive effect on development of the protonema and multiplication of the shoot. On the other hand, all tested sugars had a negative effect on both of these developmental stages in *Atrichum undulatum* (Figure 31).

Figure 53. *Bryum argenteum*, a species for which added sugars have a positive effect on development. Photo from India Biodiversity Images, through Creative Commons.

Not only do bryophytes store sugars, but as we might expect as a consequence, they also release them (Coxson et al. 1992). In the tropical montane rainforest of Guadeloupe, frequent wet-dry cycles cause the epiphytic bryophytes to accumulate 950 kg ha⁻¹ of sugars and polyols. These are released during rewetting, contributing to sugars available to other organisms in the canopy. The canopy leafy liverwort *Frullania atrata* (Figure 54) stored 17% of its dry biomass as sugar and polyol reserves, whereas the lower canopy species *Phyllogonium fulgens* (Figure 55) stored less than 6%. On the other hand, it was the lower canopy bryophytes that released the most sugars and polyols (0.9 g m⁻²) during rewetting, compared to 0.3 g m⁻² for the upper canopy. This release yielded an estimated 122 kg ha⁻¹ from the upper canopy. These sugars contribute significantly to nutrient cycling by providing an energy source for the decomposer organisms.

Figure 54. *Frullania atrata*, a species that has 17% of its dry biomass as sugars in the canopy of a tropical cloud forest. Photo by Juan Larrain, with permission.

Figure 55. *Phyllogonium fulgens*, a Neotropical species that lives in the lower canopy and stores less than 6% of its dry biomass as sugars. Photo by Michael Lüth, with permission.

**Fungal Connections – Mycorrhizae?**

One mode of uptake by bryophytes has largely been ignored by ecologists until recently, potentially causing researchers to be looking in the wrong places or not all important places for bryophyte effects on ecosystem nutrient budgets. That mode is by means of mycorrhizae (fungal associations that function in transfer of nutrients to roots or rhizoids) or similar partnerships with fungi.

In 1976, Kottke and coworkers recognized that the ability of mosses to compete was affected by differential growth stimulation of the mosses by fungi. Still, little attention was paid to moss-fungal interactions from an ecosystem perspective, but bryologists began noticing that many mosses seemed to have fungal hyphae associated with their underground parts. Meanwhile, the tree physiologists were recognizing that fungal partners were critical to the nutrient and water uptake of trees. And orchid growers recognized that the native fungi must be kept with the orchids for successful growth. Now, fungi are recognized as essential to the nutrient uptake of tree
roots, and stories about their partnerships with roots are replacing the traditional teaching emphasis on root hair mechanisms of uptake.

Ecologists estimate that 95% of all plant species are in genera that form mycorrhizal associations (Sylvia et al. 2004). In temperate and boreal forests, up to 95% of the short roots of trees form **ectomycorrhizae** [form of symbiotic relationship that occurs between a fungal symbiont and the roots (or rhizoids) of various plant species]. Mycorrhizae are critically important to most forest trees, which depend on them to increase surface area and contact nutrients in a much greater volume of soil than the tree is able to reach. Bryophytes, likewise, are able to take advantage of this partnership to reach sources otherwise unavailable to them. Even in the Antarctic, such fungal relationships can be important, as in the leafy liverwort *Cephaloziella exiliflora* (Figure 56) (Williams et al. 1994; Chambers et al. 1999). There are also indications that nutrients are transferred from the moss mat to the tree roots through mycorrhizae (Chapin et al. 1987). But we know little of the extent of these relationships.

Although Boros reported a unique parasitic fungus on mosses in 1926, most botanists considered the bryophytes to be almost immune from fungal attack; even less attention was paid to the possibility of any sort of fungal partnership. In 1970, Kamal and Singh reported on the **rhizoidosphere** fungal flora of bryophytes. In 1975, Pirozynski and Malloch offered the theory that mycorrhizae were an essential part of the invasion of land by the original bryophyte-like plants, helping them to survive in an environment that was poor in nutrients and sustained frequent periods of desiccation. But actual proof of a mycorrhizal partnership, extant or extinct, was not forthcoming.

