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Short Title:

Influence of LHCX isoforms on NPQ sites in diatoms

Corresponding authors:

Giovanni Finazzi: giovanni.finazzi@cea.fr and Angela Falciatore: angela.falciatore@upmc.fr

Title:

Dynamic changes between two LHCX-related energy quenching sites control diatom photoacclimation

Lucilla Taddei[a], Volha U. Chukhutsina[b,c], Bernard Lepetit[d], Giulio Rocco Stella[a,e], Roberto Bassi[e], Herbert van Amerongen[b], Jean-Pierre Bouly[a], Marianne Jaubert[a], Giovanni Finazzi[f], and Angela Falciatore[a]

Affiliations:

[a]Sorbonne Université, CNRS, Institut de Biologie Paris-Seine, Laboratory of Computational and Quantitative Biology, F-75005, Paris, France

[b]Laboratory of Biophysics and MicroSpectroscopy Research Facility, Wageningen University, P.O. Box 8128, 6700ET Wageningen, The Netherlands

[c]Biophysics of Photosynthesis, Department of Physics and Astronomy, Faculty of Sciences, VU University Amsterdam and LaserLaB Amsterdam, 1081 HV Amsterdam, The Netherlands

[d]Zukunftskolleg, Department of Plant Ecophysiology, University of Konstanz, 78457 Konstanz, Germany

[e]Department of Biotechnology, University of Verona, 15, Strada Le Grazie, I-37134 Verona, Italy

[f]Université Grenoble Alpes (UGA), Laboratoire de Physiologie Cellulaire et Végétale, UMR 5168, Centre National de la Recherche Scientifique (CNRS), Institut National Recherche Agronomique (INRA), Commissariat à l'Energie Atomique et aux Energies Alternatives (CEA), Institut de Biosciences et Biotechnologies de Grenoble, (BIG), CEA Grenoble, F-38054 Grenoble cedex 9, France

One sentence summary:

Multiple LHCX-related quenching sites control short- and long-term high-light acclimation in the marine diatom *Phaeodactylum tricornutum*.

Author contributions:

A.F., G.F., H.v.A., J.P.B., M.J., B.L. and V.U.C. designed the experiments and wrote the manuscript. L.T., M.J., B.L., A.F., J-P.B., and G.F., contributed to the molecular and physiological analyses; G.R.S. and R.B. performed the pigment analysis; V.U.C. and H.v.A. analyzed quenching features and generated decay-associated spectra. All authors discussed results, revised, and approved the manuscript.

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Address correspondence to giovanni.finazzi@cea.fr and angela.falciatore@upmc.fr

Abstract

Marine diatoms are prominent phytoplankton organisms that perform photosynthesis in extremely variable environments. Diatoms possess a strong ability to dissipate excess absorbed energy as heat via non-photochemical quenching (NPQ). This process relies on changes in carotenoid pigment composition (xanthophyll cycle) and on specific members of the light-harvesting complex (LHC) family specialized in photoprotection (LHCXs), which potentially act as NPQ effectors. However, the link between light stress, NPQ, and the existence of different LHCX isoforms is not understood in these organisms. Using picosecond fluorescence analysis, we observed two types of NPQ in the pennate diatom *Phaeodactylum tricornutum*, which were dependent on light conditions. Short exposure of low-light-acclimated cells to high light triggers the onset of energy quenching close to the core of photosystem II, while prolonged light stress activates NPQ in the antenna. Biochemical analysis indicated a link between the changes in the NPQ site/mechanism and the induction of different LHCX isoforms, which accumulate either in the antenna complexes or in the core complex. By comparing the responses of wild-type cells and transgenic lines with a reduced expression of the major LHCX isoform, LHCX1, we conclude that core-complex-associated NPQ is more effective in photoprotection than is the antenna complex. Overall, our data clarify the complex molecular scenario of light responses in diatoms and provide a rationale for the existence of a degenerate family of LHCX proteins in these algae.

Introduction

Marine diatoms form a group of unicellular algae that dominate the phytoplankton community across a wide range of ocean environments (Smetacek, 1999; de Vargas et al., 2015; Malviya et al., 2016). Their environmental success likely reflects their capacity to respond to numerous environmental challenges, including changes in nutrient levels and light. While the mechanisms of the responses of diatoms to nutrients have been studied in detail (Allen et al., 2008; Allen et al., 2011; Marchetti et al., 2012; Alipanah et al., 2015; Morrissey et al., 2015; Matthijs et al., 2016; McQuaid et al., 2018), little is known about light acclimation responses. Like most photosynthetic organisms, diatoms optimize light capture by enhancing their absorption capacity at low intensities, and by down-regulating the utilization of absorbed light at oversaturating energy fluxes (Müller et al., 2001; Eberhard et al., 2008). The latter process is triggered by the induction of the high-energy quenching (qE) component of non-photochemical quenching (NPQ) (Horton et al., 1996). qE reflects the increased thermal dissipation of excess light following the activation of qE effector proteins in photosystem II (PSII) and changes in the pigment composition (via carotenoid de-epoxidation through the xanthophyll cycle, XC). qE effectors include the small PSII subunit (PsbS) in plants and members of the light-harvesting complex stress-related (LHCSR) family in microalgae and mosses (Peers et al., 2009; Alboresi et al., 2010; Ballottari et al., 2016). The qE machinery of diatoms differs from that of plants and green algae in two main aspects. Diatoms possess two xanthophyll cycles catalyzing the de-epoxidation of diadinoxanthin (DD) to diatoxanthin (DT) and of violaxanthin (V) to zeaxanthin (Z) (Lohr and Wilhelm, 1999). Moreover, their qE effectors belong to the light-harvesting complex (LHC) family specialized in photoprotection, the LHCX, (Bailleul et al., 2010; Zhu and Green, 2010; Ghazaryan et al., 2016), which is related, but not identical, to the LHCSR family. Multiple LHCX genes exist in

diatoms and gene expression studies indicate that the four *Phaeodactylum tricornutum* LHCX isoforms differentially accumulate in the thylakoids upon exposure to different environmental stresses due to the existence of multiple regulatory control pathways (Allen et al., 2008; Nymark et al., 2009; Bailleul et al., 2010; Lepetit et al., 2013; Lepetit et al., 2017). These findings suggest that the functional diversity of the LHCX proteins may expand the diatom's capacity to respond to the highly variable ocean environments (Zhu and Green, 2010; Taddei et al., 2016; Lepetit et al., 2017).

In this work, we combined biochemical and spectroscopic approaches to address the role of the different LHCXs in photoprotection. We found that low-light (LL) -acclimated cells display a qE mainly driven by energy quenching in close proximity to the reaction center of PSII (the PSII core) and, to some extent, in the antennas (also called Fucoxanthin Complex Binding Proteins, FCPs). This qE is largely controlled by LHCX1, which is present in both the PSII core and the FCP complexes. On the other hand, prolonged exposure to high light (HL) enhances FCP localized quenching. Biochemical analysis suggests that this shift is related to the induction of other LHCX isoforms, which accumulate in the antenna but not in the PSII core. By comparing the physiological responses of wild-type (hereafter called WT) and knock-down lines with reduced content of the LHCX1 isoform, we conclude that qE antenna quenching is less effective than core qE in protecting cells from light damage. Overall, by relating different qE mechanisms to different molecular actors, we propose a detailed model for diatom NPQ, which is one of the key elements of the environmental flexibility of these algae in modern oceans.

