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Submitted on 15 Mar 2021

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Novel Aquaporin Regulatory Mechanisms Revealed by Interactomics*§

Jorge Bellati‡, Chloé Champeyroux‡, Sonia Hem‡, Valérie Rofidal‡, Gabriel Krouk‡, Christophe Maurel‡, and Véronique Santoni‡§

PIP1;2 and PIP2;1 are aquaporins that are highly expressed in roots and bring a major contribution to root water transport and its regulation by hormonal and abiotic factors. Interactions between cellular proteins or with other macromolecules contribute to forming molecular machines. Proteins that molecularly interact with PIP1;2 and PIP2;1 were searched to get new insights into regulatory mechanisms of root water transport. For that, a immuno-purification strategy coupled to protein identification and quantification by mass spectrometry (IP-MS) of PIPs was combined with data from the literature, to build thorough PIP1;2 and PIP2;1 interactomes, sharing about 400 interacting proteins. Such interactome revealed PIPs to behave as a platform for recruitment of a wide range of transport activities and provided novel insights into regulation of PIP cellular trafficking by osmotic and oxidative treatments. This work also pointed a role of lipid signaling in PIP function and enhanced our knowledge of protein kinases involved in PIP regulation. In particular we show that 2 members of the receptor-like-kinase (RLK) family (RKL1 (At1g48480) and Feronia (At3g51550)) differentially modulate PIP activity through distinct molecular mechanisms. The overall work opens novel perspectives in understanding PIP regulatory mechanisms and their role in adjustment of plant water status. Molecular & Cellular Proteomics 15: 10.1074/mcp.M116.060087, 3473–3487, 2016.

The absorption of soil water by roots is crucial for plants to maintain their water status. Studies in various plant species have shown that the root water permeability (root hydraulic conductivity; \(L_p\)) is constantly adjusted depending on the developmental stage of the plant, its nutritional or hormonal status, or multiple environmental stimuli (1, 2). These adjustments depend in large part on the function and regulation of aquaporins, a large class of channel proteins that facilitate the diffusion of water and small neutral solutes across cell membranes (2, 3). Aquaporins are 25–30 kDa proteins with 6 membrane-spanning domains and five connecting loops (A–E), the N- and C-terminal tails being exposed to the cytosol (4). Plant aquaporins show a high multiplicity of isoforms. Thirty-five homologs comprised in four homology subclasses have been identified in Arabidopsis. The plasma membrane intrinsic proteins (PIPs) (with 13 isoforms further subdivided in the PIP1 and PIP2 subgroups), and the tonoplastic intrinsic proteins (TIPs) (with 10 homologs) are the most abundant aquaporins in the plasma membrane and the tonoplast, respectively (5, 6). Two other subclasses include nodulin-26-like proteins (NIPs) and small basic intrinsic proteins (SIPs), with nine and three homologs, respectively (5–7).

The response of plant roots to environmental and hormonal stimuli is mediated through long-term transcriptional control of aquaporin functions, together with multiple post-translational mechanisms, such as phosphorylation, that affect the activity of aquaporins, their targeting to their destination compartment, or their stability. PIP aquaporins show a conserved phosphorylation site in their first cytosolic loop (loop B) and, in the case of PIP2 isoforms, multiple phosphorylations in adjacent sites of their C-terminal tail (1), PhosPhAt database (http://phosphat.mpimp-golm.mpg.de/). Aquaporin phosphorylation is a significant component of plant responses to stresses. For instance, following exposure of Arabidopsis roots to salt (NaCl) or hydrogen peroxide (H\(_2\)O\(_2\)), AtPIP2;1 phosphorylation was decreased and increased, respectively (8). A recent quantitative phosphoproteomic work showed a strong correlation between the level of PIP phosphorylation and \(L_p\), under a wide range of environmental conditions (1). However, knowledge of the protein kinases (PKs) that phosphorylate aquaporins is still scarce. Two PKs that phosphorylate SoPIP2;1 at Ser\(^{115}\) and Ser\(^{274}\) have been purified from...
SIRK1 was confirmed to phosphorylate Thr264 and Ser286. A rice RLK (LP2, a cytosolic cell abscisic acid (ABA) signaling, was able to phosphorylate (OST1)/Snf1-related PK 2.6 (SnRK2.6), a central PK in guard spinach (9). A very recent work revealed that Open stomata 1 Plant Aquaporin Interactomics trafficking (25, 26). In addition, the tryptophan-rich sensory interact with syntaxins, a family of proteins involved in vesicle regulation of aquaporin levels through a selective autophagic pathway, physically interact transiently induced by osmotic stress, and that is degraded membrane (22–24). PIP2s were also shown to functionally interact with three PIPs in vivo (14) but the functional role of this interactions remains unknown.

The identification of cellular interaction partners is fundamental for understanding cellular and physiological processes. In recent years, crucial experimental approaches for protein interaction mapping such as yeast two hybrid or split ubiquitin, have begun to unravel the complex interacting networks of plant proteins (15–18). Analysis of protein complexes through immuno-purification–IP followed by mass spectrometry (MS) (19) is also a widely employed technique because of its high throughput and sensitivity. Most importantly, this technique addresses the properties of protein-protein interactions occurring in the plant. However, suitable controls and quantitative proteomics are required to distinguish between bona fide binders and background contaminants (20).