Finally, in the 1980's, reports of bryophyte mycorrhizal (shouldn't it be mycorrhizoidal?) associations began to appear in the literature (Parke & Linderman 1980; Rabatin 1980; Pocock & Duckett 1985a; Iqbal et al. 1988a, b; Ligrone 1988). These have included associations with *Funaria hygrometrica* (Figure 57) (Parke & Linderman 1980; Iqbal et al. 1988a), *Sphagnum palustre* (cymbifolium) (Figure 58-Figure 59), *Polytrichum commune* (Figure 4) (Iqbal et al. 1988a), and in *Marchantia emarginata* (=*M. palmata*, Figure 60) both rhizoids and the ventral thallus (Iqbal et al. 1988b).
Ligrone and Lopes (1989) demonstrated vesicles and arbuscules ("little trees"; branched structures formed by fungi within plant cells; Figure 61) in both rhizoids and parenchyma cells of the thallose liverwort Conocephalum conicum (Figure 62), suggesting a true mycorrhizal association. The arbuscules are thought to be the site of nutrient exchange (Harrison 1999), at least in roots. Even Phaeoceros laevis (Figure 63), a member of the Anthocerotophyta and host of a Nostoc (Cyanobacteria; Figure 64-Figure 65) symbiont, has a fungal associate that appears to be mycorrhizal (Ligrone 1988). When P. laevis is infected, the plastid forms a networking structure, the vacuole mass decreases, and the organelle density increases, all modifications suggestive of a partnership.

In 1985, Pocock and Duckett (1985b) investigated the rhizoids of 206 of the 284 British liverworts. They found that swollen rhizoids occurred in 33 species of the Jungermanniales and were always associated with fungal hyphae. Swollen and branched rhizoids were particularly well developed in the Lepidoziineae (Figure 66) and Cephaloziineae (Figure 94) and often occurred on flagelliform shoots, but were better developed on the
underground axes. Duckett et al. (1991) later described the highly specialized associations between ascomycetous fungi, known for their ectomycorrhizal partnerships, and 46 species of British liverworts. They found the majority of these ascomycetous fungi to occur with the leafy liverwort suborders **Lepidoziinae** and **Cephaloziinae**. [Ascomycetous associations are found in a relatively small number of families of leafy liverworts (Read et al. 2000)]. Strikingly, 33 of these 46 British liverwort taxa form flagelliform axes (Duckett et al. 1991). These axes have elongate parenchyma cells with abundant plasmodesmata in their transverse end walls. Their apices are mucilaginous and the subapical amyloplasts appear to act in detecting gravity, much as they do in protonemata. In addition to serving as perennating structures, these axes appear to be major organs of assimilation. Is this facilitated through a mycorrhizoidal partnership?

In all these leafy liverwort cases, the fungi infect the individual rhizoids independently, but most of these 46 taxa nevertheless have abundant fungi-infected rhizoids that extend 20-30 cm into the peaty substrate (Duckett et al. 1991). What an extension for a tiny bryophyte! In the liverworts *Lepidozia* (Figure 66), *Kurzia* (Figure 67), and *Telaranea* (Figure 68), but known in no others, the rhizoids swell prior to fungal infection. In *Cladopodiella* (Figure 69), the fungi form a pseudoparenchymatous sheath around the swollen rhizoidal tips.

By 1988, Boullard had presented evidence that the fungal symbiotic relationship with the liverworts was evolutionarily very old. Yet, in 1990, During and van Tooren pointed out that "in only very few cases have these interactions been analysed functionally."

