Function of *Nicotiana tabacum* Aquaporins as Chloroplast Gas Pores Challenges the Concept of Membrane CO$_2$ Permeability

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Photosynthesis is often limited by the rate of CO$_2$ diffusion from the atmosphere to the chloroplast. The primary resistances for CO$_2$ diffusion are thought to be at the stomata and at photosynthesizing cells via a combination resulting from resistances of aqueous solution as well as the plasma membrane and both outer and inner chloroplast membranes. In contrast with stomatal resistance, the resistance of biological membranes to gas transport is not widely recognized as a limiting factor for metabolic function. We show that the tobacco (*Nicotiana tabacum*) plasma membrane and inner chloroplast membranes contain the aquaporin Nt AQP1. RNA interference–mediated decreases in Nt AQP1 expression lowered the CO$_2$ permeability of the inner chloroplast membrane. In vivo data show that the reduced amount of Nt AQP1 caused a 20% change in CO$_2$ conductance within leaves. Our discovery of CO$_2$ aquaporin function in the chloroplast membrane opens new opportunities for mechanistic examination of leaf internal CO$_2$ conductance regulation.

INTRODUCTION

The existence of a water-facilitating component in membranes was hypothesized after the discovery that the transepithelial water flow across amphibian skin was ~5 times higher than expected if it was only controlled by simple diffusion across a lipid bilayer (Hevesy et al., 1935). Aquaporins were identified as this component in the early nineties (Preston et al., 1992; Agre, 2004). Since then, these proteins were identified in almost all organisms. Aquaporins display variable conductivity for water, in some cases along with permeability to solutes such as glycerol, urea, amino acids, and even ions (Biela et al., 1999; Eckert et al., 1999; Yasui et al., 1999; Ikeda et al., 2002). Furthermore, some aquaporins were found to facilitate gas transport through biomembranes in heterologous expression systems (Nakhoul et al., 1998; Jahn et al., 2004).

Within plant membranes, Uehlein et al. (2003) provided evidence for a protein-mediated pathway for CO$_2$ transport in vivo by altering the expression of the aquaporin 1 from *Nicotiana tabacum* (Nt AQP1). The Nt AQP1 protein belongs to the PIP1 subfamily (plasma membrane intrinsic proteins) and shares functionally important amino acid residues in the pore region with human AQP1 (de Groot et al., 2001). High expression of Nt AQP1 resulted in increased photosynthesis and growth. However, the precise mechanism of action and mechanistic relation to photosynthesis was uncertain (Uehlein et al., 2003).

In many cases, photosynthesis is limited by the availability of CO$_2$, which is dependent on the process of diffusion from the bulk atmosphere to the site of photosynthetic CO$_2$ fixation in the chloroplast stroma. There are three commonly defined resistances in this diffusion pathway: the leaf boundary layer, the stomata, and the internal leaf structure. Over the past 30 years, speculation about the magnitude and variability of resistance to CO$_2$ diffusion inside leaves ($r_l$) has ranged from assumptions that it is insignificant to that it is large and variable. The importance of characterizing $r_l$ for improving models of photosynthesis has recently been revisited (Ethier et al., 2006; Warren and Adams, 2006), though the mechanism of its variability remains poorly understood. Internal leaf resistance was subdivided into resistance within the intercellular air space, liquid phase resistance in cell walls and cytosol, and resistance of biological membranes (Evans et al., 1994; Gillon and Yakir, 2000).

Because they have been difficult to measure, membrane resistances cause the greatest uncertainties. Using measurements of the CO$_2$ permeability of a lecithin-cholesterol bilayer, and assuming an equal resistance for each of the three membranes in the diffusive pathway from the air spaces to the chloroplast stroma, Evans et al. (1994) calculated that the plasma membrane and the chloroplast outer and inner membranes together account for 49% of $r_l$. However, not all three membranes may have equal permeability.