Data on plant aquaporin interactomes are starting to emerge. Yeast-two hybrid (18) and split-ubiquitin (15, 16) studies have identified, about 200 proteins that seem to interact, with a high confidence, with PIP aquaporins (21) for review. In addition, more focused recent studies have revealed that PIPs can functionally interact with several classes of proteins. For instance, PIP1–PIP2 interactions were shown to be required for in planta trafficking of PIP1s to the plasma membrane (22–24). PIP2s were also shown to functionally interact with syntaxins, a family of proteins involved in vesicle trafficking (25, 26). In addition, the tryptophan-rich sensory protein/translocator (TSPO), a multistress regulator that is transiently induced by osmotic stress, and that is degraded through a selective autophagic pathway, physically interact with AtPIP2;7 (27). PIPs also functionally interact with Rma1H1, a membrane-anchor E3 ubiquitin ligase homolog, to regulate aquaporin levels via ubiquitination (28).

One major objective of the present work was to investigate as a whole the plant PIP1;2 and PIP2;1 interactome. A quantitative IP–MS strategy, together with data from available databases, allowed to build an interconnected PIP network of about 900 proteins. Next, we focused on those protein interaction partners (next called interactants) that show a physical interaction with PIPs. We hypothesized that these interactants may provide novel insights into the molecular regulation of PIP aquaporins. Here, we explore novel functional roles of phospholipases D and RLKs. The latter can have opposite effects on aquaporin activity through specific molecular mechanisms.

**EXPERIMENTAL PROCEDURES**

**Biological Materials and Plant Treatments—**Arabidopsis thaliana (Col-0 ecotype) transgenic plants expressing GFP, GFP-PIP2;1, GFP-PIP1;2 under the control of a constitutive 35S promoter were used (29) for proteomic analysis (see below). Arabidopsis seeds were sown in vitro on a MS/2 medium (30) complemented with 1% sucrose, 0.05% MES and 7 g/l agar. The effect of NaCl and H2O2 were studied by bathing plantlets with 100 mM NaCl during 2 h, and 500 μM H2O2 for 20 min, respectively. Additional transgenic plants were used: promAMT1;3::AMT1;3-GFP (31), promPGP4::PGP4-GFP in a pggp background (32), promPGP19::GFP-PGP19 (33). Nicotiana tabacum plants were cultivated in soil for 4–6 weeks (8 h light (120 μmol photons/m²/s, 20 °C, 65% relative humidity).

**Vectors and Constructs—**All constructs were obtained using Gateway cloning technology (Invitrogen) according to the manufacturer’s instructions. The cDNAs of RKL1 (At1g48480), RKL902 (At3g17840), Feronia (At3g51550), PDL6 (At4g35790), PDL1 (At4g11850), and NTL3 (At5g06320) were amplified by PCR using the primers described in supplemental Table S1 followed by a second PCR with the primers AtIB1 or AtIB1’ and AtIB2 or AtIB2’ (supplemental Table S1) allowing the addition of attB recombination sites and cloned into a pDONOR 207 vector using a Gateway® BP Clonase enzyme mix (Invitrogen). Annexin4 clone (At2g38750) was obtained from ARBC (U15576 clone). For FLIM experiments, cDNAs were transferred into binary destination vectors pGWB5 and pGW86 (Dr. Nakagawa, Shimane University, Matsue, Japan) to allow fusion of eGFP at the C- and N terminus of the proteins of interest, respectively, by using a Gateway® LR Clonase enzyme mix (Invitrogen). To fuse mRFP at the C terminus of the proteins, cDNAs were transferred into a pB7WGR2 vector. GFP- and mCherry- tagged PIP2;1 are described in (34, 35). A. tumefaciens strain GV3101 was transformed with the constructs of interest, selected for resistance toward rifampicin (50 mg/l), gentamycin (25 mg/l), and kanamycin (50 mg/l) in the case of pGW86 vector, and for resistance toward spectinomycin (100 mg/l) in the case of pB7WGR2 vector.

**Immunopurification—**IP of GFP-tagged proteins was performed at 4 °C from 9 day-old plantlets. Plantlets were treated with 1% formaldehyde for 15 min under gentle shaking. Formaldehyde was then quenched with the addition of 300 mM glycine under continuous shaking. After for 30 min, plantlets were rinsed twice with phosphate-buffered saline (4 mM KH2PO4, 16 mM Na2HPO4, 115 mM NaCl, pH 7.4). One to two grams of roots were collected, chpped with a scalpel in the presence of 2.1 mg/l fresh weight (FW) of a buffer made of NaCl 300 mM, Triton X-100 1% (w/v), Na deoxycholate 0.5% (w/v), SDS 0.1% (w/v), Tris-HCl 100 mM (pH 8), leupeptin 2 mg/l, EDTA 2 mg/l, and AEF 5% B, and then proteinated. After centrifugation at 10,000 × g for 15 min, the supernatant was again centrifuged in the same conditions. IP was performed from the supernatant with an antibody against GFP using a μMACSTM anti-GFP Microbeads kit (Milenyi Biotec, Paris, France) according to manufacturer’s conditions. Briefly,
sample was incubated at 4 °C for 1 h with a volume equivalent to 35 μg/g FW of an anti-GFP Microbeads solution. The sample was then loaded onto μMACS columns that were previously conditioned with 200 μL of a lysis buffer (NaCl 150 mM, Triton X-100 1% (w/v), Tris-HCl 50 mM pH 8). After 4 washings with 200 μL of a buffer made of NaCl 150 mM and lgepal CA-630 1% (v/v) and an additional washing step with 100 μL of Tris-HCl 20 mM pH 7.5, proteins were eluted with 50 μL of elution buffer (Tris-HCl 50 mM pH 6.8, DTT 50 mM, SDS 1% (w/v), EDTA 1 mM, bromophenol blue 0.005% (v/v), glycerol 10% (v/v)). Formaldehyde fixation was reversed by heating eluted proteins at 100 °C during 20 min.