Other associations have been documented in the field. Although not truly mycorrhizoidal because they lack the composite structure definitive of this relationship, bryophytes now are known to enter into partnerships. Even buried wood, inoculated with $^{32}$P, was able to provide P for the living tips of *Hypnum cupressiforme* (Figure 70-Figure 71) through a saprotrophic fungus, *Phanerochaete velutina* (Figure 72), that connected to the older parts of the moss (Wells & Boddy 1995).
Figure 71. *Hypnum cupressiforme*, a species that is able to derive phosphorus from buried wood. Photo by David Holyoak, with permission.

Figure 72. *Phanerochaete velutina*, a fungal associate of *Hypnum cupressiforme* on wood. Photo by James K. Lindsey, with permission.

The fungal association may in some small way benefit the neighboring plants, and they in turn the bryophyte (Duckett & Read 1995). As noted earlier, Chapin *et al.* (1987) have found an association that may indeed benefit the trees. In an Alaskan forest they found that the mycorrhizal fungi of the black spruce (*Picea mariana*, Figure 3) stimulated the moss carpet above to release phosphorus to the tree roots! When the mycorrhizae were inhibited, more P remained with the mosses and less escaped from the plots, where it presumably went to tree roots.

Rhizoids of at least some leafy liverworts in the Lepidoziaceae (Figure 66), Calypogeiaceae (Figure 73), Cephalozia (Figure 94), and Cephaloziiellaceae (Figure 56) can be infected by the same fungus, *Hymenoscyphus ericae* (Figure 74), an ascomycetous fungus, that infects members of the Ericaceae such as Calluna (Figure 75), Erica (Figure 76), Rhododendron (Figure 77), and Vaccinium (Figure 78-Figure 79) (Duckett & Read 1995). So far, there appears to be no evidence of a transport pathway from moss to fungus to ericaceous plant or vice versa, but the presence of one of these host plants would enhance the opportunities for the fungus to grow there and thus provide greater opportunities for the fungus to join with the other host. This is similar to the partnership between *Monotropastrum humile* (an achlorophyllous flowering plant; Figure 80), a fungus, and a beech tree (*Fagus crenata*, Figure 81) (Kasuya *et al.* 1995). The fungus in the *F. crenata* appears to be the same as that in the *M. humile*, and evidence implies that the fungus joins the two tracheophytes. In this way, the *M. humile* could take advantage of the sunlight reaching the canopy of *Fagus crenata* by receiving carbohydrates from the canopy transferred through the fungus to the *M. humile*. The fungus appears to be a member of the Russulaceae (Figure 82) (Yamada *et al.* 2008; Matsuda *et al.* 2011).

Figure 73. *Calypogeia azurea*, a leafy liverwort that can be infected with *Hymenoscyphus ericae*. Photo by Hermann Schachner, through Creative Commons.

Figure 74. *Hymenoscyphus ericae* on *Rhododendron* root, a species that can infect leafy liverworts in Lepidoziaceae (Figure 66), Calypogeiaceae (Figure 73), Cephalozia (Figure 94), and Cephaloziiellaceae (Figure 56). Photo by Mark C. Starrett, David A. Heleba, and Adam R. Wheeler, through Creative Commons.

Figure 75. *Calluna vulgaris*, a host for the fungus *Hymenoscyphus ericae*. Photo by Janice Glime.
Figure 76. *Erica* sp., a host for the fungus *Hymenoscyphus ericae*. Photo by Janice Glime.

Figure 77. *Rhododendron ferrugineum*, a host for the fungus *Hymenoscyphus ericae*. Photo by Albert Kok, through Creative Commons.

Figure 78. *Vaccinium angustifolium* in *Pinus banksiana* forest. *Vaccinium* is a host for the fungus *Hymenoscyphus ericae*. Photo by Photo by Mricon, through Creative Commons.

Figure 79. *Vaccinium angustifolium*, a host for the fungus *Hymenoscyphus ericae*. Photo by Mricon, through Creative Commons.