Developmentally controlled morphological changes in leaves, such as the amount of surface area exposed to intercellular air spaces, are commonly invoked to explain differences in $r_l$ (Evans et al., 1994; Evans and Loreto, 2000). The hypothesis that protein expression may reduce $r_l$ was initially proposed as a function of carbonic anhydrase, as it could maintain equilibrium CO$_2$ concentrations adjacent to membranes (Evans et al., 1994; Price...
et al., 1995; Gillon and Yakir, 2000; Bernacchi et al., 2002). This helps to control ribulose-1,5-bis-phosphate carboxylase/oxygenase (Rubisco), it must cross the plasma membrane and chloroplast membranes. An obvious assumption is that Nt AQP1 fulfils its function as a CO2 transport facilitator in the plasma membrane since it belongs to the PIP1 family of plant plasma membrane intrinsic proteins (Biela et al., 1999). However, plasma membrane CO2 permeability only varies slightly with the level of Nt AQP1 expression (see results below). Consequently, the chloroplast membranes are another possible location for aquaporin-mediated modification of CO2 flux. To date, we know of no reports describing aquaporins in organelles apart from mitochondria in animals (Amiry-Moghaddam et al., 2005; Calamita et al., 2006). However, no conclusive proof of organelle aquaporin function was provided (Yang et al., 2006).

Here, we show that Nt AQP1 is localized in chloroplast membranes in addition to plasma membranes, and we give evidence for its function in CO2 transport in isolated chloroplasts in situ, in membrane vesicles, and in vivo using mature leaves. By demonstrating the CO2 (rather than H2O) transporting function for this
organelle located aquaporin, we call into question the appropri-
ateness of referring to these proteins as aquaporins and chal-
lenge the concept of free diffusion of gases like CO₂ through
biomembranes.

RESULTS

Localization of Nt AQP1

The subcellular localization of Nt AQP1 in chloroplast membranes
was determined by protein gel blot analysis, electron micros-
copy, and fluorescence microscopy using an Nt AQP1-mGFP (for
modified green fluorescent protein) fusion protein. For protein gel
blotting, chloroplast membranes were isolated and separated
into outer, inner, and thylakoid membranes. The identity of
envelope membranes was verified with antibodies directed to
the phosphate translocator in the inner chloroplast membrane
and the 24-kD outer chloroplast membrane protein, respectively
(see Supplemental Figure 1 online). Using an antibody directed to
the N terminus of Nt AQP1, we confirmed that this aquaporin is a
constituent of plasma membranes. In addition, a clear signal
corresponding to the inner chloroplast membrane was detected.

Figure 3. Detection of a Translational Fusion of Nt AQP1 to GFP in Tobacco Chloroplasts after Transient Transformation Using the Biolistic Method.
Panels (A) to (C) show intact guard cells (bar = 20 μm), and (D) to (F) show mesophyll cells (bar = 25 μm). Bright-field images (A and D), chlorophyll
fluorescence (B and E; red), and mGFP fluorescence (C and F; green) are shown.
the plasma membrane and in regions of chlorophyll fluorescence. Taken together, this supports the notion that in mesophyll cells and in guard cells, Nt AQP1 was targeted to the chloroplast in addition to the initially discovered localization in plasma membranes.

**CO₂ Permeability of Isolated Membranes**

Nt AQP1 function in green leaf cell membranes was analyzed with regard to water and CO₂ permeability. Plasma membrane or chloroplast membrane vesicles were loaded with carboxyfluorescein and carbonic anhydrase, and, in a stopped flow spectrophotometer, the suspension was subjected to a CO₂-saturated buffer solution. As a consequence of CO₂ uptake and conversion into carbonic acid by carbonic anhydrase, the cell acidified and the kinetics of the resulting fluorescence decrease were recorded. Using these data, CO₂ permeability could be calculated and revealed just slight differences in plasma membranes isolated from leaf cells of control plants (8.54*10⁻³ ± 1.71*10⁻⁴ cm/s [n = 15]) or those of plants lacking Nt AQP1 from disruption via RNA interference (RNAi) (7.73*10⁻³ ± 2.95*10⁻⁴ cm/s [n = 15]; t test, P = 0.1). The plasma membrane water permeability changed, and it was 6.34*10⁻³ ± 1.47*10⁻⁴ cm/s for controls (n = 20) and 3.39*10⁻³ ± 9.72*10⁻⁵ cm/s for RNAi plants (n = 20; t test, P = 3.4*10⁻¹⁰). However, the water permeability of chloroplast envelopes was not significantly different (i.e., 1.91*10⁻² ± 6.27*10⁻⁴ cm/s [n = 25] for controls and 1.70*10⁻² ± 6.34*10⁻⁴ cm/s [n = 40] for RNAi plants; t test, P = 0.22). By contrast, the CO₂ permeability of chloroplast envelopes with or without AQP1 differed (Figure 4A). It was 18.5*10⁻⁴ ± 4.06*10⁻⁴ cm/s (n = 22) for control and 2.06*10⁻⁴ ± 4.90*10⁻⁵ cm/s (n = 28) for RNAi plants (t test, P = 1.89*10⁻⁵). This corresponds to an 89% reduced CO₂ uptake rate compared with chloroplast vesicles with a natural expression of Nt AQP1. Comparison of Pₐ₃ data from chloroplast envelope and plasma membranes indicate that the plasma membrane is ~5 times more permeable to CO₂ than the chloroplast membrane (Figure 4B).