Protein Digestion—In-solution reductions/alkylations were performed simultaneously with detergent removing by the filter-aided sample preparation protocol (1, 36). These steps were followed by a endolysin-C (Sequencing Grade Modified, Promega, Madison, WI) digestion 3 h at 37 °C followed by a trypsin (Sequencing Grade Modified, Promega) digestion overnight at 37 °C. Peptides were eluted by step elutions with 50 mM ammonium bicarbonate, followed by 50% acetonitrile and then 0.5 M NaCl. Peptides were desalted on C18 columns (Sep-Pak® Vac1C18 cartridge 3cc, Waters, Guyancourt, France). After solvent evaporation, peptides were resuspended in 2% formic acid.

Experimental Design and Statistical Rational for Protein Identification and Quantification by Mass Spectrometry—The protein digests were analyzed using a Q-TOF mass spectrometer (Maxis Impact; Bruker Daltonik GmbH, Bremen, Germany), interfaced with a nano-HPLC U3000 system (Thermo Scientific, Waltham, MA). Samples were concentrated with a pre-column (Thermo Scientific, C18 PepMap100, 300 μm × 5 mm, 5 μm, 100 A) at a flow rate of 20 μL/min using 0.1% formic acid. After preconcentration, peptides were separated with a reversed-phase capillary column (Thermo Scientific, C18 PepMap100, 75 μm × 250 mm, 3 μm, 100 A) at a flow rate of 0.3 μL/min using a two-step gradient (8% to 28% acetonitrile in 40 min, then 28% to 42% in 10 min), and eluted directly into the mass spectrometer. Proteins were identified by MS/MS by information-dependent acquisition of fragmentation spectra of multiple charged peptides. Up to twenty data-dependent MS/MS spectra were acquired in positive ion mode. MS/MS raw data were analyzed using Bruker Compass Data Analysis software (Automatic Engine Version 4.1 (Build 359)) to generate the peak lists. The non-redundant Arabidopsis protein database (TAIR10, version 20110823, 35386 entries, http://www.arabidopsis.org) was locally queried using XTandem search engine (version 2013.09.01; http://www.thegpm.org/tandem/) with the following parameters: trypsin as enzyme, 1 allowed missed cleavage, carbamidomethylation of cysteine as fixed modification and N-terminal acetylation of protein, deamidation of asparagine and glutamine, N-terminal-pyroglutamylamidation of glutamine and glutamate, oxidation of methionine, phosphorylation of serine, threonine and tyrosine, and methylation of glutamate and aspartate as variable modifications. Mass tolerance was set to 10 ppm on full scans and 0.05 Da for fragment ions. Identified proteins were filtered and quantified in the present work together with the reported interactants of these PIP interactants. Protein-protein interaction networks were visualized using Cytoscape (40). Networks were analyzed using the Cytoscape 2.8.2 Molecular Complex Detection (MCODE) plugin (41) to detect clusters (i.e. densely connected regions) that are predictive of functional protein complexes. The parameters used for MCODE to generate the clusters were as follows: loops included, degree cut-off of 2, deletion of single connected nodes from the cluster (haircut option disabled), expansion of the cluster by one neighbor shell (fluff option enabled), node density cut-off of 0.1, node score cut-off of 0.2, k-core of 2, and maximum depth of the network equal to 100.

Fluorescence Lifetime Imaging Microscopy (FLIM)—For transient expression in leaves, 4- to 6-week-old tobacco (N. tabacum cv SR1) plants were infiltrated with Agrobacterium tumefaciens strain GV3101 with the desired construct as described previously (42) and exhibiting an optical density at 600 nm between 0.025 and 0.05. Infiltration was performed at an induced wound in leaves to facilitate the transformation. In the case of multiple transformations, transformed agrobacteria were mixed. Tobacco plants were kept in the same culture condition during transient expression. Tobacco epidermal cells were observed on a portion of −25 mm² of transformed leaf 72 h after infiltration. FRET ( Förster resonance energy transfer) and FLIM measurements were obtained with a multiphoton confocal microscope (ZEISS LSM 780, Göttingen, Germany) by the so-called TCSPC (Time Correlated Single Photon Counting) method (43). GFP was excited at 920 nm with a pulsed infra-red laser Ti:Saphir (Chameleon ULTRA II, COHER-ENT) for 3 min. The emitted fluorescence was detected by a detector HPM-100 Hybrid (Hamamatsu R10467–40 GaAsP) in descanned position. The laser synchronization and measurement of photon life time were performed using a capture card SPC-830 (B&H). Cells were selected provided that (1) the fluorescence intensity of mCherry/ mRFP was higher than that of eGFP to optimize FRET conditions and (2) cells were isolated from any cell expressing GFP alone, in order not to dilute the FRET signal. One to three biological replicates were
performed by tested couples. For each repetition, 5 to 10 images were obtained and analyzed. From the fluorescence intensity images, the decay curves were calculated per pixel and fitted with either a mono- or double-exponential decay model using the SPImage software (http://www.becker-hickl.de/software/tcspc/software/tcspcspecial.html). The mono-exponential model function was applied for donor samples with only GFP present. For samples containing two fluorophores, GFP/mCherry or GFP/RFP, a double-exponential model function was used without fixing any parameter. The lifetime of GFP expressed alone and the lifetime of GFP co-expressed with mCherry or mRFP were compared using Student’s t-test, by grouping all the repetitions for the same couple. The FRET efficiency is calculated according to the following formula: FRET = 1 - (GFP lifetime in the presence of mCherry/RFP)/(GFP lifetime expressed alone).