Results

***The LHCX1* knock-down line recovers WT-NPQ levels upon prolonged high-light exposure**. In *P. tricornutum,* LHCX1 is the only member of the *LHCX* gene family that is substantially expressed in cells grown in LL (30 µmol photons $m^{-2} \cdot s^{-1}$, 12:12 h light:dark cycle) (Bailleul et al., 2010; Taddei et al., 2016; Lepetit et al., 2017). In these conditions, LHCX1 is the main NPQ effector. This role is evidenced by the phenotype of a transgenic line with down-regulated expression of *LHCX1* (hereafter named *lhcx1*), which contained less LHCX1 and showed a lower qE capacity than the WT when grown in LL (Bailleul et al., 2010) (Fig. 1A and Supplemental Fig. S1). Exposure to HL (500 µmol photons $m^{-2} \cdot s^{-1}$, 12:12 h light:dark cycle, Fig. 1B) for two days enhanced NPQ in both WT and transgenic cells. However, the NPQ increase was larger in the mutant, and therefore, the quenching capacity of the two strains became indistinguishable in HL. Immunoblot analysis of HL-treated cells showed that LHCX1 levels were increased in both strains, even if the *lhcx1* knock-down cells maintained a lower LHCX1 content than the WT cells (Fig. 1C). This observation suggests that the increase in LHCX1 alone cannot account for the observed difference in NPQ amplitude in LL- and HL-treated cells. To further elucidate the effect of quenching on antenna protein domains, we compared the XC pigments in WT and *lhcx1* cells in LL and HL and found a similar DD/DT content in LL-treated cells (Supplemental Table S1), which was in agreement with previous results (Bailleul et al., 2010). HL triggered a significant increase of DD+DT but also led to the appearance of violaxanthin (V), antheraxanthin (A) and zeaxanthin (Z), i.e. xanthophyll precursors of DD+DT synthesis under HL stress (Lohr and Wilhelm, 1999). The *lhcx1* knock-down line displayed a slightly increased amount of total DD+DT compared to the WT, while

its de-epoxidation state was the same as of the WT. This increase of DT could, in principle, account for the recovery of WT-like NPQ levels in the *lhcx1* cells. Nevertheless, previous work has shown that induction of DT by prolonged light stress cannot enhance NPQ without a concomitant increase of LHCX proteins (Bailleul et al., 2010; Lepetit et al., 2013; Lepetit et al., 2017). On the other hand, we detected a substantial accumulation of LHCX3 in both strains in HL (Fig. 1C). Induction of the LHCX2 isoform was also visible at the mRNA level (Fig. 1E), and at the protein level after over-exposure of the western blot membrane (Fig. 1D). Because of their possible role in NPQ, this finding prompted us to further investigate the link between the induction of these proteins and the acquirement of WT levels of NPQ in HL-grown *lhcx1* cells.

Different quenching capacities in LL- and HL-treated cells reflect a heterogeneous distribution of the LHCX isoforms in different chloroplast fractions. Previous studies have localized LHCX1 either in the FCP complexes (Lepetit et al., 2010; Schaller-Laudel et al., 2015) or in photosystem I (PSI) (Grouneva et al., 2011). However, no information is available for the other LHCX isoforms. Therefore, we reinvestigated the localization of the various LHCX proteins in the different photosynthetic complexes isolated by sucrose density gradient centrifugation (Fig. 2A) from detergent-treated thylakoid membranes of *P. tricornutum.* Five distinct fractions were recovered from LL- and HL-treated WT and *lhcx1* strains (Fig. 2A). Using western blot analysis, we identified them as free pigments, trimeric FCPs (i.e. the physiological antenna form in *P. tricornutum* (Lepetit et al., 2007; Joshi-Deo et al., 2010; Gardian et al., 2014), PSII monomers (PSII m), PSI, and PSII dimers (PSII d) in accordance with earlier results obtained with clear native polyacrylamide gel electrophoresis (PAGE) (Nagao

et al., 2013). LHCX1, the only isoform strongly expressed in LL-treated cells, is ubiquitous as it co-localizes with the FCP and PSI as well as with the PSII dimer fractions (Fig. 2B). As expected, samples isolated from *lhcx1* cells had a lower content of this protein. On the other hand, LHCX3, which is induced in HL-treated cells (Nymark et al., 2009; Bailleul et al., 2010; Lepetit et al., 2013; Taddei et al., 2016; Lepetit et al., 2017), seems to have a more specific localization, being found only in the FCP and PSI fractions (Fig. 2B). The LHCX2 isoform could not be detected by western blot analysis, likely because of a lower level of accumulation as compared to LHCX3 in prolonged HL stress and a lower affinity of the antibody generated against the Chlamydomonas LHCSR3 for this isoform.

Prolonged exposure to HL induces a change in the NPQ quenching site in *P. tricornutum* cells.

We employed picosecond spectrally-resolved fluorescence measurements to analyze the quenching features in LL- and HL-treated WT and *lhcx1* cells. To discriminate between quenching in the antennas and PSII cores, we selectively excited different pigment populations. We excited fucoxanthin (FX), which is only found in the FCP antenna complexes (Lepetit et al., 2010), with 540-nm light. Conversely, we used 400-nm light to excite chlorophyll *a* (Chl *a*) (Szabó et al., 2008; Chukhutsina et al., 2013), which is present in PSI and PSII cores and in the FCPs. Time-resolved fluorescence emission data were globally fitted to obtain the fluorescence lifetimes and the corresponding decay-associated spectra (DAS).

Global analysis of time-resolved fluorescence data was performed in WT and *lhcx1* lines in LL (Fig. 3) and HL (Fig. 4) excited at 400 and 540 nm, both in the quenched and unquenched states. We found that five components are required for accurately describing

193 the fluorescence kinetics in LL (Fig. 3) and the results on WT cells upon excitation with 400
194 nm light are described in more detail below. The three components with the shortest
195 lifetimes (14 ps, 64 ps, and 242 ps) mainly reflected excitation energy transfer from short-
196 wavelength (high-energy) pigments to long-wavelength (low-energy) pigments. This downhill
197 energy transfer could easily be recognized because the corresponding DAS were positive on
198 the short-wavelength side and negative on the long-wavelength side (Van Stokkum et al.,
199 2008). For example, the 14-ps DAS displayed a positive band at 675 nm and two negative
200 bands at 690 nm and 717 nm. This reflected excitation energy transfer (EET) from
201 chlorophyll (Chla) with fluorescence peaking around 675 nm (Chl_{675}) to Chl_{690} and Chl_{717}, and
202 mainly represented EET from FCPs to both photosystems, and possibly Lhcf15, a member of
203 the Fucoxanthin Chlorophyll a/c binding Protein family (Chukhutsina et al., 2014). Indeed,
204 the peak position of Chl_{717} strongly resembled that of the red (long-wavelength) antennas
205 composed of Lhcf15, which emit at 716 nm at 77 K (Herbstová et al., 2015; Herbstová et al.,
206 2017). The assignment of this long-wavelength emission (to PSI or Lhcf15, or both) is further
207 discussed in the next section. The second DAS represented a similar process occurring on a
208 slower timescale (64 ps). The 242-ps (3^{rd}) DAS reflected energy equilibration between Chl_{687}
209 and Chl_{717}, and surprisingly, was an order of magnitude slower than observed previously for
210 FCPs of *Cyclotella meneghiniana* (Chukhutsina et al., 2014). This suggested that the red
211 antennas are part of a large antenna system, in which it takes a relatively long time to reach
212 some of the red-emitting species. The 894-ps DAS represented fluorescence decay processes
213 in PSII and PSI emitting at 690 nm and 712 nm, respectively. The 4-ns DAS, emitting at 717
214 nm, again reflected relaxation of the "red-most" emitters of *P. tricornutum.* Fig. 3 also shows
215 the results for LL-treated WT cells in which NPQ has been induced. In this case, we found
216 similar lifetimes (14 ps, 61 ps, 219 ps, 816 ps, and 4 ns) as for unquenched cells (Fig. 3,

Supplemental Table S2) but the DAS are different. The 14-ps spectrum was virtually identical for the quenched and unquenched sample, including the EET part to the long-wavelength pigments (Fig. 3). On the other hand, the 61-ps and 219-ps components showed strongly reduced EET to the long-wavelength band. As a result, the amplitude of the 4-ns DAS decreased substantially, corresponding to a decrease of the average fluorescence lifetime (Supplemental Table S3) and quenching of the fluorescence. We directly compared the "steady-state" fluorescence spectra, which have been reconstructed from the DAS as explained in Materials and Methods. The resulting quenched and unquenched spectra for LL-treated WT cells are presented in Fig. 5A. Both spectra were dominated by the red-shifted fluorescence band around 717 nm, but the spectrum corresponding to the NPQ state was substantially smaller. We also observed quenching of the 687-nm emission (Fig. 5 insert).