**Photosynthetic Capacity**

We also examined the influence of chloroplast envelope CO₂ permeability on the photosynthetic capacity of leaves from controls and Nt AQP1–deficient RNAi plants. During illumination, the leaf internal CO₂ partial pressure (ci) was similar in RNAi plants and control plants. However, photosynthetic rates were reduced

<table>
<thead>
<tr>
<th>A (µmol m⁻² s⁻¹)</th>
<th>gₛ (mol m⁻² s⁻¹)</th>
<th>cᵢ (Pa)</th>
<th>A/cᵢ (µmol m⁻² s⁻¹ Pa⁻¹)</th>
</tr>
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<tbody>
<tr>
<td>Control</td>
<td>19.1 ± 1.0±</td>
<td>0.49 ± 0.01±</td>
<td>22.4 ± 0.4±</td>
</tr>
<tr>
<td>RNAi</td>
<td>16.2 ± 0.8±</td>
<td>0.38 ± 0.03±</td>
<td>22.2 ± 0.4±</td>
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Net photosynthesis (A), stomatal conductance to water vapour (gₛ), and intercellular CO₂ partial pressure (cᵢ) in AQP1 RNAi and control tobacco plants. Measurements reported at an atmospheric pressure of ~100 kPa and a light intensity of 1500 µmol m⁻² s⁻¹. Different letters indicate significant differences (t test, P < 0.05), and se is shown as ± (n = 12).
by 15% in the former (Table 1), confirming our findings on Nt AQP1 antisense plants (Uehlein et al., 2003). The inhibition of AQP1 expression also resulted in a slight reduction of stomatal conductance by limiting uptake of CO2 into leaves. However, the ratio of net assimilation rate (A) to c, was significantly higher in controls (Table 1). As Flexas et al. (2006) showed that tobacco plants with impaired expression of AQP1 exhibit the same amount and activity of Rubisco, the change in A/c, points to a change of mesophyll conductance.

**Leaf Internal Conductance**

In a separate experiment, we combined leaf gas exchange with tunable diode laser spectroscopy (TDL) to perform online measurements of carbon isotope discrimination (Δ) by leaves (Flexas et al., 2006; Barbour et al., 2007). Following the procedure given by Evans et al. (1986), and Farquhar (Farquhar et al., 1982; Farquhar and Richards, 1984), we calculated internal leaf conductance to CO2 (g, = 1/r) and the CO2 partial pressure at the site of carboxylation (c,). Plants for this experiment were grown in New Mexico at higher light intensities (due to greenhouse characteristics) and lower ambient partial pressures of CO2 (due to higher elevation above sea level) than the plants for the previously described gas exchange experiment performed in Darmstadt, Germany.

At the assay light intensity of 2000 μmol photons m⁻² s⁻¹, A was lower in the RNAi line, though only significant as a trend (P = 0.10), while g, and c, did not differ significantly between control and RNAi lines (Table 2). The ratio of A to c, in New Mexico–grown wild-type plants was ∼25% higher than that of the wild-type plants grown in Germany, suggesting that the New Mexico plants either had a higher photosynthetic capacity (greater amounts or more active Rubisco) or a higher g,.

Application of these data to the Evans et al. (1986) model, we calculated that the mesophyll conductance for CO2 (g, in the RNAi line is 21% lower (or r, is 27% higher) than for the control line. Since c, = c, / (A/g), this equates with a 1.7 Pa (13%, P = 0.16) lower c, in the RNAi line (Table 2). Lower rates of photosynthesis in the RNAi line have the effect of increasing the calculated c, so it is not surprising that the c, decrease is only a trend.

**Membrane Resistances as a Percentage of Total Resistance**

In combination, our data show that the purified inner chloroplast membranes from RNAi plants have a 56% higher resistance to CO2 transport, but in vivo the entire r, only increased 27%. Therefore, if we assume that the changes in the Nt AQP1 content of the inner chloroplast membrane are the sole factor generating the change in r, we would estimate that the resistance of the inner chloroplast membrane constitutes 48% of r, in tobacco rather than the 16% estimated by Evans et al. (1994) for tobacco.