Oocyte Expression—cRNA production, expression in Xenopus laevis oocytes, and osmotic water permeability (Pf) measurements were performed as previously described (44).

RESULTS

Interactomics Methodology—We used an IP method coupled to protein identification by MS to characterize proteins copurifying with PIP1:2 and PIP2:1 (further referred to as PIP1:2 and PIP2:1 interactants). Root extracts were prepared from transgenic plants expressing GFP-PIP2:1 or GFP-PIP1:2 under the control of a 35S promoter and paramagnetic anti-GFP microbeads were used to purify the fusion proteins and their interacting proteins. This approach was performed in plants grown in standard conditions or treated with salt (100 mM NaCl, 2h) or hydrogen peroxide (500 μM H2O2, 20 min), two treatments described to inhibit aquaporin activity and root water transport (34, 45). Aquaporins are hydrophobic membrane proteins that require detergents for solubilization. However, these detergents may not be compatible with a proper recovery of interacting proteins. To circumvent this problem, plant tissues were treated with formaldehyde (46) to cross-link protein complexes in vivo. A protein was considered as a genuine PIP interactant when it was identified in at least 3 over 4 replicates with at least two significant different peptides (see methods). In addition, an IP-MS/MS from plants expressing 35S::GFP allowed to identify 22 GFP interactants that were removed from the initial lists of PIP2:1 and PIP1:2 interactants (supplemental Table S2). Overall, a total of 436 and 388 proteins were identified as putative interactants of PIP1:2 and PIP2:1, respectively (supplemental Tables S3, S4), taken into account all plant physiological treatments.

The identification of similar proteins with independent PPI methodologies can be used to validate true interactants. Previous yeast two-hybrid (http://Interactome.dfci.harvard.edu/A_thaliana; Arabidopsis interactome mapping consortium 2011, (18)) and split-ubiquitin approaches (www.associomics.org, (15, 16)) identified as a whole, 29 and 35 proteins interacting with PIP1:2 and PIP2:1, respectively (supplemental Table S5). Three and five of these PIP1:2 and PIP2:1 interactants, respectively, were also identified in the present work. Among them, five are PIPs (24) whereas others correspond to a Ca2+-ATPase, and sugar or phosphate transporters. This comparative analysis confirms the usually low overlap between binary approaches and whole protein complex analysis (47). A reverse IP-MS strategy was used to confirm the interaction between PIP2:1 and PIP1:2 and a few selected interactants using transgenic plants expressing promAMT1:3::AMT1:3-GFP (31, 48), promPGP4::PGP4-GFP in a pgp4 background (32), and promPGP19::PGP19-GFP (49). A total of 282, 22, and 31 proteins were identified in the interactomes of AMT1:3, PGP4, and PGP19, respectively (supplemental Tables S6, S7). The bait itself and both PIP1:2 and PIP2:1 were recovered in each interactome, thereby supporting the relevance of IP-MS to identify PIP interactants.

PIP Interaction Network—PIP1:2 and PIP2:1 shared about 80% of their interactants (Fig. 1A). Given such a high similarity between the two interactomes, all interactants were further considered as defining a “PIP interactome.” One third of them exhibited at least one transmembrane domain (http://aramemnon.botanik.uni-koeln.de/) (supplemental Table S3) and the remaining are soluble proteins possibly bound to membranes. Indeed, 32 and 8% of PIP interactants are predicted to localize to the plasma membrane and endoplasmic reticulum, respectively (http://suba.plantenergy.uwa.edu.au/) (supplemental Table S3). Thus, the PIP2:1 and PIP1:2 interactomes are somewhat enriched in proteins sitting in the same subcellular compartments as their bait. According to Gene Ontology annotation (http://pantherdb.org/), almost 90% of assigned accessions had catalytic activities, binding activities and belonged to proteins contributing to structural integrity of cellular complexes and to transport activity (Fig. 1B). These functions are clearly over-represented when compared with the Arabidopsis genome (Fig. 1B). As for biological processes, metabolic process and localization (Fig. 1C) were predominant, the latter being mostly enriched through the term “vesicle-mediated transport” (Fig. 1C).

Several previous studies pointed to an enrichment of plasma membrane proteins in microdomains according to abiotic, biotic, and chemical treatments (50–53). Interestingly, 22% of PIP interactants (n = 108) were shown to be enriched in microdomains in these studies (supplemental Table S3). In addition, PIP interactome contained 38 PKs (supplemental Table S3) including 23 RLKs and 6 calcium-dependent PKs.