Figure 80. *Monotropastrum humile*, an achlorophyllous flowering plant that partners with a beech tree through a fungal partner. Photo by Qwert, through Creative Commons.

Figure 81. *Fagus crenata*, host tree for *Monotropastrum humile* and its fungal partner. Photo by Alpsdake, through Creative Commons.
Cryptothallus mirabilis

It appears that the fungi may be to some liverworts what the mycorrhizae are to the grape fern Botrychium and to many of the saprophytic forest floor flowering plants – a means of getting sufficient energy when the canopy is blocking an extensive portion of the light. Such a relationship is essential to the thallose liverwort Cryptothallus mirabilis (Figure 83), a European species known as ghostwort. It occurs nestled in mires and lacks chlorophyll. Certainly for it, a partnership is essential. But this liverwort has a Basidiomycota fungus as its ectomycorrhizal partner (Ligrone et al. 1993). They concluded that this liverwort is a parasite! It was thought that its fungal partner joined it to a species of Betula (birch), from which it ultimately obtained its carbohydrate energy source (Wiehle 1988; Pocock & Duckett 1984; Frey & Kürschner 1991; Read et al. 2000), much like the parasitic flowering plant Monotropa uniflora (Figure 84), the Indian pipe. However, Ligrone et al. (1993) disagree. They found that the fungi in Betula roots had a different morphology from those in the associated C. mirabilis. It appears that the association of C. mirabilis is more like that of the goblin fern Botrychium mormo, wherein the fungus derives carbon from decomposing litter and transfers some of it to the fern, permitting it to live in low light (Gundale 2002). But could it also be that the form of the fungus depends on the host, thus differing between that of the C. mirabilis and that of the Betula?

Bidartondo et al. (2003) determined that Cryptothallus mirabilis (Basidiomycota; Figure 83-Figure 86) is an epiparasite, depending on a species of the fungus Tulasnella (Figure 85-Figure 86). This fungus forms ectomycorrhizal (symbiotic relationship between fungal symbiont and roots of plant species) associations with surrounding trees. It is able to transfer labelled $^{14}$C from birch (Betula) seedlings in the lab, and presumably from tree roots in the field. Species of this same genus are also associated with Aneura pinguis (Figure 87) (Kottke et al. 2003) and some orchids (Clements & Ellyard 1979; Roche et al. 2010).
When it develops, the *Cryptothallus mirabilis* (Figure 83) fungus (Figure 88-Figure 91) forms large, intracellular coils in the liverwort (Ligrone *et al* 1993). Then the liverwort cytoplasm proliferates and the starch content of its plastids decreases. As the hyphae die back and aggregate into large masses, the liverwort cells senesce. In *C. mirabilis*, the fungal hyphae contain abundant glycogen and occasionally amyloid deposits. It is interesting that the fungal partner in *C. mirabilis* is identical to the one in *Aneura pinguis* (closely related but photosynthetic; Figure 87) from alpine sites but different from the fungus in *A. pinguis* from a chalk pit and sand dunes. In *C. mirabilis*, net carbon transfer is to the liverwort, and it is likely that there is transfer from the fungus to the liverwort in *A. pinguis* as well. In addition to the morphological similarities, further support for this hypothesis in *A. pinguis* is that spores of both liverwort species fail to develop beyond a few cells in *axenic* (sterile) culture.
Bidartondo and Duckett (2010) concluded that most of the thalloid liverworts contain **Glomeromycota** (Figure 92) that form arbuscular mycorrhizae with them. Many leafy liverwort species and members of the thalloid **Aneuracea** have a relationship with **Basidiomycota**. Whereas the **Aneuracea** associate almost exclusively with species of **Tulasnella**, eight leafy liverwort genera predominately associate with members of **Sebacina vermifera** (**Basidiomycota**; see Figure 93). **Sebacina** species have a habit of surrounding plants, so some of them may envelop the plants and prevent photosynthesis. It is interesting to note that when multiple species of bryophytes occur together, they rarely share the same fungal species. Furthermore, the bryophyte symbioses are not like those of the tracheophytes.