The same measurements were performed with a 540-nm excitation to preferentially excite the antenna (Fig. 3B), and the obtained DAS were very similar to those obtained upon the 400-nm excitation (see Fig. 3A). Also, the reconstructed steady-state spectra for quenched and unquenched cells (Fig. 5B) were very similar to those obtained with 400-nm excitation.

Time-resolved fluorescence measurements were also performed on LL-treated *lhcx1* cells and the results for unquenched cells were very similar to those of WT cells (Fig. 3, C and D). The spectra for the quenched cells were also reminiscent of those of the WT cells. However, the amount of quenching was smaller as can be seen for the reconstructed steady-state spectra in Fig. 5 (C and D). Moreover, quenching of the 687-nm emission was not present in the transgenic cells (Fig. 5 inserts). It is important to note that the differences induced by NPQ cannot be directly compared to those obtained during fluorescence induction

measurements, which were performed at room temperature (RT) (Fig. 1). Indeed, the fluorescence steady state emission (Fig. 5) calculated at 77K was dominated by the long-lived PSI fluorescence or other red-most emitting species, at variance with fluorescence measured at room temperature.

Similar measurements were then performed on HL-treated cells (Fig. 4). Four decay components were sufficient to fit the data for both types of cells upon excitation with either 400 nm or 540 nm. In contrast to the results on LL-treated cells, there were substantial differences between the two excitation wavelengths, which were clearly visible when comparing the reconstructed steady-state spectra (Fig. 5 E to H). Excitation of the antenna at 540 nm led to enhanced fluorescence quenching (Fig. 5F) as compared to the 400-nm excitation (Fig. 5E), and this was also observed for the *lhcx1* cells (Fig. 5 G and H). Therefore, it can be concluded that in HL-treated cells, a substantial portion of the quenching is localized in the FCPs. Earlier studies using time-resolved fluorescence (Miloslavina et al., 2009; Chukhutsina et al., 2014) identified two quenching sites in diatoms. The first one (Q1) was mainly localized in the antenna complexes that detach from the photosystems during quenching. The second one (Q2) was found in close proximity to the PSII core (Chukhutsina et al., 2014). In the frame of this model, our data suggest that Q1 quenching becomes more prominent in *P. tricornutum* cells under HL conditions. Under LL conditions, this antenna-related quenching is not observed and we conclude that the WT cells (and to a far lesser extent, the *lhcx1* cells) develop NPQ mainly based on Q2 localized quenching.

Overall, the results of picosecond fluorescence decay spectroscopy and biochemistry suggest that LHCX1 is responsible for the large NPQ associated to the PSII core that is observed in LL. Hence, LHCX1 is mandatory for Q2. Conversely, the onset of the “extra”

antenna-related NPQ observed in HL-exposed cells (Q1) should be due to the specific induction of other LHCX isoforms, such as LHCX3, predominantly detected in the antenna fraction.

Consequences of "antenna-" and "PSII core-" localized quenching on light acclimation of *P. tricornutum* cells. Based on the conclusions from the picosecond fluorescence measurements (i.e. the preferential detection of "antenna" (Q1) and "PSII core" (Q2) quenching in diatoms in HL and LL conditions, respectively), we tried to evaluate the relative efficiency of the two quenching mechanisms in protecting the photosynthetic apparatus from photodamage. To this aim, we compared the photosynthetic performances of WT and *lhcx1* cells grown either in LL, where the "PSII core" quenching is mostly active, or in HL, where antenna quenching becomes the prominent component of NPQ.

A shift from LL to HL for two days largely increased the photosynthetic activity of WT cells, indicating that the cells properly acclimate to the higher photon flux (Fig. 6 A) and therefore sustain growth (Supplemental Fig. S2). In parallel to the increased photosynthesis, respiration was also enhanced (Supplemental Table S4) as expected because of the tight link between the two processes in diatoms (Bailleul et al., 2015). On the contrary, *lhcx1* cells were unable to increase their photosynthetic and respiratory capacities upon HL exposure for two days (Fig. 6B and Supplemental Table S4). By calculating PSII inactivation (either from changes in the Fv/Fm ratio or using the 1/Fo - 1/Fm parameter (Campbell and Tyystjärvi, 2012), we observed a stronger photoinactivation in *lhcx1* knock-down cells compared to the WT (Supplemental Table S4) when exposed to HL. This also suggested that despite the similar NPQ capacity of both cell lines in HL, *lhcx1* cells were more prone to photoinhibition

than the WT. This conclusion was supported by a biochemical analysis of the thylakoid main complexes using antibodies for the LHCX proteins, the PSII (D2) and PSI (PsaF) photosynthetic subunits (Fig. 6, C and D), and the ATPase complex subunit (βCF1). We found that both genotypes displayed reduced levels of PSII and PSI proteins upon exposure to HL for 2 days. Overall, these data confirm that *P. tricornutum* responds to increasing light intensities by reducing the number of reaction centers, a strategy known in diatoms and other microalgae as the n-type photoacclimation (Falkowski and Owens, 1980). However, the effect on PSII was exacerbated in the *lhcx1* cells, suggesting that PSII was specifically degraded upon the high light shift in *lhcx1* as a consequence of photoinhibition.

Discussion

Our biochemical, spectroscopic, and physiological investigation suggests a model for photoprotection in *P. tricornutum* (Fig. 7), in which the differential accumulation of LHCX isoforms in different photosynthetic complexes modulates the efficiency of NPQ via different quenching mechanisms. We show that two main LHCX isoforms present in the light (LHCX1 and LHCX3 (Taddei et al., 2016)) are located in different regions in the photosynthetic complexes of this alga. While LHCX1 is ubiquitously distributed in PSI, PSII, and the FCPs, LHCX3 is only associated with the PSI and FCP complexes. Moreover, LHCX1 and LHCX3 are differentially expressed depending on the light regime. LHCX1 is the predominant isoform in LL, while LHCX1 and LHCX3 accumulate in HL-exposed cells. We suggest that in LL-acclimated cells, LHCX1 would provide a constitutive NPQ capacity mainly localized near the PSII core (Q2 (Miloslavina et al., 2009; Chukhutsina et al., 2014)) where LHCX1 is found (Fig. 7). The existence of a link between this quenching and LHCX1 is supported by the finding that the NPQ amplitude is reduced when the content of LHCX1 is diminished, e.g. in the *lhcx1* knock-

down line, in the Pt4 ecotype, and also in WT cells at the end of the day (Bailleul et al., 2010). In addition to this "basal" quenching process, an additional quenching is observed upon HL exposure for a few days (Lepetit et al., 2013; Lepetit et al., 2017). This quenching is mostly localized in the FCPs, and therefore corresponds to the previously identified Q1 type of quenching (Miloslavina et al., 2009; Chukhutsina et al., 2014). In the antenna, Q1 would benefit from the additional presence of the HL-inducible LHCX3 isoform (Fig. 7), and possibly the LHCX2 isoform in this process, which also accumulates to some extent under HL stress (Taddei et al., 2016; Lepetit et al., 2017).

Our physiological data also allowed us to assess the relative efficiency of the two LHCX-related NPQ mechanisms. While both the WT and *lhcx1* lines have a comparable NPQ capacity in HL, *lhcx1* cells are more prone to photoinhibition. However, antenna quenching, Q1, is prominent in cells with a deregulated LHCX1 expression, while Q2 quenching ("PSII core" quenching) dominates in the WT. Overall, this observation suggests that "PSII core" quenching is more efficient in protecting diatoms against photoinhibition of PSII, as recently hypothesized (Kuzminov and Gorbunov, 2016; Giovagnetti and Ruban, 2017). In the frame of this model, the differences in NPQ (Fig. 1) and photosynthesis (Fig. 6) between WT and *lhcx1* cells in LL vs. HL conditions can be explained based on the presence of distinct complexes that are differentially quenched by members of the LHCX family.