Assuming that interacting proteins contribute to similar molecular functions or cellular processes, we next built a network including all PIP interactants identified in the present work or in previous Y2H (18) and Split-Ub approaches (15–17) sharing interactions (16). Thus, interactants of hub proteins with more than 70 interactants were removed, to prevent corruption of the PIP network with low-affinity interactions. The final network consists of 883 proteins linked by 1620 edges (supplemental Table S8, supplemental Fig. S1). In addition, five main clusters were iden-
Clusters 1, 2, and 3 comprise aquaporins, transporters, and GTPases, respectively, that were already pinpointed in Fig. 1 as enriched molecular functions. Clusters 4 and 5 grouped proteins from exocyst complex or involved in brassinosteroid (BR) signaling, respectively (Fig. 2; supplemental Table S8). The exocyst complex comprises 8 subunits engaged in docking and tethering of secretory vesicles, providing a spatial and temporal regulation of exocytosis (54, 55). BRs are phytohormones that regulate plant growth and development. Thus, this clustering analysis points to novel functional features of the PIP interactome.
Fig. 2. Clustering analysis of PIP network. The MCODE plugin of Cytoscape (41) was used to distinguish 5 major clusters in the PIP network. Corresponding AGI numbers can be found in supplemental Table S8. AGI of proteins are represented by nodes in a color-code way: Blue: interactants identified in the present work with no quantitative variations upon treatments. Red: interactants identified in the present work with quantitative variations upon treatments. Gray: interactants of PIP interactants that are reported in databases. Solid line: data from (18); dashed line: data from (15, 16).
A label-free quantitative mass spectrometry approach was used to measure the variations in abundance of PIP1;2 and PIP2;1 interactants, according to NaCl and H₂O₂ treatments. Forty-nine and 12 proteins of the PIP1;2 and PIP2;1 interactomes, respectively, responded to NaCl (supplemental Table S3, supplemental Table S4) whereas 39 and 38, respectively, showed an altered abundance, in response to H₂O₂ (supplemental Table S3, supplemental Table S4C, S4E). PIP networks were built considering all PIP interactants with variations in abundance according to H₂O₂ (H-network) and NaCl (N-network) treatments, and revealed 243 proteins and 182 connected proteins, respectively (supplemental Table S8, supplemental Fig. S2A, S2B). Interestingly, according to GO annotation, those PIP interactants were enriched in vesicle-mediated transport process (GO:0016192) (supplemental Table S9) suggesting that the two treatments rather interfere with PIP cellular trafficking.

Physical Interaction Between Interactants and PIP2;1—With the objective of identifying proteins that physically act on PIP2;1 function, we selected a subset of plasma membrane localized-proteins, based on their biological function, quantitative variations according to H₂O₂ and NaCl treatments, putative enrichment in microdomains, or presence in a specific cluster. Putative physical interactions with PIP2;1 were then investigated using FLIM. The prevalence of RLKs (56) in PIP interactome made us consider three of them. RKL1 (At1g48480) and RKL902 (At3g17840) are close homologs belonging to the LRRRII-RLK family. Expression of RLK902 in root tips and lateral primordia suggests a role in cell proliferation (57). Feronia (At3g51550) is a well-described RLK that

Fig. 3. Physical interaction between PIP2;1 and selected members of the PIP interactome. FLIM technology was used to study the physical interaction between PIP2;1 and Annexin4, HIR3, RKL1, RKL902, Feronia, NHL3, PLDδ, and PLDγ1. Analyses were performed in leaf epidermal cells transiently expressing fluorescent tagged proteins (A, E, G, I, K, M, O, Q, S) alone or in combination with mCherry- or RFP-tagged proteins (B, D, F, H, J, L, N, P, R, T). Controls for a positive interaction were obtained from the interaction between PIP2;1-GFP and PIP2;1-mCherry (B) and PIP2;1-GFP and PIP2;1-RFP (C). Control for a negative interaction was obtained from the co-expression of PIP2;1-GFP with RFP (D). The donor fluorescence lifetime τ was calculated as described under Experimental Procedures and is indicated by a color code from red for τ values of 1500 ps to blue for τ values of 2500 ps. Values are indicated in Table I. Scale bar: 10 μm.
fine-tunes cell growth by controlling apoplastic pH (58) and ROS (59, 60), thereby balancing wall rigidity for cell integrity and flexibility for cell expansion. These are the reasons why RKL1, RLK902, and Feronia were selected despite their abundance was not altered by H2O2 and NaCl treatments. Phospholipases D (PLDs) fulfill diverse roles in lipid metabolism and cellular regulation (61, 62). Here, we selected PLDδ (At4g35790) and PLDγ1 (At4g11850) that show quantitative variations according to treatments (supplemental Table S3) and are present in cluster 2 of PIP interactome (Fig. 2). Annexin4 (At2g38750) that belongs to the same cluster as PLDs was also selected. In animals, annexins associate with membrane phospholipids and facilitate fusion of cytoplasmic vesicles with the plasma membrane (63) and one of these (annexin A2) mediates c-AMP-induced trafficking of Aquaporin-2 in the collecting duct (64). Finally, we also selected HIR3 because of its localization in microdomains (51, 53) and NHL3 because of its high degree of protein connection (19 proteins including 3 PIPs) (supplemental Fig. S3).

FLIM analyses were performed in tobacco leaf epidermal cells that transiently expressed the two putative interacting partners tagged with GFP and mCherry- or RFP, respectively (Table I, Fig. 3). Physical interactions were assessed based on a significant p value (<0.01), whatever the FRET efficiency. Well established homo-tetrameric interactions of PIPs homologs (24) were used as positive controls. Thus, PIP2;1 fused to GFP showed strong interaction with PIP2;1 fused to RFP and mCherry (Table I, Fig. 3). Negative controls were obtained from coexpression of PIP2;1 fused to GFP with soluble RFP and with HIR3 (At3g01290) that is an anchored membrane protein (Table I, Fig. 3). Whereas no relevant physical interaction could be detected between PIP2;1 and Annexin4, a significant physical interaction was revealed between PIP2;1 and 3 RLKs (RKL1, RLK902, and Feronia), two PLDs (PLDδ and PLDγ1), and NHL3 (At5g06320) (Table I, Fig. 3).