**DISCUSSION**

Tobacco AQP1 belongs to the so-called Plasma Membrane Intrinsic Protein family 1 (PIP1). Our data show that the classification with regard to plant aquaporin localization is insufficient because the cellular distribution of this aquaporin is not restricted to the plasma membrane. In addition to its location in the plasma membrane, Nt AQP1 is also a component of the inner chloroplast membrane even though it lacks a classical chloroplast transit peptide. We could demonstrate that Nt AQP1 in chloroplast

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**Table 2. Gas Exchange Analysis and Online Photosynthetic Discrimination**

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<thead>
<tr>
<th>A (μmol m⁻² s⁻¹)</th>
<th>g, (mol H₂O m⁻² s⁻¹)</th>
<th>c, (Pa)</th>
<th>Δ (<em>g</em> yüzde)</th>
<th>g, (μmol CO₂ m⁻² s⁻¹ Pa⁻¹)</th>
<th>c, (Pa)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Control</td>
<td>20.7 ± 0.4a</td>
<td>0.66 ± 0.07a</td>
<td>19.6 ± 0.5a</td>
<td>17.0 ± 0.4a</td>
<td>3.0 ± 0.3a</td>
</tr>
<tr>
<td>RNAi</td>
<td>19.6 ± 0.7a</td>
<td>0.56 ± 0.05a</td>
<td>19.4 ± 0.7a</td>
<td>16.3 ± 0.7a</td>
<td>2.4 ± 0.2a</td>
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Net photosynthesis (A), stomatal conductance to water vapor (g,), intercellular CO₂ partial pressure (c,), online photosynthetic ΔC₁³CO₂ discrimination (Δ), internal leaf conductance (g,), and chloroplast CO₂ partial pressures (c,) are reported for AQP1 RNAi and control tobacco plants. All measurements were made at a PPFD of 2000 μmol m⁻² s⁻¹, except for g, which was calculated from measurements at multiple light intensities. Measurements are reported at an atmospheric pressure of ~79 kPa, and leaves were provided with a c, of ~25.1 Pa at a light intensity of 2000 μmol m⁻² s⁻¹, except g, data, which were generated from multiple light intensities. Values reported for g, can be converted to the more familiar, non-SI units of mol CO₂ m⁻² s⁻¹ Pa⁻¹ by dividing by 10. Different letters indicate significant differences (t test, P < 0.05), and SE is shown as ± (n = 6).
envelopes fulfills an important physiological function in vivo by showing that CO₂ transport through the plant leaf to the site of CO₂ fixation in the chloroplast stroma is strongly impacted by the function of Nt AQP1 as a CO₂ membrane transport facilitator.

Plants with reduced Nt AQP1 expression have an increased chloroplast membrane resistance to CO₂ transport of 56%, which is manifested as a 27% increase of in vivo internal leaf resistance. This causes a reduced rate of photosynthesis in the RNAi line, and, despite this lower sink strength, the chloroplast CO₂ partial pressure in this RNAi line is lower than that in controls. Reduced expression of Nt AQP1 has effects on the characteristics of membranes in chloroplast-containing mesophyll cells. The plasma membrane showed decreased water permeability, while the CO₂ permeability was not considerably affected. In chloroplast envelopes, the water permeability was just slightly reduced, while the CO₂ permeability decreased significantly.

Thus, Nt AQP1 seems to switch function from a water transport facilitating channel to a CO₂ transport facilitating channel depending on its cellular location or on the intrinsic CO₂ permeability of the membrane where it resides. Comparative analysis of the CO₂ permeability properties indicates that the plasma membrane is ~5 times more permeable to CO₂ than the chloroplast envelope. Thus, the chloroplast envelope could potentially meet the requirements for CO₂ permeation through aquaporins in a membrane with low CO₂ permeability as demanded by Hub and de Groot (2006). The concept that certain membranes could be more mosaic than fluid would support this view (Engelman, 2005) and is consistent with the concept of Nt AQP1 being a CO₂ pore.