Consistent with the above scenario, previous studies have revealed a fine-tuning of qE based on changes in the amount/localization of the qE protein effectors in other photosynthetic organisms**.** In plants, Bergantino and colleagues have proposed that PsbS could trigger different types of NPQ via its association with either the PSII core or the LHCII (light harvesting complex II) antenna complexes (Bergantino et al., 2003). This would occur

via a hypothesized protein monomerization, which has recently been experimentally observed *in vitro* (Fan et al., 2015). Consistent with this idea, a fast-developing NPQ is lost in the NoM mutant lacking PSII core-bound monomeric LHCs, while the slow-developing quenching was unaffected (Dall'Osto et al., 2017). In green algae, differential binding of LHCSR3 to PSI and PSII has been reported and related to changes in NPQ (Allorent et al., 2013). Recently, Pinnola and colleagues have also shown that PSI-bound LHCSR1 induces NPQ in this complex (Pinnola et al., 2015) in the moss *Physcomitrella patens*. Our findings that LHCX3 is bound to PSI are consistent with the occurrence of a similar quenching process in diatoms as well. However, testing this possibility is difficult in diatoms due to the peculiar nature of the long-wavelength fluorescence band around 717-720 nm, which is seen in *P. tricornutum* cells, especially in LL-treated cells. In plants and green algae, PSII and PSI fluorescence emission can be easily distinguished by their spectral features and lifetimes, with PSI emitting at longer wavelengths with a shorter lifetime. In diatoms, the 717-720 nm band has been attributed to emission by a PSII-associated red-shifted antenna (Herbstová et al., 2015; Herbstová et al., 2017). In our global analysis of WT cells excited at 400 nm, we observed 3 DASs that show EET towards the long-wavelength pigments. The fastest component of 14 ps is characteristic for EET in PSI and is observed for PSI from the plant *Arabidopsis thaliana* (Wientjes et al., 2011; Tian et al., 2017) and the green alga *Chlamydomonas reindhartii* (Ünlü et al., 2016; Wlodarczyk et al., 2016). The observed time constants for the major part of the transfer range from 5-11 ps in *A. thaliana* to 7-29 ps in *C. reinhardtii*, which is similar to the 14 ps observed for LL-treated WT unquenched cells. The other components reflecting EET to the long-wavelength pigments are far slower (64 and 242 ps) than usually observed for PSI and we ascribe them to transfer to Lhcf15 proteins, which are known to emit at 716 nm at 77K (Herbstová et al., 2015; Herbstová et al., 2017). It

357 is also worthwhile to mention that no nanosecond component with PSI characteristics has
358 been observed for diatoms (Chukhutsina et al., 2014), in contrast to what has been reported
359 for native membranes of higher plants or cyanobacteria, where DAS with 2-ns and 7.4-ns
360 lifetimes represent slow PSI trapping from red pigments (van der Weij-de Wit et al., 2011;
361 Chukhutsina et al., 2015). Interestingly, when NPQ is induced in the LL-treated WT cells, the
362 fastest DAS remains entirely unchanged as well as the 14-ps lifetime. This finding suggests
363 that LHCX1 does not induce quenching of PSI but rather induces quenching on the red-
364 shifted antennas of PSII. Consistent with this, transfer to the long-wavelength pigments of
365 Lhcf15 is reduced considerably, leading to substantial quenching of their fluorescence. The
366 results obtained for the 540-nm excitation are also consistent with this picture and the same
367 is true for the results on LL-treated *lhcx1* cells. When the cells are grown in HL, the long-
368 wavelength fluorescence is strongly reduced. However, upon induction of NPQ, the long-
369 wavelength band is also quenched. Again, no clear difference is observed in the fastest DAS,
370 which corresponds to EET to the long-wavelength band, while quenching is mainly due to the
371 reduction of the 4-ns component. These findings might suggest that like LHCX1, LHCX3 does
372 not induce NPQ in PSI but rather in the red-shifted antennas of PSII, although the possibility
373 of quenching in PSI cannot be unambiguously ruled out.

374 Overall, we propose that active regulation of the two forms of quenching by different
375 LHCX isoforms provides a rationale for the existence of several isoforms of these qE
376 effectors, their number being, on average, larger than that found in all the other algal
377 species studied thus far (see (Taddei et al., 2016; Mock et al., 2017)). Multiple regulation of
378 the LHCX family members by nutrient starvation (Taddei et al., 2016) and other stresses
379 (e.g., light fluctuation (Lepetit et al., 2017) and prolonged darkness (Taddei et al., 2016))
380 would provide additional degrees of flexibility in controlling responses to environmental

changes, as required for efficient acclimation to the continuous changes of the ocean environment.

Materials and Methods

Strains and culture conditions: Axenic *P. tricornutum* (Pt1 8.6, CCMP2561) wild type and the *lhcx1* knock-down (Bailleul et al., 2010) strains were grown in f/2 medium at 19°C in a 12-h-light:12-h-dark photoperiod. Cells were first acclimated to 30 µmol photons $m^{-2} \cdot s^{-1}$ (LL) and then shifted to 500 µmol photons $m^{-2} \cdot s^{-1}$ (HL) white light for two days. Cells were collected during the exponential phase of growth.

Oxygen evolution and consumption: Rates of oxygen evolution and consumption were measured with a Clark electrode (Hansatech, UK) at different light intensities (0, 90, 200, 450, 750, and 2300 µmol photons $m^{-2} \cdot s^{-1}$) and the measurement was performed when the signal was stable. Illumination was maintained for 2 minutes at every intensity to attain steady-state oxygen evolution while avoiding an excessive illumination that could lead to photoinhibition. Net photosynthesis was calculated as light-driven oxygen evolution minus dark respiration.

Pigment analysis: Pigment extraction was performed on cells grown either in LL or HL for two days. Cells were irradiated for 10 min with strong HL before being collected by quick filtration. Pigments were extracted on ice using 96% ethanol, buffered with Na_2CO_3, in the dark for 30 minutes and centrifuged. The supernatant was loaded in a high-performance liquid chromatograph (Thermo-Fisher) with a detector diode array to analyze the visible region with a C18 spherisorb column (7.3 x 30mm) using an aqueous mixture of acetonitrile/methanol/0.1 M Tris-HCl buffer (pH 8.0) (72:8:3, v:v:v, buffer A) and a

methanol/hexane mixture (4:1, v:v, buffer B). The runs were done at a flux of 1.5 mL, starting with 100% buffer A: 0-5 min 97% A, 5-17 min a gradient to 80% A, 17-18 min to 100% of buffer B, 18-23 min 100% B. Pigments are distinguishable by the retention time and by the absorption spectrum. The de-epoxidation state was calculated as (Z+1/2 A)/(Z+A+V) or as (DT)/(DT+DD) (Ruban et al., 2004; Bonente et al., 2011).

Isolation of pigment-protein complexes: Thylakoid membrane isolation and solubilization was conducted following the protocol by Lepetit et al., 2007. Equal amounts of isolated thylakoids, corresponding to 0.5-1 mg of total chlorophyll, were solubilized with n-dodecyl β-D-maltoside (DM, Carl Roth, Germany) at detergent/chlorophyll ratios of 30 corresponding to 3% DM (w/v). The solubilized thylakoids were immediately applied to linear sucrose gradients (from 0 to 0.6 M sucrose (w/v) in isolation medium B complemented with 0.03% DM. Samples were centrifuged for 17 h at 110,000 g using a swing-out rotor. After the separation, sucrose gradient bands were harvested with a syringe and stored for further characterization at -20°C.