**RKL1, RLK902, and Feronia Modulate PIP2;1 Intrinsic Water Transport Activity**—With regard to their putative PK function, RKL1, RLK902, and Feronia were chosen for further investigating a functional role on PIP2;1 activity, using coexpression in X. laevis oocytes. Expression of PIP2;1 alone conferred, with respect to native oocytes, a 8-fold increase in cell osmotic water permeability ($P_f$) (supplemental Fig. S4A, S4B). Coexpression of PIP2;1 with RKL1 and not with RLK902 resulted in a further increase in $P_f$ by 50% (Fig. 4A, 4B, supplemental Fig. S4A, S4B). Three phosphorylation sites have been described in PIP2;1, at Ser121, Ser280, and Ser283 (1, 4, 8). The $P_f$ of oocytes expressing a PIP2;1 form with Ser-to-Ala mutations at positions 280 and 283 (Ser280Ala-Ser283Ala) was almost similar to $P_f$ of oocytes expressing wild-type PIP2;1 (Fig. 4A, 4B). In addition, coexpression of RKL1 with PIP2;1 Ser280Ala-Ser283Ala resulted in a similar stimulation of $P_f$ as with wild-type PIP2;1 (supplemental Fig. S4A). These results suggest that phosphorylation of PIP2;1 at Ser280 and Ser283 does not contribute to PIP2;1 activity in oocytes, and, in particular, to its enhancement upon coexpression with RKL1. One hypothesis is that Ser121, a residue described to be involved in aquaporin gating could be the target of RKL1. However punctual mutations of Ser121, to an Ala or Asp residue, led to inactive PIP2;1, thereby preventing to test this hypothesis.

By contrast to RKL1, coexpression of PIP2;1 with Feronia resulted, with respect to oocytes expressing PIP2;1 alone, in...
The interaction between GFP-PIP2;1 and mCherry-Annexin4 gave similar results as the interaction between GFP-Annexin4 and PIP2;1-mCherry (data not shown).

a decrease in P_i by 60% (Fig. 5). Lys565 is a residue located in the catalytic domain of Feronia. Punctual mutation of Lys565 to an Arg (Lys565Arg), or deletion of the C terminus of Feronia were shown to abolish its kinase activity (65). When coexpressed with PIP2;1, these mutated forms of Feronia failed to interfere with the water transport activity of the aquaporin (Fig. 5A). These results suggest that Feronia inhibits PIP2;1-mediated water transport through its PK activity. To investigate PIP2;1 residues that would be targeted by Feronia, we coexpressed the latter with forms of PIP2;1 carrying individual or combined mutations of Ser280 and Ser283 to Ala or Asp (Fig. 5B, supplemental Fig. S5). For all PIP2;1 forms carrying individual mutations, coexpression with Feronia resulted in a decrease in P_i by 15% to 38%, whereas wild-type PIP2;1 showed an inhibition by 45%. In addition, Feronia did not alter the P_i of oocytes expressing a Ser280Ala-Ser283Ala form of PIP2;1 whereas the Ser280Asp-Ser283Asp form showed a residual inhibition (Fig. 5B). These results indicate that Feronia possibly acts through Ser280 and Ser283, but not exclusively through one of these residues. Furthermore, phosphodefficient mutations of both Ser280 and Ser283 appeared to fully prevent the inhibitory effect of Feronia whereas phosphomimetic mutations would allow partial inhibition.

**DISCUSSION**

**PIP Interactome: A Platform Regrouping a Wide Range of Transport Activities—**Plant Aquaporin Interactomics

<table>
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<th>Donor</th>
<th>Acceptor</th>
<th>τ (ps) ± S.E.</th>
<th>FRET efficiency</th>
<th>N/n</th>
<th>p (t-test)</th>
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<td>2320 ± 7</td>
<td>20%</td>
<td>1/9</td>
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<td>PIP2;1-GFP</td>
<td>RFP</td>
<td>2319 ± 19</td>
<td>0%</td>
<td>3/12</td>
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<tr>
<td>HIR3-GFP</td>
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<td>HIR3-GFP</td>
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<td>GFP-Annexin4</td>
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<td>2387 ± 37</td>
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<td>0.37</td>
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<td>2436 ± 20</td>
<td>7%</td>
<td>3/20</td>
<td>2.59 × 10^{-8}</td>
</tr>
<tr>
<td>RK1-GFP</td>
<td>-</td>
<td>2429 ± 7</td>
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<td>4.88 × 10^{-9}</td>
</tr>
<tr>
<td>RLK902-GFP</td>
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<td>8%</td>
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<td>2.97 × 10^{-6}</td>
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<tr>
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<td>11%</td>
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<td>9.92 × 10^{-6}</td>
</tr>
<tr>
<td>GFP-NHL3</td>
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<td>13%</td>
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<td>1.33 × 10^{-4}</td>
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<tr>
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<td>13%</td>
<td>2/8</td>
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</tr>
<tr>
<td>PLD6-GFP</td>
<td>-</td>
<td>2319 ± 13</td>
<td>16%</td>
<td>3/12</td>
<td>1.39 × 10^{-8}</td>
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<tr>
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</tr>
<tr>
<td>PIP2;1-GFP</td>
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<td>2390 ± 12</td>
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<tr>
<td>PIP2;1-GFP</td>
<td>PIP2;1-RFP</td>
<td>2075 ± 36</td>
<td>13%</td>
<td>3/13</td>
<td>-</td>
</tr>
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</table>

The interaction between GFP-PIP2;1 and mCherry-Annexin4 gave similar results as the interaction between GFP-Annexin4 and PIP2;1-mCherry (data not shown).