It is known that aquaporin function can be regulated by posttranslational modification (e.g., via phosphorylation) or by interaction with other aquaporin isoforms (Johansson et al., 1998; Moshelion et al., 2004). Although phosphorylation or regulation by phosphorylation was not demonstrated for PIP1-type aquaporins like Nt AQP1, it is possible that one of the above-mentioned regulatory mechanisms causes the change in transport specificity. Besides aquaporin regulation, simple differences in intrinsic membrane properties between the plasma membrane and chloroplast membranes could explain the assumed switch in aquaporin function. If the intrinsic CO₂ permeability of plant plasma membranes is large enough to ensure high enough rates of CO₂ transport, the presence or absence of CO₂ pores may not have a measurable effect. Theoretical studies have demonstrated that significant aquaporin-mediated CO₂ permeation is only relevant in membranes with a low intrinsic CO₂ permeability (Hub and de Groot, 2006). As confirmed by our studies in membrane vesicles and in CO₂ uptake measurements in vivo, the chloroplast envelope lacking Nt AQP1 appears to be rate limiting for CO₂ transport.

It was shown that an unstrirred layer could affect fast membrane transport processes in vesicles of artificial bilayers, and the question could arise if these could also be rate limiting in experiments with cell membranes (Gutknecht et al., 1977; Hill et al., 2004). Because we detect clear differences in membrane CO₂ permeability with or without Nt AQP1 and since all CO₂ transport experiments were conducted under iso-osmotic conditions, it is unlikely that a membrane water layer, which could be assumed to have the same thickness in membrane vesicles from RNAI or controls, is the rate limiting step. Determination of P CO₂ values revealed that CO₂ permeates the chloroplast membrane ~100 times slower than estimated for an artificial bilayer. The absolute values for CO₂ transport rates are in perfect agreement with those obtained on human aquaporin1 by a comparable experimental setup (Prasad et al., 1998). The determined CO₂ permeability is not in the range where unstirred layers significantly affect CO₂ transport; it is rather in the range of that for water. Consequently, it is suggested that the contribution of Nt AQP1 to CO₂ permeability is as significant as that of water-conducting aquaporins to membrane water permeability. Taken together, we conclude that Nt AQP1 as a CO₂ facilitator increases CO₂ transport at the inner chloroplast membrane, not at the plasma membrane.

The localization of Nt AQP1 in chloroplast membranes (in addition to the plasma membrane) suggests that its classification and possibly that of other PIP aquaporins as plasma membrane intrinsic protein is misleading. Also, the demonstrated CO₂ transporting function in addition to water transport makes the term “CO₂ aquaporin” or “COOporin,” as it was already suggested by Terashima et al. (2006) more appropriate than aquaporin. The physiological relevance of this gas transporting function also suggests a mechanism that plants may use to modify photosynthetic function. Furthermore, the possibility that a CO₂ transport function or gas transport function in general may be important for mitochondrial membranes in many different species, including animals, has not escaped our notice.

**METHODS**

**Plant Material**

Tobacco plants (*Nicotiana tabacum* cv Petit Havana SR1) were cultivated under standard greenhouse conditions in Germany (25°C, 80% RH, PAR ~800 μmol m⁻² s⁻¹, ~36.9 Pa CO₂, 14 h light) and in New Mexico (26°C, 20% RH, max PAR ~1200 μmol m⁻² s⁻¹, ~28.8 Pa CO₂, 14 h light). The RNAi tobacco plants with reduced expression of Nt AQP1 were a gift from M. Bots and C. Mariani. The production of the transgenic plants was described elsewhere (Bots, 2004). Existence of the RNAI effect was verified on the protein level using a PIP1-specific antiserum kindly provided by C. Mariani and an Nt AQP1–specific antiserum (Otto and Kaldenhoff, 2000; see Supplemental Figure 2 online).

**Purification of Plasma Membranes**

Prior to purification of plasma membranes, crude membrane fractions (microsomes) were isolated following the procedure of Kjellbom and Larsson (1984). Briefly, 100 g fresh weight leaf material were homogenized three times for 30 s in 300 mL 0.33 M sucrose using a kitchen blender, 50 mM HEPES/KOH, pH 7.5, 5 mM EDTA, 5 mM DTT, 5 mM ascorbic acid, 0.5 mM phenylmethanesulfonyl fluoride, 0.2% BSA, 0.2% caseine (boiled for 10 min), and 0.6% polyvinylo polypyrrolidone. The homogenate was filtered through three layers of miracloth, and the bulk of chloroplasts and mitochondria were sedimented by centrifugation at 5000 g for 10 min. The membranes in the supernatant were precipitated at 100,000 g for 1 h, and the pellet was resuspended to a total volume of 10 mL in 0.33 M sucrose, 5 mM HEPES/KOH, pH 7.5, 5 mM KCl, 1 mM DTT, and 0.1 mM EDTA. Plasma membranes were purified from the crude membrane fraction using the two-phase partitioning system (Widell and Larsson, 1981; Lundborg et al., 1981). Briefly, 9 g of the microsomal suspension were added to 27 g of the phase mixture to give a total phase system of
36 g with a final composition of 6.2% (w/w) Dextran T 500, 6.2% (w/w) polyethylene glycol 3350, 0.33 M sucrose, 5 mM potassium phosphate buffer, pH 7.8, and 5 mM KCl, 1 mM DTT, and 0.1 mM EDTA. The phase system was mixed by several inversions and centrifuged in a swing-out rotor for 10 min at 1500g. The upper phase was reextracted three times. The plasma membrane–enriched final upper phase was diluted fivefold with buffer and centrifuged at 100,000g for 1 h. The pellet was resuspended in 3 mM HEPES/KOH, pH 7.5, 0.25 M sucrose, 50 mM KCl, and 1 mM DTT.