Expression analyses: Total RNA were extracted and analyzed by RT-qPCR as described (Taddei et al., 2016). Total proteins were extracted and analyzed by western blot as previously described (Bailleul et al., 2010). Proteins from photosynthetic complexes were analyzed by charging equal amounts of chlorophyll (1 µg), quantified according to Lohr and Wilhelm (Lohr and Wilhelm, 2001). Proteins were detected by specific antibodies: anti-LHCSR (dilution 1: 5000, gift of Prof. G. Peers, University of California, Berkeley, CA, USA), anti-D2 (dilution 1:10,000; gift of Prof. J.-D. Rochaix, University of Geneva, Switzerland), anti-PsaF and anti-ßCF1 for the chloroplastic ATPase (dilution 1:1000 and 1:10,000, respectively, gifts of F.-A. Wollman, Institut de Biologie Physico-Chimique, Paris, France), and anti-LHCF1-11 (dilution 1:2000, gift of Prof. C. Büchel, Institut für Molekulare Biowissenschaften

Universität Frankfurt, Frankfurt, Germany). Densitometry measurements of each protein signal were performed using ImageJ (Schneider et al., 2012). Protein signals in the linear range of detection were adjusted for loading according to the corresponding βCF1 signal, and values were normalized to the value of the WT in the LL condition.

Room-temperature chlorophyll fluorescence measurements: The kinetics of chlorophyll fluorescence yields at room temperature were measured using a fluorescence CCD camera recorder (Speezen1, JBeamBio, France (Johnson et al., 2009)) on cells at $1x10^6$ to $2x10^6$ cells/ml. Before the measurements, all samples were adapted to ambient, dim light for 15 min at 18°C to relax the reaction centers. The Fv/Fm ratio was calculated as (Fm-Fo)/Fm, where Fm and Fo are the maximum and the minimum fluorescence emission levels during a saturating pulse and in the dark, respectively. NPQ was calculated as (Fm-Fm')/Fm' (Bilger and Björkman, 1990), where Fm' is the maximum fluorescence emission level in cells exposed to actinic light, measured with the saturating pulse of light. The maximal NPQ response was measured upon exposure for 10 minutes to saturating 950 µmol $m^{-2}{\cdot}s^{-1}$ green light. Photoinactivation was determined as (1/Fo - 1/Fm) (Park et al., 1995; He and Chow, 2003; Wu et al., 2011; Campbell and Tyystjärvi, 2012). Here, the value (1/Fo - 1/Fm) of LL-acclimated or two days of HL-acclimated cells was taken before starting a short HL treatment (950 µmol $m^{-2}{\cdot}s^{-1}$) and was set to 100%. The percentage of functional PSII was estimated by calculating (1/Fo - 1/Fm) after the 10-min HL treatment and 15 min of darkness.

Low temperature time-resolved fluorescence emission spectra measurements using a streak-camera: Time-resolved emission spectra were recorded using a synchroscan streak-camera system as described (van Oort et al., 2009). An excitation wavelength of 540 nm was used to preferentially excite fucoxanthin (FX) in the antenna, while 400 nm was used to preferentially excite chlorophyll *a* (Chl *a*) in the antenna and the cores. All samples were

452 measured in two different states: the original ("unq") state (10 min of dark adaptation) and
453 the "quenched" state (~10 min of preillumination with white light at ~400 µmol photons m^{-2}
454 s^{-1}). The laser power was 40-60 µW, the time-window was 2 ns, the spot size was 100 µm,
455 and the repetition rate was 250 kHz. An average of 100 images, all measured for 10 s, was
456 used to achieve a high signal/noise ratio. Before analysis, the images were corrected for
457 background signal and detector sensitivity and sliced into traces of 5 nm. The streak-camera
458 images were analyzed as described previously (Chukhutsina et al., 2013) with a singular-
459 value-decomposition (SVD) algorithm (van Stokkum et al., 2004). In short, the total dataset
460 was fitted with the function f (t, λ):

$$f(t,\lambda) = \sum_{1,2\ldots}^{N} DAS_i(\lambda)\, \exp(-\frac{t}{\tau_i}) \oplus i(t)$$

461 where DAS (decay-associated spectra) are the wavelength-dependent amplitude factors
462 associated with decay component i having a decay lifetime τ_i (van Stokkum et al., 2004). The
463 number of significant decay components was determined by the SVD algorithm analysis of
464 the data and was 5 for LL and 4 for HL. A Gaussian-shaped instrument response function
465 (i(t)) was used as input for the analysis with the width as a free-fitting parameter. The full
466 width at half maximum (FWHM) values of this function, obtained from the fitting procedure,
467 were in the range of 28±2 ps. The slowest component was always fixed to 4 ns. Due to the
468 limited time window of our setup, it was not possible to resolve this component in an
469 accurate way, but for the presented analysis the exact value is not important. When we add
470 all 5 DAS for a specific sample and excitation wavelength, then we obtain the fluorescence
471 spectrum immediately after excitation (t=0) before any spectral evolution has taken place in
472 the corresponding wavelength region (unless some processes are too fast to be detected).
473 This can be derived by filling in t=0 in the equation above, which leads to $\exp(-t/\tau_i) = 1$ for all

values of *i.* The resulting t=0 spectra (before the fluorescence decays sets in) do not depend on the state of the cells (unquenched/quenched), as expected. For comparison of fluorescence emission in quenched and unquenched states (Fig. 5), the total fluorescence spectra at t = 0 are normalized to their maximum, while the DAS are scaled accordingly. These scaled DAS were also used to reconstruct the steady-state fluorescence spectra by multiplying the individual, scaled DAS with their corresponding lifetime and taking their weighted sum.

Accession Numbers

Sequence data from this article can be found on the *P. tricornutum* genome browser (annotation Phatr3) on the Ensembl portal (http://protists.ensembl.org/Phaeodactylum_tricornutum/Info/Index) under the following ID numbers: LHCX1: Phatr3_J27278; LHCX2 Phatr3_EG02404; LHCX3: Phatr3_J44733; LHCX4 Phatr3_J38720.

Supplemental Data

Supplemental Figure 1: Representative fluorescence traces used to calculate NPQ values in Figure 1.

Supplemental Figure 2: Growth rate analysis of wild-type (WT) and LHCX1 knock-down (*lhcx1)* lines after a low light (LL) to high light (HL) shift.

Supplemental Table 1. Pigment composition of *P. tricornutum* wild-type (WT) and *lhcx1* knock-down lines grown either in low (LL) or high light (HL) for two days.

Supplemental Table 2. Results of global fitting of the streak-camera data upon 400 nm and 540 nm excitation in unquenched (unq) and quenched (q) states.

Supplemental Table 3. Calculated averaged lifetimes at characteristic wavelengths in *P. tricornutum* wild-type (WT) and *lhcx1* line grown in low light (LL) or high light (HL).

Supplemental Table 4. Photosystem II efficiency in wild type (WT) and *lhcx1* lines.

Figure Legends

Figure 1. *LHCX1* knock-down cells recover their NPQ capacity during prolonged HL exposure. For all the experiments shown in this figure, cells were grown in 12-h-light/12-h-dark cycles either in low light (LL) (30 µmol photons m-2 s-1) or in high light (HL) (500 µmol photons m-2 s-1) for 2 days, following a shift from LL to HL. The samples were taken 2 hours after the onset of light. A and B, NPQ capacity in *P. tricornutum* wild-type (WT, black) and *LHCX1* knock-down (*lhcx1*, white) cells grown under LL (A) or HL (B). Note that different vertical axes were used in panels (A) and (B) to better highlight the differences in NPQ between WT and *lhcx1* cells in LL and HL conditions. Bars indicate +/- standard deviation of five independent experiments. C and D, Accumulation of the different *P. tricornutum* LHCX proteins in WT and *lhcx1* cells detected with an antibody against LHCSR/LHCX. Thirty micrograms from each protein extract were used, and the protein levels were quantified using a serial dilution of proteins from wild-type cells as standard. The relative amount of protein loaded on the gel and the three detected *P. tricornutum* LHCX isoforms (Taddei et al., 2016) are indicated. βCF1 was used as a loading control. The longer exposure time of the membrane in D allowed us to detect the accumulation of the LHCX2 isoform in HL. The vertical lines indicate non-adjacent lanes taken from the same blot. E, Analysis of the relative transcript levels of *LHCX* by RT-qPCR in WT and lhcx1 cells. The *RPS* (ribosomal protein small subunit 30S; Phatr3_J10847) was used as a reference gene, and for each LHCX, the values are relative to the WT level in LL. Bars represent +/- SD of 3 technical replicates.