Underground and Other Partnerships

It appears that **Cryptothallus** (Figure 83) is not the only liverwort capable of living below ground with an **Ascomycota** fungal partner (Duckett *et al.* 1989). In bog communities, the leafy liverworts (**Jungermanniales**) **Cephalozia** (Figure 94), **Cladopodiella** (Figure 69), **Kurzia** (Figure 67), **Lepidozia** (Figure 95), **Odontoschisma** (Figure 96), and **Telaranea nematodes** (Figure 68) can all develop extensive underground stem systems with numerous rhizoids that have swollen, fungus-containing tips. These liverworts can produce new shoots down to 24-30 cm in peat and to 10 cm in rotten logs (**Lepidozia reptans**, Figure 95).
Figure 96. *Odontoschisma macounii*, a species that houses fungi in swollen rhizoid tips in bogs. Photo by Michael Lüth, with permission.

In Malaysia, members of the leafy liverwort family *Lepidoziaceae* (Figure 95) can produce such axes down to 1.5 m in the peaty soil of the upper montane rainforest (Duckett *et al*. 1989). When these develop in the dark, they retain their partnership morphology, but when the shoots are exposed to light they regenerate into leafy shoots and lose their gravitropic response. This loss of fungal partnership morphology appears to be related to the disappearance of subapical amyloplasts, known to have a gravimetric response. Duckett and coworkers suggest that these liverworts may be acting as alternative hosts to ericaceous mycorrhizae, particularly in places like Malaysia. In Great Britain, less than 20% of the *Jungermanniales* (Figure 94-Figure 95) have rhizoidal fungi, whereas in the montane forests of Malaysia, where ericaceous shrubs are extensive, the percentage may be as high as 80-90%.

As the search continues, more and more fungal taxa are being described in bryophyte associations, but not all are mycorrhizal (Khan *et al*. 1997; Döbbeler 1997; Brouwer 1999). In fact, a number appear to be parasitic; others are just coexisting, perhaps benefitting from the modulated temperature and moisture. Nevertheless, approximately 300 species of *Ascomycota* appear to grow obligately on bryophytes (Döbbeler 1997). More than 40 species of *Ascomycota* in six orders occur on the *Polytrichaceae* alone, primarily on *Polytrichum s.l.* (Figure 4, Figure 34) and *Dawsonia* (Figure 97) (Felix 1988). Some fungi, for example *Lemprospora* (Figure 98) and *Octospora* (Figure 99), are known only from bryophytes (Döbbeler 1997; Brouwer 1999); in other cases, the bryophyte has never been found without its fungal associate (Döbbeler 1997). *Octospora* and other genera infect the subterranean rhizoids of *Polytrichaceae* (Figure 4, Figure 34, Figure 97), while others occupy the spaces between the vertical leaf lamellae (Felix 1988). In fact, 20 different *Ascomycota* species are known to occupy that unusual habitat without apparently having any effect on the moss.

Figure 97. *Dawsonia superba*, a genus that has *Ascomycota* associates. Photo by Jan-Peter Frahm, with permission.

Figure 98. *Lamprospora seaveri*, a fungus that only occurs on bryophytes. Photo by G. Moyne, through Creative Commons.

Figure 99. *Octospora excipulata*, a fungus that lives exclusively on bryophytes. Photo by Malcolm Storey, through Creative Commons.

Raspe and De Sloover (1998) suggested that the discomycetous fungus *Mniaecia jungermanniae* (Figure 100-Figure 101), which lives exclusively on leafy liverworts in the *Jungermanniales* (Figure 100), might have achieved the first step toward mutualism. This destructive parasite grows inside the bryophyte rhizoids but
does not seem to afford any direct benefit to the liverwort. It appears it has a long way to go to reach mutualism.