**Protein Gel Blotting**

Ten micrograms of protein per lane were separated on a 12% polyacrylamid gel (SDS-PAGE) (Laemmli, 1970). Equal lane loading was monitored by Coomassie Brilliant Blue stain in a parallel experiment. Protein transfer was performed in a tank transfer system (Amersham) with 10 mM CAPS to a nitrocellulose membrane for 2 h at 80 V. Transfer efficiency was monitored by a reversible, colloidal silver stain. For this, the membrane was incubated in a solution containing 2% sodium citrate, 0.8% FeSO4 heptahydrate, and 0.2% AgNO3 for 10 min and washed with water. The membrane was destained with 15 mM potassium hexacyanoferrat (III) and 50 mM Na-thiosulfate. Subsequently, it was blocked with fat-free milk powder in PBS (137 mM NaCl, 2.7 mM KCl, 10 mM Na2HPO4, and 2 mM KH2PO4, pH 7.4) for 15 min and incubated with the respective antibodies. Detection was achieved via a chemiluminescent substrate according to the protocol provided by the manufacturer (CDP-Star; Applied Biosystems).

**Electron Microscopy and Immunocytochemistry**

Electron microscopy was done essentially following the procedure given in Kaldenhoff et al. (1995). Briefly, tissues were fixed in 2% glutaraldehyde, buffered phosphate, pH 7.2, for 60 min at 4°C. Specimens were washed with ice-cold phosphate buffer and dehydrated in a graded acetone series and embedded in EM resin (TAAB). Sections were cut with a diamond knife on an ultramicrotome. Immunostaining was performed by treating the sections with 1% BSA in 20 mM phosphate buffer, pH 7.2, for 60 min at 4°C. Tobacco leaves were washed with distilled water and homogenized using a modified blender (Kannangara et al., 1977) in ice-cold grinding buffer (300 mM sorbitol, 1 mM sodium pyrophosphate, 2 mM EDTA, pH 8.0, 5 mM MgCl2, 5 mM 2-mercaptoethanol, 5 mM ascorbate, and 50 mM HEPES, pH 6.8). The homogenate was filtered through four layers of miracloth and centrifuged at 4000 rpm in a Sorvall GSA rotor. The supernatant was discarded and the pellet resuspended in 5 mL of ice-cold grinding buffer and layered on top of a two-step Percoll gradient (80 and 40% percoll in grinding buffer). Centrifugation at 5750 rpm in an Sorvall HB-4 rotor for 20 min resulted in separation of intact chloroplasts from residual starch granules, mitochondria, broken chloroplasts, and other cellular membranes. Chloroplasts were washed three times by careful resuspension in ice-cold grinding buffer and centrifugation for 1 min at 3000 rpm in a Sorvall HB-4 rotor. Purified chloroplasts were carefully resuspended in 0.6 M sorbitol in Tricine-EDTA buffer (10 mM Tricine and 1 mM EDTA, pH 7.0) to cause shrinkage for 30 min. Chloroplasts were subjected to a freeze-thaw cycle. After homogenization, thylakoid membranes were removed by centrifugation at 2000g. The sorbitol concentration in the supernatant was diluted to 0.3 M with Tricine-EDTA buffer. Envelope membrane vesicles were collected from the supernatant by centrifugation on a three-step sucrose gradient (0.8, 0.64, and 0.4 M in Tricine buffer) at 113,000g for 3 h. Verification of membrane fraction identity was done using antibodies directed to phosphate translocator in inner or 24-kd outer chloroplast membrane protein from spinach (Spinacia oleracea; kindly provided by U.I. Flügge, University of Cologne, Germany).