**PIP Interactome:** PIP Interactome: A Platform Regrouping a Wide Range of Transport Activities—Plant Aquaporin Interactomics

<table>
<thead>
<tr>
<th>Donor</th>
<th>Acceptor</th>
<th>τ (ps) ± S.E.</th>
<th>FRET efficiency</th>
<th>N/n</th>
<th>p (t-test)</th>
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<tbody>
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<td>PIP2;1-GFP</td>
<td>-</td>
<td>2320 ± 7</td>
<td>20%</td>
<td>1/9</td>
<td>3.21 × 10^{-17}</td>
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<tr>
<td>PIP2;1-GFP</td>
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<tr>
<td>PIP2;1-GFP</td>
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</tr>
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root water transport involves reactive oxygen species-activated cell signaling and PIP internalization (45). Another study, revealed, using these constructs, that endoplasmic reticulum-retained PIP2;1-GFP may interact with other PIP aquaporins to hamper their trafficking to the plasma membrane, contributing to inhibition of root cell hydraulic conductivity (69). Finally, the use of these constructs for single-molecule analysis of PIP2;1 dynamics and membrane partitioning allowed to
interactants from the membrane bait. In addition, we used formaldehyde-mediated protein cross-linking that recently emerged as an additional means for preserving cellular protein interactions while being compatible with numerous purification strategies (reviewed in (71)). An intrinsic complication of IP-MS is that it may give rise to false hits, in particular when interactions of unwanted proteins with GFP. A high number of unexpected interactions, stress, polarized growth, and membrane transport (74). Interestingly, more than 20% of PIP interactants have been described as enriched in microdomains (supplemental Table S3) although 33% have a transmembrane transport activity (not shown). Under resting conditions, PIP2;1 shows constitutive cycling from the plasma membrane via clathrin-mediated endocytosis, whereas under salt stress conditions, the protein follows an additional clathrin-independent internalization route (70), which is associated with flotillins, a marker of membrane microdomains (75). The PIP interactome may highlight a partitioning of aquaporins in several types of microdomains to ensure transport activities or trafficking at specific submembrane locations.

A network clustering procedure also revealed molecular functions that appear to be tightly connected to PIPs. These include the exocyst complex, in relation to the trafficking processes, BR signaling and GTPases. BR may control aquaporin activity (76) and induce genes involved in water transport, cell-wall organization, and biogenesis in relation to root cell elongation (77). Another link is H$_2$O$_2$, which inhibits aquaporin activity (45) and the production of which seems to be critical for BR-induced stress tolerance in plants (78–81). In the present work, we showed that the abundance of one component of BR cluster (At3g09840) in the PIP interactome was decreased upon H$_2$O$_2$ treatment (supplemental Table S3). In addition, the hormonal network related to auxin and BRs appeared to be affected by PIP downregulation in poplar leaf (82). Thus, we hypothesize that BR signaling pathways could be linked to regulation of PIP function in roots.

**Regulation of PIP Trafficking**—Osmotic stresses induce a partial internalization of PIPs (34, 83) leading to a reduced abundance of PIPs at the root cell surface, which may contribute to a decrease of root water permeability. In addition, stimulus-induced PIP trafficking can be counteracted by reactive oxygen species (ROS) scavengers, in agreement with the central role played by ROS in stress and hormonal signal-
ing in plants (83). Thus, deciphering the molecular and cellular mechanisms that govern PIP dynamics is central to understand the perception and transduction of stress signals in plants. Syntaxins are among the few proteins known to be involved in PIP trafficking (25, 84). In particular, post-Golgi trafficking of Arabidopsis PIP2;7 was shown to depend on a physical interaction with two specific syntaxins, SYIP61 and SYIP121 (26). As mentioned above, clathrin-dependent and -independent endocytic mechanisms allow PIPs to cycle between the plasma membrane and early endosomes under resting conditions and osmotic or oxidative stresses, respectively (25, 70). The latter stresses also induce quantitative changes in the double C-terminal phosphorylation (Ser280 and Ser283) of AtPIP2;1 (1, 8) whereas phosphorylation of Ser283 interferes with trafficking of internalized PIPs to spherical bodies (8). In the present work, quantitative analysis of PIP interactome revealed that H2O2 and NaCl treatments preferentially modify the abundance of interactants involved in vesicle-mediated trafficking (supplemental Table S9). These results support the previously described role of proteins such as syntaxins and clathrin in aquaporin regulation and point to additional protein partners acting on PIP trafficking.