Figure 2. Localization of the LHCX isoforms in different chloroplast fractions. A, Sucrose density gradient fractionation of solubilized thylakoids from wild-type (WT) and *lhcx1* cells grown in LL and in HL for two days. FP, free pigments; FCP, fucoxanthin chlorophyll binding protein complex; PSI, photosystem I; PSII m, photosystem II monomers; PSII d, PSII dimers. B, Western blot analysis of the proteins extracted from the thylakoids and the FCP, PSII, and PSI fractions and detected with antibodies against LHCSR/LHCX, LHCF (antenna proteins), D2 (PSII), and PsaF (PSI). Grey panels represent no signal detected after hybridization with the indicated antibodies. Samples were loaded at an equal chlorophyll amount (1 µg). CBB, Coomassie Brillant Blue staining of the protein gels.

Figure 3. Time-resolved fluorescence analysis of *P. tricornutum* cells adapted to low light. A and B, Decay-associated spectra (DAS) for wild-type (WT) cells upon a 400-nm excitation (A) and a 540-nm excitation (B) in unquenched (unq, solid lines) and quenched (q, dotted lines) states. C and D, DAS for lhcx1 cells upon a 400-nm excitation (C) and a 540-nm excitation (D) in unquenched (unq, solid lines) and quenched (q, dotted lines) states. Measurements were performed at 77 Kelvin. DAS were calculated as explained in methods.

Figure 4. Time-resolved fluorescence analysis of *P. tricornutum* cells adapted to high light. A and B, Decay-associated spectra (DAS) for wild-type (WT) cells upon a 400-nm excitation (A) and a 540-nm excitation (B) in unquenched (unq, solid lines) and quenched (q, dotted lines) states. C and D, DAS for *lhcx1* cells upon a 400-nm excitation (C) and a 540-nm excitation (D) in unquenched (unq, solid lines) and quenched (q, dotted lines) states. Measurements were performed at 77 K. DAS were calculated as explained in methods.

Figure 5. Reconstructed steady-state emission spectra in unquenched (solid line) and quenched (dashed line) states at 77 K of LL-adapted (first row) and HL-adapted (second row)

cells. Excitation wavelengths and analysed strains are indicated in every figure panel. Spectra were reconstructed as explained in methods.

Figure 6. Physiological analysis of wild-type (WT) and *lhcx1* cells. A and B, Net photosynthesis calculated from the oxygen evolution rates minus oxygen consumption measured with a Clark electrode at different light intensities (0, 90, 200, 450, 750, and 2300 µmol photons m^{-2} s^{-1}). WT (A) and *lhcx1* (B) cells were grown in low light (LL, 30 µmol photons m^{-2} s^{-1}) or high light (HL, 500 µmol photons m^{-2} s^{-1}) for two days, following a shift from LL to HL. Bars indicate the standard deviation of three biological replicates. C, Western blot analysis of total protein extracts (30 µg) from cells grown in the same condition as in A and B. Antibodies against LHCSR/LHCX, LHCF, D2, PsaF, and βCF1 were used for protein detection. D, Densitometric analysis of D2 and PsaF obtained from independent western blot analyses (three independent biological experiments) from WT and *lhcx1* cells grown as in (C). Signals for D2 and PsaF were adjusted according to those of βCF1, used as loading control, and normalized on the WT LL signal.

Figure 7. Model for NPQ in *P. tricornutum* wild-type (WT) and *lhcx1* cells adapted to low light (LL) and after short and long exposures to high light (HL). In LL-grown WT cells experiencing a short HL-treatment (from seconds to minutes), the major quenching site is close to the reaction centre (PSII QS, Q2). *lhcx1* cells show a reduced quenching capacity because of the reduced content of LHCX1, the highly expressed isoform in LL. After prolonged HL treatment (days), the quenching sites are mainly in the FCP red-shifted antenna (Antenna QS, Q1). The *lhcx1* line recovers its NPQ capacity. Because of the similar quenching capacity in WT and *lhcx1* cells, this quenching could be related to LHCX3, which is highly induced in HL and is detected in the FCP fraction.

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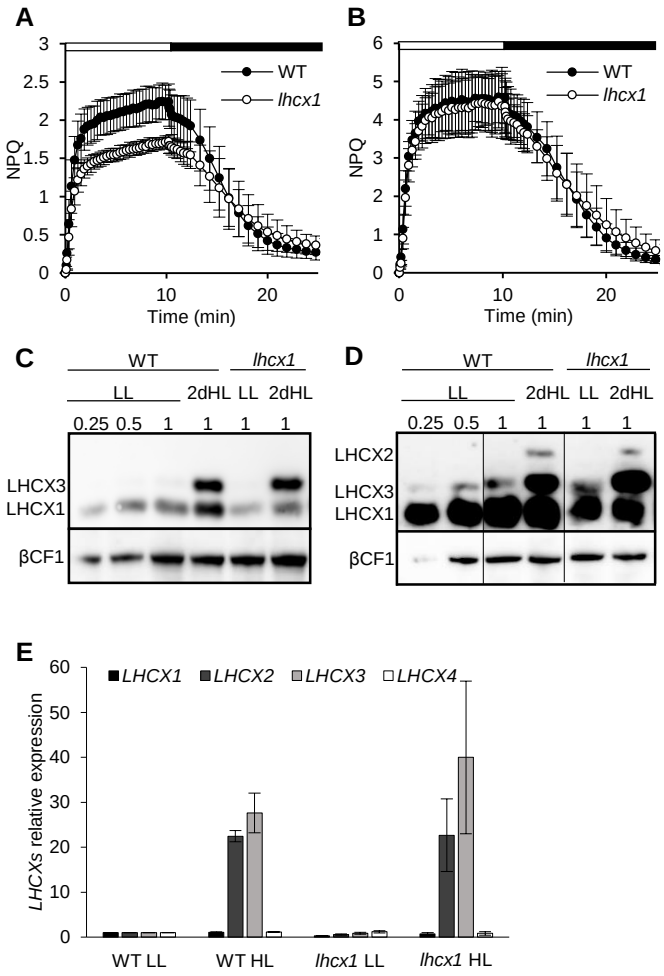
Figure 1 – Taddei et al.



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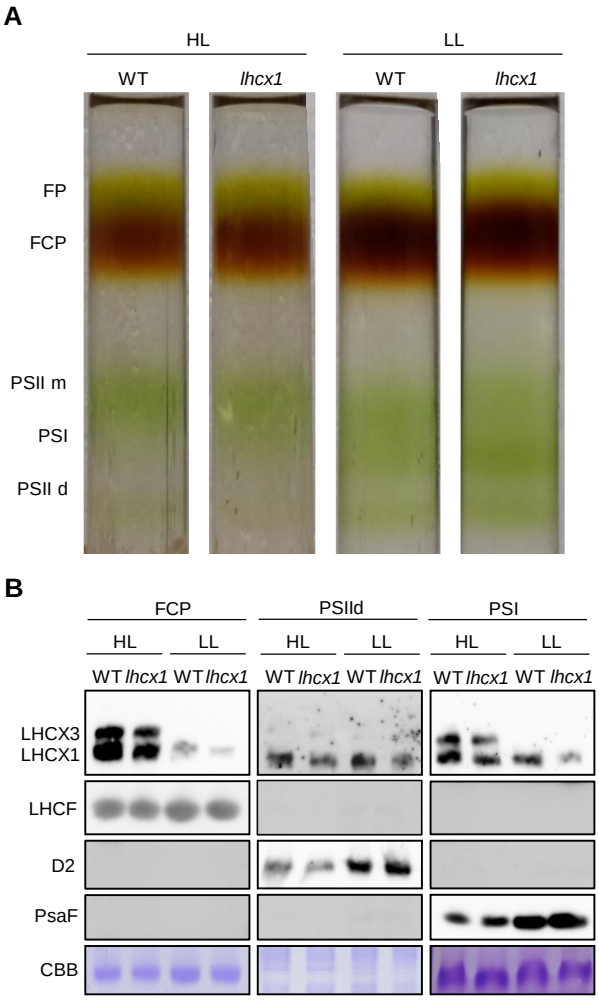