**Purification of Chloroplast Envelope Membrane Vesicles**

Prior to the isolation of chloroplasts, plants were incubated in darkness for 24 h to reduce the starch content of the chloroplasts. Intact chloroplasts were isolated from tobacco using a Percoll gradient based method as described by Cline (1985) with modifications by Bock (1998) in buffer containing sorbitol as osmoticum. All steps were performed at 4°C. Tobacco leaves were washed with distilled water and homogenized using a modified blender (Kannangara et al., 1977) in ice-cold grinding buffer (300 mM sorbitol, 1 mM sodium pyrophosphate, 2 mM EDTA, pH 8.0, 5 mM MgCl2, 5 mM 2-mercaptoethanol, 5 mM ascorbate, and 50 mM HEPES, pH 6.8). The homogenate was filtered through four layers of miracloth and centrifuged at 4000 rpm in a Sorvall HB-4 rotor. The supernatant was discarded and the pellet resuspended in 5 mL of ice-cold grinding buffer and layered on top of a two-step Percoll gradient (80 and 40% percoll in grinding buffer). Centrifugation at 5750 rpm in an Sorvall HB-4 rotor for 20 min resulted in separation of intact chloroplasts from residual starch granules, mitochondria, broken chloroplasts, and other cellular membranes. Chloroplasts were washed three times by careful resuspension in ice-cold grinding buffer and centrifugation for 1 min at 3000 rpm in a Sorvall HB-4 rotor. Purified chloroplasts were carefully resuspended in 0.6 M sorbitol in Tricine-EDTA buffer (10 mM Tricine and 1 mM EDTA, pH 7.0) to cause shrinkage for 30 min. Chloroplasts were subjected to a freeze-thaw cycle. After homogenization, thylakoid membranes were removed by centrifugation at 2000g. The sorbitol concentration in the supernatant was diluted to 0.3 M with Tricine-EDTA buffer. Envelope membrane vesicles were collected from the supernatant by centrifugation on a three-step sucrose gradient (0.8, 0.64, and 0.4 M in Tricine buffer) at 113,000g for 3 h. Verification of membrane fraction identity was done using antibodies directed to phosphate translocator in inner or 24-kd outer chloroplast membrane protein from spinach (Spinacia oleracea; kindly provided by U.I. Flügge, University of Cologne, Germany).

**Purification of Chloroplast Envelope Membrane Vesicles**

Vesicles were suspended in a buffer solution containing 50 mM KCl, 50 mM NaCl, and 20 mM HEPES, pH 7.0, supplemented with 50 μM carboxyfluorescein and 1 mg/mL carbonic anhydrase and incubated at 4°C overnight. Carboxyfluorescein– and carbonic anhydrase–loaded vesicles were pelleted by centrifugation (100,000g, 30 min) and washed once with buffer aerated with N2. Vesicles loaded with carbonic anhydrase and fluorescein were rapidly mixed with the above-mentioned buffer solution aerated with CO2 in a stopped flow spectrometry device (SMF-300; Bio-Logic Scientific Instruments). Uptake of CO2 resulted in an intravesicular acidification and consequently a decrease of fluorescein fluorescence. To calculate membrane CO2 permeability, the intravesicular pH was assessed prior to CO2 uptake and after the reaction was completed following the procedure given by Slavik (1982). For this, the ratio of pH-independent to pH-dependent fluorescein fluorescence (435 nm versus 490 nm) at different pH values was determined. The resulting calibration curve (pH/ratio of fluorescence intensity) was used to calculate CO2 concentrations at the inlet and outlet of the cuvette were measured using a differential infrared gas analyzer (IRGA). The cuvette flow was adjusted to 200 mL min⁻¹. The cuvette area was 4.5 cm², and white light of 500 to 1500 μmol m⁻² s⁻¹ PPF was used. Leaf temperature was kept at 25°C. Measurements were made on the youngest fully expanded leaves. Data were recorded at 10-s intervals. Data analysis and calculations were performed using Microsoft Excel. Statistically significant differences between plants with reduced Nt AQP1 expression and control plants were analyzed using a Student’s t test (P < 0.05). Data are given as means ± SE.
the intracellular pH from the ratio of fluorescence intensity as measured prior to and after the reaction. Consequently, the kinetics in fluorescence change as detected by the stopped flow spectrophotometer could be converted into a pH-change kinetics. The exponential time constant of the acidification was determined over the initial 100 ms. CO₂ permeability was calculated using the method given by Yang et al. (2006). The average vesicle diameter was 276 and 365 nm for plasma membrane and chloroplast envelope vesicles, respectively, giving surface-to-volume ratios of 2.17 × 10⁵ cm⁻¹ and 1.64 × 10⁵ cm⁻¹. Vesicle size was determined using a Zetasizer system (Malvern Instruments).