**A Role for Lipid Signaling in PIP Function**—IP-MS studies do not allow to distinguish between proteins that physically or indirectly interact with the bait. With the objective of focusing on physical PIP interactants, we applied the FLIM technology to eight PIP interactants selected according to their molecular function, their quantitative variations according to H2O2 and NaCl treatments, their putative enrichment in microdomains or their presence in a specific cluster. Among them, Annexin 4 and HIR3 did not show any physical interaction with PIP2;1 (Fig. 3, Table I). By contrast, members of phospholipase D and RLK families as well as NHL3 showed a physical interaction with PIP2;1. Multiple lines of evidence indicate that, in humans, Aquaporin-3 (AQPD3) and phospholipase D both contribute to skin function. AQPD3 co-localizes with phospholipase D2 in caveolin-rich membrane micro-domains, the former delivering glycerol to the latter, for synthesis of phosphatidylglycerol, a lipid messenger regulating keratinocyte proliferation and differentiation (85). In plants, PLDs and their enzymatic product phosphatidic acid (PA) play roles in cellular responses to hormonal and abiotic stimuli, as well as in plant-microbial interactions and plant defense against bacterial and fungal pathogens (62). PA is therefore regarded as a universal lipid signaling molecule. It often directly binds to proteins to alter their localization, enzymatic activity, or interactions with membrane or cytoskeleton (86). More than 30 plant proteins from diverse physiological pathways have been identified as PA targets and the list is still being extended (62, for review). In particular, a recent work identified AtPIP2;1 and AtPIP2;2 as PA-binding (87) but the functional effects of these interactions remain to be elucidated. Additionally, AtPLD5 is required for ABA-induced stomatal closure, acting downstream of H2O2 and nitric oxide in the ABA signaling pathway (88). Here, we showed that, in roots, the quantity of PLDδ in the PIP2;1 and PIP1;2 interactome decreased upon H2O2 treatment (supplemental Table S3). Thus, we hypothesize that a reduced physical interaction between PLDδ and PIP2;1 upon H2O2 treatment would result in a decreased PA binding to PIP2;1, thereby altering PIP2;1 function. Such a role for lipid signaling in PIP2;1 function is consistent with the suggested role of lipid-mediated signaling in the transduction of stress signals arising from the soil (89).

**Differential Roles of Receptor-like Kinases in PIP Function**—The PKs known to act on PIPs include two PKs phosphorylating spinach SoPIP2;1 at Ser115 and Ser274 (9), OST1/SnRK2.6, a PK involved in guard cell ABA signaling (10), SIRK1 and BSK8, (12, 13). Our IP-MS strategy identified 37 kinases as putative PIP interactants, of which 16 are RLKs. RKL1 and RLK902 physically interact with PIP2;1 and both belong to the LRR III subfamily. RLK902 and RKL1 were described as functional PKs able to auto-phosphorylate (57). At the macroscopic level and in standard culture conditions, neither the rkl1 and rkl902 mutant lines nor the rkl1/rkl902 double knockout line showed any significant phenotypes (57). Coexpression of PIP2;1 with RKL1 in oocytes enhanced PIP2;1 water transport activity (Fig. 4A). However, despite a 75% amino acid sequence identity with RKL1, RLK902 was unable to activate PIP2;1 water transport activity (Fig. 4B), suggesting a strong specificity of these two PKs toward their substrates. Previous structure-function analyses have revealed the role of several cytosol-exposed phosphorylation sites of PIPs in controlling their water transport activity (21). In our study, PIP2;1 mutant analysis showed that stimulation by RKL1 is not mediated through two well-characterized C-terminal phosphorylation sites, Ser280 and Ser283. Because a recent study showed that Ser121 of loop B is the target of OST1/SnRK2.6 (10) we also investigated punctual mutations at this site. However, none of the mutated forms (Ser121Ala, Ser121Asp) yielded an active PIP2;1. Thus, in the absence of direct biochemical evidence, we cannot definitely conclude about the phosphorylation site(s) recognized by RKL1 in PIP2;1.

By contrast to RKL1, co-expression of PIP2;1 with Feronia decreased PIP2;1 water transport activity by 60% (Fig. 5). Feronia belongs to the Catharanthus roseus RLK1-like kinase family (CrRLK1Ls) characterized by their extracellular carbohydrate-binding malectin domains (90). Several recent works demonstrated that Feronia is critical to fine-tuning cell growth. In particular, it controls apoplastic pH (58) and ROS (59, 60), thereby balancing wall rigidity for cell integrity and flexibility during cell expansion. A recent study demonstrated that, upon binding to a small secreted peptide, RAPID ALKALIZATION FACTOR (RALF) (58), Feronia acts as a growth inhibitor in the root post-elongation zone. In the present work, punctual mutation of the kinase domain of Feronia or its deletion prevented the inhibitory effect of Feronia on PIP2;1 (Fig. 5) demonstrating that this effect is mediated through phosphor-
ylation. Functional analysis of PIP2;1 punctual mutants showed that inhibitory effect of Feronia was not individually mediated through Ser280 or Ser283 (Fig. 5). By contrast, a double phospho-deficient form (Ser280Ala-Ser283Ala) but not a double phospho-mimetic form (Ser280Asp-Ser283Asp) of PIP2;1 had become resistant to the inhibitory effect of Feronia (Fig. 5B). Thus, the combined phosphorylation of these two sites may favor the inhibitory action of Feronia. We do not believe that Ser280 and Ser283 serve as phosphorylation sites for Feronia. We rather showed that binding of Feronia to PIP2;1 may be favored by their combined phosphorylation (Fig. 6, Table II). Because phosphorylation of both Ser280 and PIP2;1 may be favored by their combined phosphorylation for Feronia. We rather showed that binding of Feronia to Physiological Molecular of Plants, INRA, 2 place Viala, Montpellier, France.

This work is supported by a research contract with Syngenta. This article contains supplemental material.

To whom correspondence should be addressed: Biochimie et Physiologie Moléculaires des Plantes, INRA, 2 place Viala, Montpellier, Cécile Fizames and Olivier Langella for bioinformatics assistance.

REFERENCES


Molecular & Cellular Proteomics 15.11