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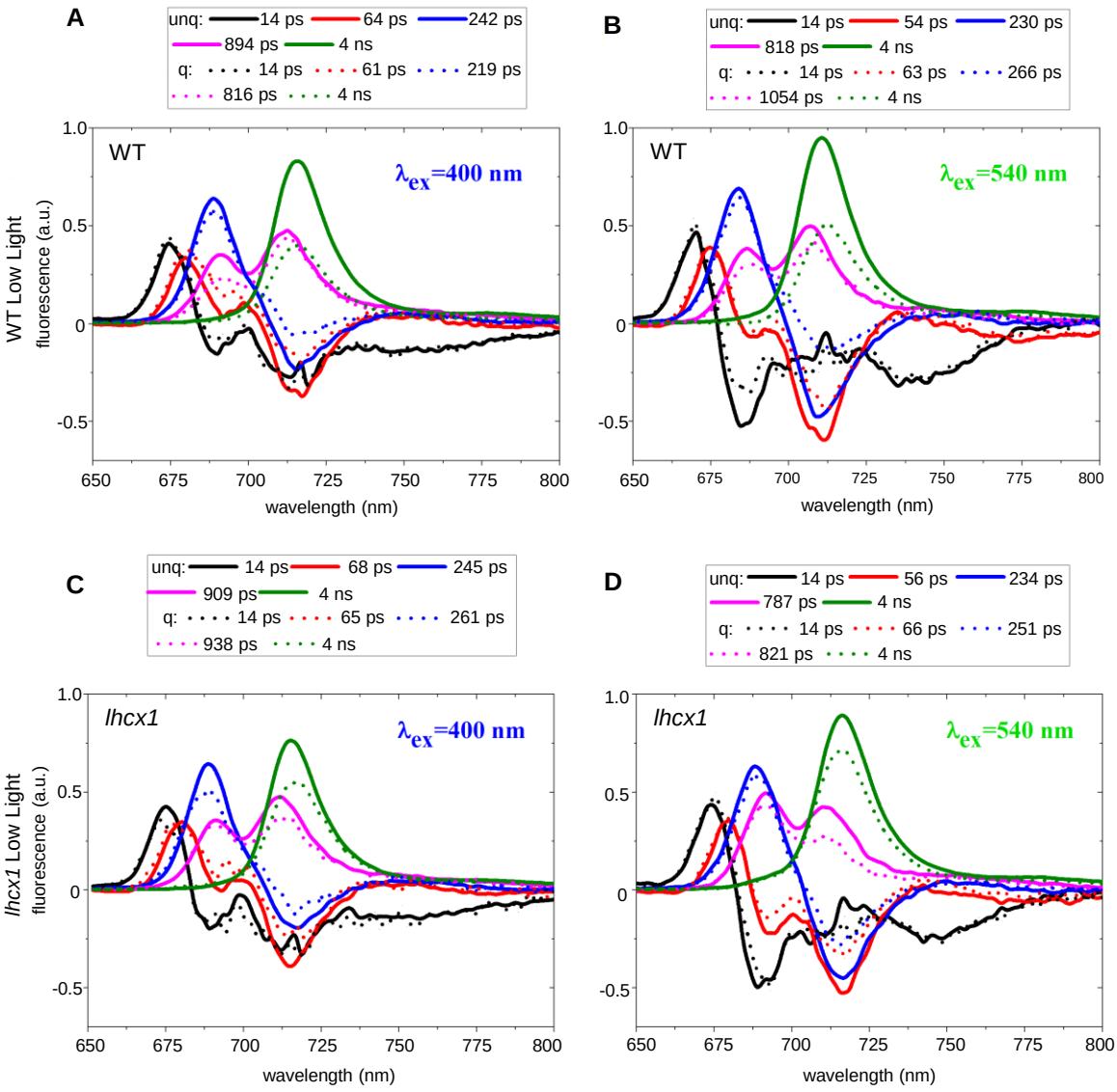

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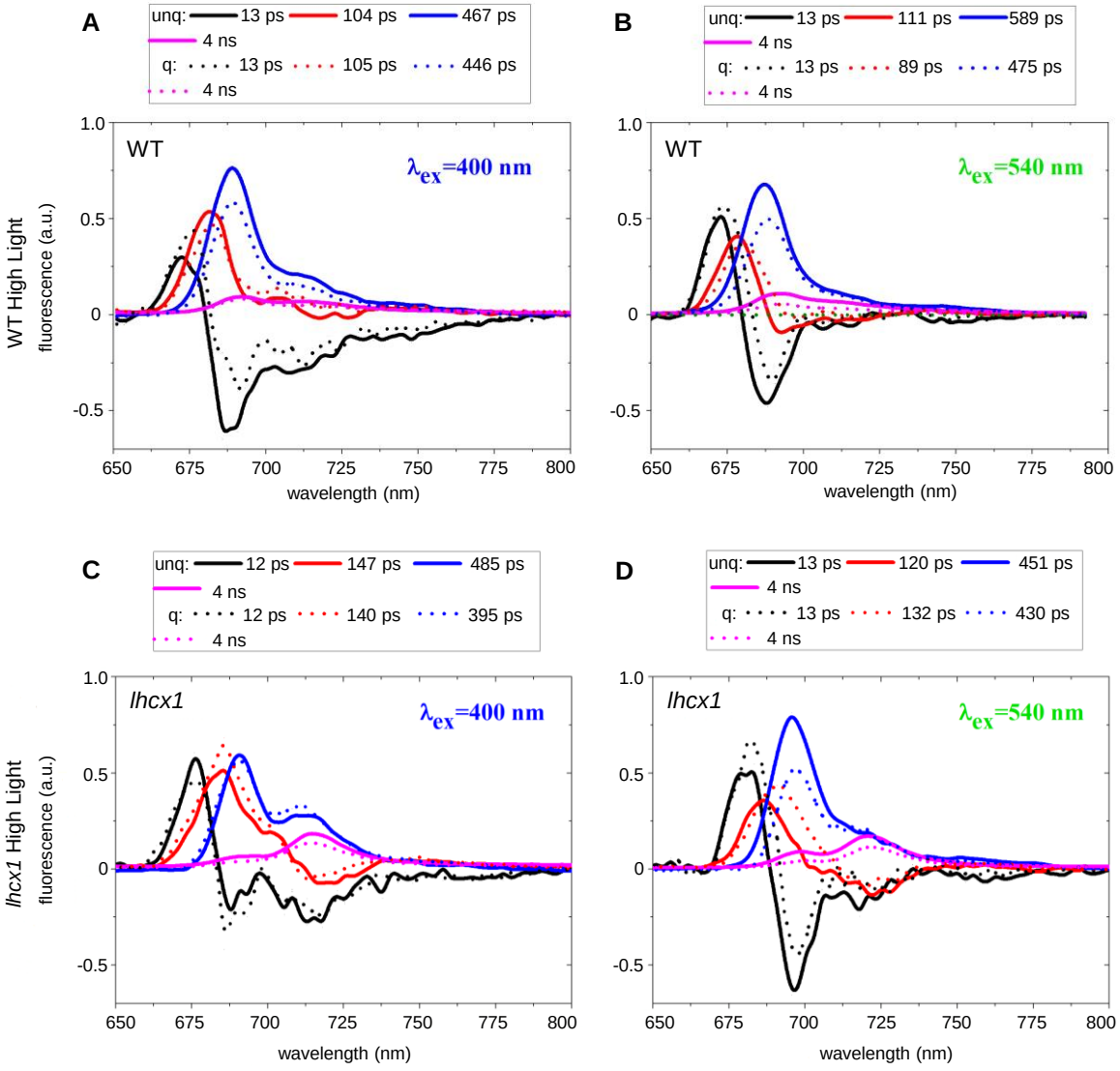