Analysis of H₂O Permeability of Membrane Vesicles

Water permeability of membrane vesicles was analyzed by measuring the intensity of light scattering following an osmotic challenge as described by Maurel et al. (1997). Purified plasma membrane vesicles were diluted to a concentration of 100 μg protein·mL⁻¹ in a hypotonic solution (50 mM NaCl, 50 mM mannitol, and 10 mM Tris-MES, pH 8.3), which induced a transient opening of vesicles and equilibration of the intravesicular material with the buffer solution. The vesicles were mixed with an equal volume of the same solution as used for equilibration supplemented with 500 mM mannitol in an SFM 300 stopped flow spectrometer (Bio-Logic Scientific Instruments). Vesicle shrinkage was monitored via measurement of 90° scattered light intensity increase.

Estimation of gₙ and cₑ

Online measurements of photosynthetic ¹³CO₂ discrimination from a combined IRGA-tunable diode laser system were used to calculate gₙ and cₑ using the equations of Evans et al. (1986). The TGA-100 (Campbell Scientific) measures absolute concentrations of ¹³CO₂ and ¹²CO₂ at a frequency of 10 Hz from dry air before and after exposure to a photosynthesising leaf. The 10 Hz data were averaged over 15 s every 3 min (between calibrations) to calculate the isotopic composition (δ¹³C) of sampled air with a precision of 0.05 to 0.09‰. Measurements of photosynthesis were made using IRGAs in an LI-6400 portable gas exchange system with a 6 cm² leaf chamber and a red/blue LED light source (LI-COR). All data except δg were reported for a light intensity of 2000 μmol m⁻² s⁻¹ PPFD. Estimates of gₙ were generated from online measurements of Δ made at light intensities ranging from 500 to 2000 μmol m⁻² s⁻¹ PPFD to vary the ratio of A to cₑ (ambient partial pressure of CO₂). We report the units of gₙ per Pascal (μmol m⁻² s⁻¹ Pa⁻¹) rather than the traditional method that uses non-SI units of per bar (mol m⁻² s⁻¹ bar⁻¹). Conversion to the per bar values is achieved by dividing per Pa data by 10. Leaf temperature was kept at 25°C, and leaves were provided with 30 Pa CO₂ with a δ¹³C of approximately −4‰. Measurements were made on the youngest fully expanded leaves.

We calculated cₑ using the models of photosynthetic discrimination (Δ) presented by Farquhar et al. (Farquhar, 1982; Farquhar and Richards, 1984). The simple model describing predicted discrimination (Δ) is Δ = a(Δg) + bΔg, where cₑ, cₑ, and cₑ are the ambient (well mixed air surrounding the leaf), leaf surface, and intercellular partial pressures of CO₂, respectively, a(Δg) is the fractionation occurring from diffusion through the leaf boundary layer (2.9‰), a is the fractionation occurring from diffusion in air (4.4‰), and b is the net fractionation caused by Rubisco and PEPC (29‰, for tobacco) (Evans et al., 1994). In the simple model, cₑ is related to Δ by making the additional assumption that gₙ is infinite (cₑ = cₑ). The slope of the plot of Δ versus (A/cₑ) (see Figure 5 for an example) is inversely proportional to gₙ(Evans et al., 1986). We used the relationship cₑ = cₑ − (A/gₙ) to calculate CO₂ partial pressure at the chloroplast. Statistically significant differences were analyzed using a Student’s t test (P < 0.05). Data are given as means ± SE.

Accession Numbers

Sequence data of genes appearing in this article can be found in the GenBank/EMBL data libraries under accession numbers AJ001416 (Nt AQP1), NM_198098 (human AQP1), and SCU87624 (mGFP4).

Supplemental Data

The following materials are available in the online version of this article.

Supplemental Figure 1. Identification of Chloroplast Envelope Proteins by Immunoblot.

Supplemental Figure 2. Nt AQP1 in RNAi and Control Plants.

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Function of *Nicotiana tabacum* Aquaporins as Chloroplast Gas Pores Challenges the Concept of Membrane CO₂ Permeability

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