Figure 100. *Mniaecia jungermanniae* (fungus in center) on leafy liverworts. Photo by Malcolm Storey (DiscoverLife), with online permission.

Figure 101. *Mniaecia jungermanniae* asci and ascospores. Photo by Malcolm Storey (DiscoverLife), with online permission.

We have noted several times that bryophytes obtain phosphate, and possibly other nutrients, from the bedrock. It is likely that at least in some cases fungi contribute to this nutrient source. Calling them "rock-eating mycorrhizae," Schöll *et al.* (2008) demonstrated that tunnels occur in mineral grains. They attributed these to hyphae from ectomycorrhizae that can dissolve mineral grains. Whether these fungi are directly associated with bryophytes, or they use litter or other plants for their carbon source, these fungi permit phosphates and other nutrients to enter nutrient cycling, potentially making some of them available to the bryophytes.

### Arbuscular Mycorrhizae

Harrison (1999) reported that arbuscular mycorrhizae, restricted to the fungal order *Glomales* (*Zygomycota*, more recently named *Glomeromycota*; Figure 92), infected some bryophytes. Schüßler (2000) reported that a member of this order, *Claroideoglomus claroideum* (Figure 92), formed a mycorrhiza-like symbiosis with the hornwort *Anthoceros punctatus* (Figure 102-Figure 103). Following inoculation with spores, Schüßler found branched hyphae within the thallus within 20 days. This was the first definite experimental establishment of an arbuscular mycorrhiza-like association between a member of the *Glomales* and a bryophyte, although Felix (1988) had reported mycorrhiza-like associations in a number of taxa (Table 3). In 2003, Jakucs *et al.* found vesicles of a glomalean fungus in the moss *Hypopterygium* (Figure 104), suggesting that there might indeed be a mutualistic relationship in which the fungus also benefits, but that hypothesis still awaits verification.

Figure 102. *Anthoceros punctatus* with young sporophytes; *Claroideoglomus claroideum* forms a mycorrhizal association with this species. Photo by Des Callaghan, with permission.

Figure 103. *Anthoceros punctatus* antheridial pit and *Nostoc* colony (dark area). Photo by Des Callaghan, with permission.
Figure 104. *Hypopterygium didictyon*, a species that associates with a glomalean fungus (Figure 92). Photo by Juan Larrain, through Creative Commons.


<table>
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<tr>
<th>Fungus</th>
<th>Bryophyte</th>
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<tr>
<td></td>
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There is a certain degree of specificity among the bryophyte species that have fungal associations. Russell and Bulman (2004) found that *Marchantia foliacea* (Figure 107-Figure 109) from two locations in New Zealand supported *Glomus* (Figure 109) (*n.b.*, many species of *Glomus* are now placed in *Claroideoglomus*; Figure 92) arbuscular fungi internally (Figure 109), but that *M. polymorpha* (Figure 110) did not. Every *M. foliacea* thallus they examined contained this *Glomus* species in the parenchyma tissue around the midrib. The fungus invaded the thallus through the smooth rhizoids and grew upward through the thallus, forming arbuscules only in the upper portion of the thallus. The hyphae crossed directly through the cell walls of the liverwort. This same fungus forms mycorrhizal associations with the conifer, *Podocarpus* (Figure 111), and it may be that this fungus is shared by both plants. Unfortunately, we still have no evidence if this relationship between the fungus and the liverwort is truly symbiotic.

Figure 105. *Mycena* sp.; *M. cinerella* forms mycorrhizae with *Atrichum undulatum*, *Brachythecium rutabulum*, and *Funaria hygrometrica*. Photo by James K. Lindsey, with permission.

Figure 106. *Endogone pisiformis*, a genus known to form mycorrhizae with bryophytes. Photo by Adolf and O. Ceska, with permission.