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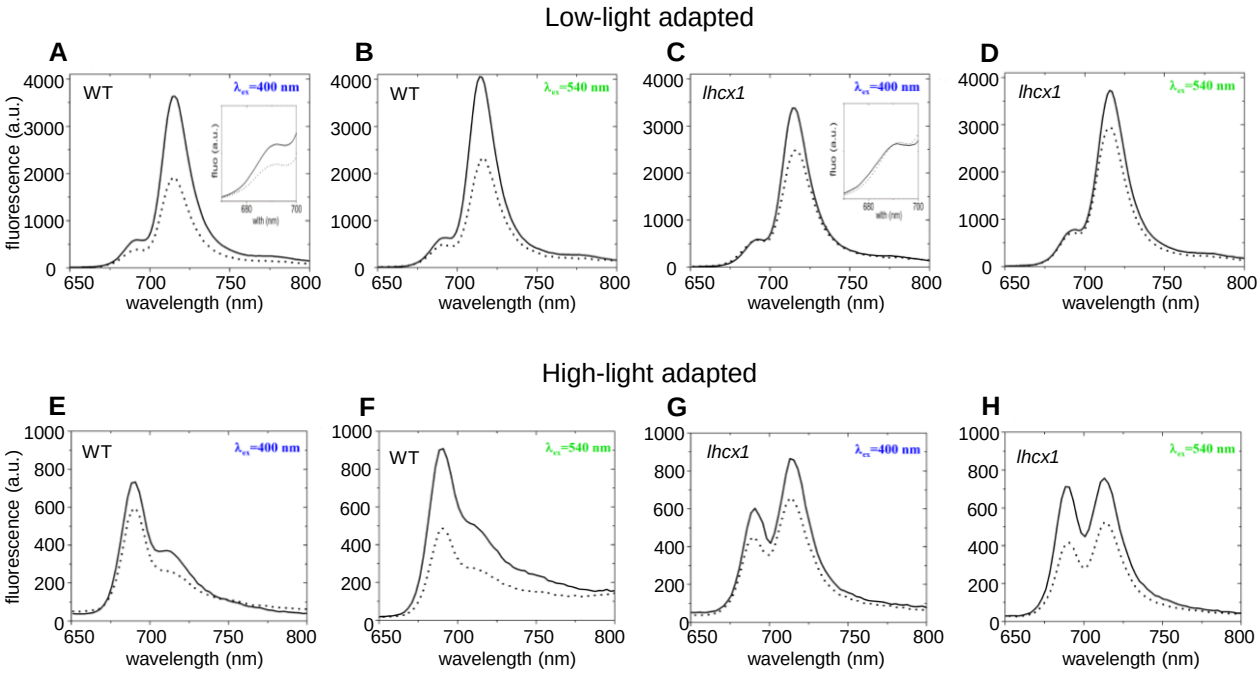

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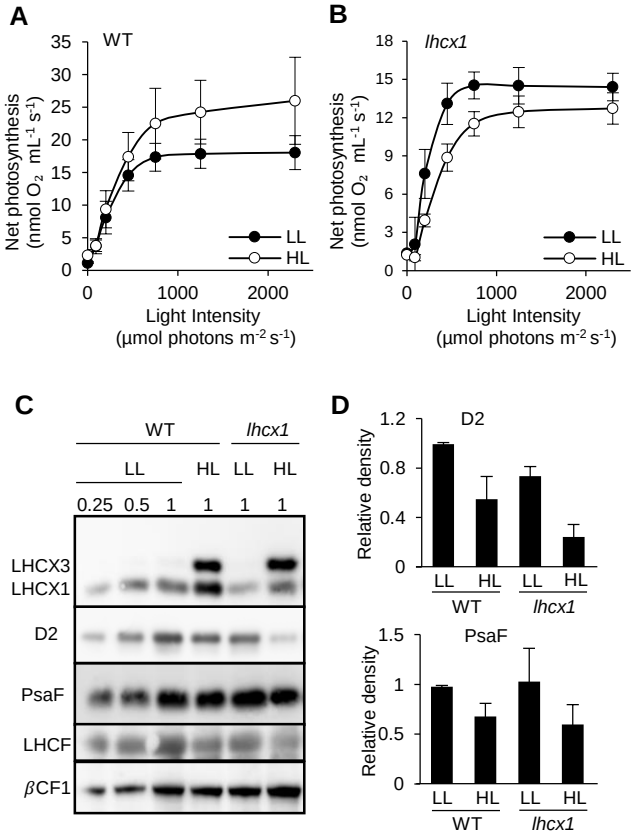

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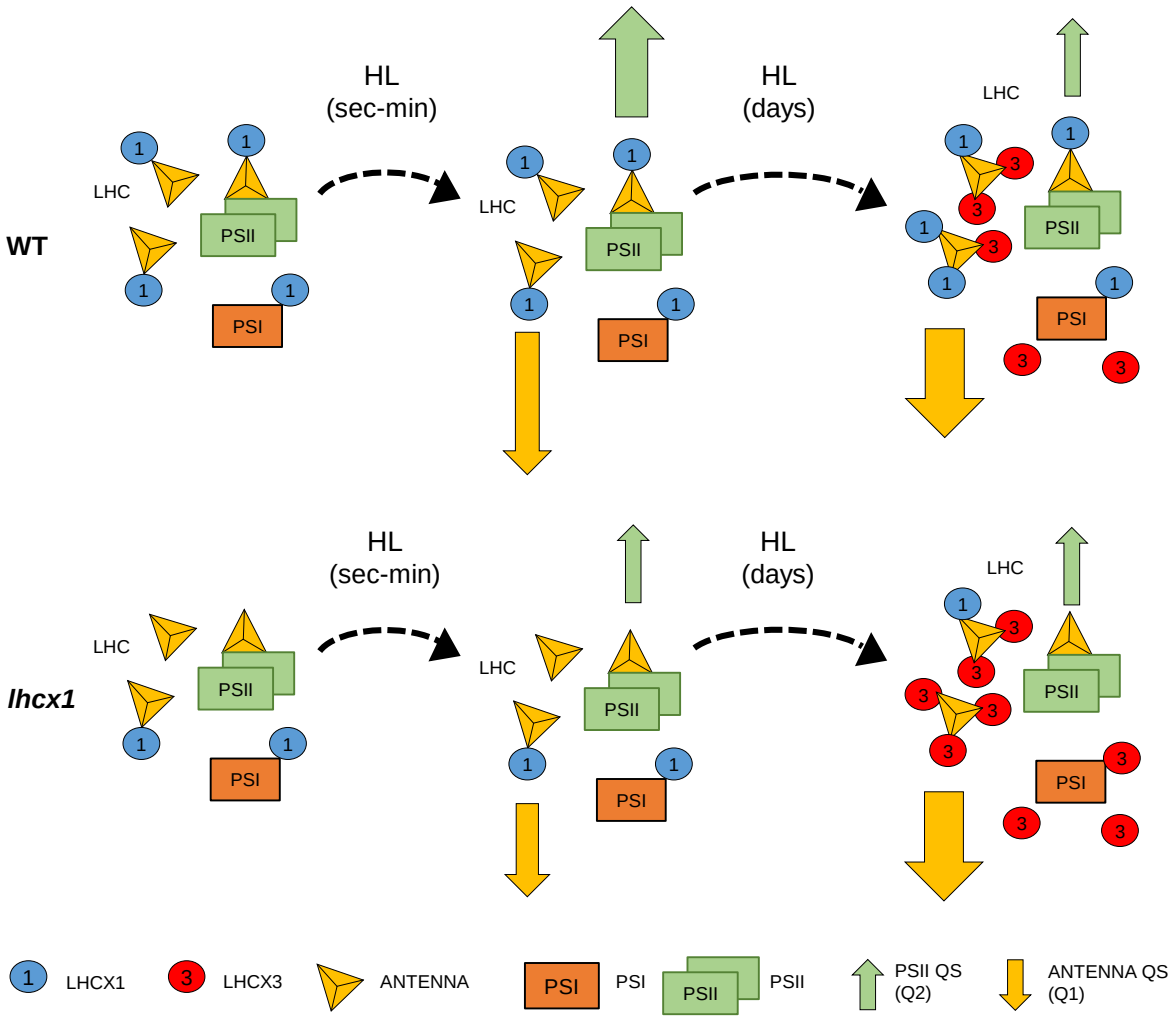

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