Figure 107. *Marchantia foliacea* thallus, a species that houses arbuscular growth of the mycorrhizal fungus *Glomus* (Figure 92) around the midrib. Photo courtesy of Julia Russell.
Figure 108. *Marchantia foliacea* thallus with arbuscular growth of the mycorrhizal fungus *Glomus* (Figure 92) around the midrib. Photo courtesy of Julia Russell.

Figure 109. SEM *Marchantia foliacea* thallus with arbuscular growth of the mycorrhizal fungus *Glomus* in cells around the midrib. Photo courtesy of Julia Russell.

Figure 110. *Marchantia polymorpha* developing archegoniophores, a species that does not form an association with the fungus *Glomus* (Figure 92). Photo by Bob Klips, with permission.

The specificity of some of the groups for specific plant phyla is fascinating. For example, *Zygomycota* (Figure 112) colonize members of *Anthocerotophyta* (Figure 102) and *Marchantiophyta* (Figure 110, Figure 113), but not *Bryophyta* (Figure 104) (Read et al. 2000). On the other hand, members of the *Glomales* (Figure 92) isolated from the flowering plant *Plantago lanceolata* were able to colonize the thallose liverwort *Pellia epiphylla* (Figure 113) and produce arbuscules and vesicles.

Figure 111. *Podocarpus*, a genus whose roots serve as host for *Glomus* (Figure 92) and may share it with bryophytes. Photo by Koppchen, through Creative Commons.

Figure 112. *Zygomycota* sporangia, a phylum that colonizes hornworts and liverworts, but not mosses. Photo by Kristi Yim, through Creative Commons.

Figure 113. *Pellia epiphylla*, a species that can be colonized by the same member of *Glomales* as those found on the flowering plant *Plantago lanceolata*. Photo by Michael Lüth, with permission.
Beneficial or Harmful?

The fungal associates are not always beneficial to the bryophytes. Zobel et al. (1999) treated a sub-Arctic forest community with fungicide and found that the bryophytes and dwarf shrubs increased in biomass relative to the community with fungicide and found that the bryophytes and dwarf shrubs increased in biomass relative to the control. Could it be that the fungi are frequently stealing from the bryophytes and making nutrients available to trees?

Summary

Unlike tracheophytes, bryophytes take up nutrients over their entire surface. With leaves only one cell thick in most taxa every leaf cell is thus exposed to environmental sources of nutrients. The three most limiting nutrients (N, P, K) accumulate in the upper parts of the plants through active uptake, whereas Ca, Mg, and Na accumulate through passive cation exchange. Bryophytes have high cation exchange capacity (CEC) due to polyuronic acids in their cell walls. Once ions are bound on exchange sites, a proton pump removes H+ ions from the cell, creating a charge gradient that brings in positive ions. These bring along negative ions by cotransport. It appears that bryophytes have two, perhaps more, types of exchange sites, permitting differential binding of ions. They also seem to have specificity for things they need over things they do not. Anion exchange sites can contribute to phosphate uptake. Abundance of cation sites compared to anion sites can account for the preference of ammonium (cation) over nitrate (anion).

Further active processes are able to distinguish ions formed by N, P, and K from more exchangeable cations such as those of Ca++ or Mg++, and they are generally able to maintain relatively constant levels of these essential nutrients despite changes in environmental concentrations. Increasing temperatures increase the uptake, which is also pH-dependent. Some uptake occurs through pinocytosis and entry of nanoparticles.

Fungi are often associated with the rhizoids of bryophytes. It may be that a large number of bryophytes are afforded the advantages of fungal partner relationships, providing them with considerably more surface area for acquiring nutrients. The thallose liverwort Cryptothallus mirabilis (Tulasnella) has a fungal partner that provides carbohydrates for this non-chlorophyllous plant.

Many bryophyte-fungal associations have been discovered, but the types of interaction lack our understanding. We know that glomalean fungi are frequently associated with bryophytes, but the association has not been clearly described. This could be a very fruitful area for further research.

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