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The \( \alpha_2\delta-1 \) subunit remodels \( \text{CaV}1.2 \) voltage sensors and allows \( \text{Ca}^{2+} \) influx at physiological membrane potentials

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Excitation-evoked calcium influx across cellular membranes is strictly controlled by voltage-gated calcium channels (\( \text{CaV} \)), which possess four distinct voltage-sensing domains (VSDs) that direct the opening of a central pore. The energetic interactions between the VSDs and the pore are critical for tuning the channel’s voltage dependence. The accessory \( \alpha_2\delta-1 \) subunit is known to facilitate \( \text{CaV}1.2 \) voltage-dependent activation, but the underlying mechanism is unknown. In this study, using voltage clamp fluorometry, we track the activation of the four individual VSDs in a human L-type \( \text{CaV}1.2 \) channel consisting of \( \alpha_{1C} \) and \( \beta_1 \) subunits. We find that, without \( \alpha_2\delta-1 \), the channel complex displays a right-shifted voltage dependence such that currents mainly develop at nonphysiological membrane potentials because of very weak VSD–pore interactions. The presence of \( \alpha_2\delta-1 \) facilitates channel activation by increasing the voltage sensitivity (i.e., the effective charge) of VSDs I–III. Moreover, the \( \alpha_2\delta-1 \) subunit also makes VSDs I–III more efficient at opening the channel by increasing the coupling energy between VSDs II and III and the pore, thus allowing \( \text{Ca}^{2+} \) influx within the range of physiological membrane potentials.

\textbf{INTRODUCTION}

Calcium influx through voltage-activated calcium (\( \text{CaV} \)) channels translates electrical signals into a variety of physiological outcomes such as cell contraction, neurotransmitter or hormonal release, and gene expression. The specificity of the \( \text{Ca}^{2+} \) signal relies on the activity of the \( \text{CaV} \) channel complex being perfectly tuned to voltage signals. \( \text{CaV} \) channels are multimeric proteins formed by the pore-forming \( \alpha_1 \) subunit and at least three auxiliary subunits, \( \beta, \alpha_2\delta, \text{and calmodulin, in a 1:1:1:1 stoichiometry, resulting in an asymmetric structural architecture (Fig. 1; Findeisen and Minor, 2010; Catterall, 2011; Dolphin, 2013; Ben-Johny and Yue, 2014; Neely and Hidalgo, 2014; Campiglio and Flucher, 2015; Wu et al., 2015). The \( \alpha_2\delta \) auxiliary subunit is a large (\~{}170 kD), mostly extracellular protein with a single membrane-anchoring segment (Davies et al., 2010) that binds to the \( \alpha_1 \) subunit from the extracellular side (Cassidy et al., 2014). \( \alpha_2 \) and \( \delta \) proteins are the products of the same gene as a preprotein that posttranslationally proteolysed and then linked by a disulfide-bond to form the mature \( \alpha_2\delta \) protein (Calderón-Rivera et al., 2012). Four genes (\text{CACNA2D1}–\text{4}) encode for distinct \( \alpha_2\delta \) isoforms (\( \alpha_2\delta-1–4 \)), which are all expressed in the brain (Dolphin, 2013). In addition to brain tissue, \( \alpha_2\delta-1 \) is strongly expressed in cardiac, smooth, and skeletal muscles, whereas \( \alpha_2\delta-4 \) is found in endocrine tissues and the retina. Mutations in the \( \alpha_2\delta-1 \) gene can lead to Brugada (Burashnikov et al., 2010) and short QT (Templin et al., 2011; Bourdin et al., 2015) syndromes and are associated with epilepsy and mental disability (Vergult et al., 2015). In mice, naturally occurring mutations in the \( \alpha_2\delta-2 \) gene lead to ataxia and epilepsy (Barclay et al., 2001), whereas the \( \alpha_2\delta-3 \) protein is important for synaptic morphogenesis (Kurshan et al., 2009) and nociception (Neely et al., 2010). Mutations in \( \alpha_2\delta-4 \) can result in night blindness (Wycisk et al., 2006). Moreover, \( \alpha_2\delta-1 \) and -2 have been identified as the molecular targets of gabapentinoid drugs (such as gabapentin and pregabalin), mediating their analgesic action in neuropathic pain (Field et al., 2006; Hendrich et al., 2008; Uchitel et al., 2010). Finally, it has been shown that \( \alpha_2\delta \) proteins also play an important role in synapse formation (Eroglu et al., 2009).

Several studies report that the interaction of \( \alpha_2\delta \) with the pore-forming \( \alpha_{1C} \) subunits (L-type \( \text{CaV}1.2 \)) favors channel activation, as manifested by a hyperpolar-
The $\alpha_2\delta$-1 subunit remodels CaV1.2 voltage sensors | Savalli et al.

The $\alpha_2\delta$-1 subunit remodels CaV1.2 voltage sensors | Savalli et al.

izing shift of channel opening (Felix et al., 1997; Platano et al., 2000; Bourdin et al., 2015) and an accelerated time course of activation (Bangalore et al., 1996). Accordingly, channel activation is diminished with $\alpha_2\delta$-1 down-regulation (Tuluc et al., 2007; Fuller-Bicer et al., 2009) and enhanced with $\alpha_2\delta$-1 up-regulation (Li et al., 2006). Thus, by facilitating channel opening, $\alpha_2\delta$-1 allows CaV1.2 channels to operate at physiological membrane potentials. However, the molecular mechanism by which $\alpha_2\delta$-1 facilitates CaV1.2 activation is as yet poorly understood.

Because $\alpha_2\delta$-1 modulates the voltage-dependent properties of CaV1.2 channels (Felix et al., 1997; Platano et al., 2000; Bourdin et al., 2015) and associates with $\alpha_{1C}$ subunits asymmetrically (Walsh et al., 2009a), we hypothesized that $\alpha_2\delta$-1 differentially modulates each of the four voltage-sensing domains (VSDs), as well as their contribution to channel opening. In fact, the pore-forming $\alpha_{1C}$ subunit consists of four homologous, but nonidentical, concatenated repeats (I–IV), each composed of a VSD (transmembrane helices S1–S4) and a quarter of the pore domain (S5–S6; Catterall, 2011). Using voltage clamp fluorometry (VCF), we have recently revealed the functional heterogeneity of the four CaV1.2 VSDs, whereby each undergoes structural changes during channel activation, with unique voltage- and time-dependent properties, such that the activation of VSDs II and III, and to a lesser extent VSD I, energetically contributes to channel opening (Pantazis et al., 2014). VCF is a powerful investigative tool that allows for simultaneous measurements of ionic current kinetics and structural rearrangements occurring within specific protein domains, the latter tracked using environmentally sensitive fluorophores (Claydon and Fedida, 2007; Gandhi and Olcese, 2008; Talwar and Lynch, 2015; Zhu et al., 2016). VCF has been a successful approach in the study of numerous voltage-sensitive proteins (Mannuzzu et al., 1996; Cha et al., 1999; Smith and Yellen, 2002; Savalli et al., 2006; Kohout et al., 2008; Osteen et al., 2010; Tombola et al., 2010), transporters (Meinnild et al., 2002; Larsson et al., 2004; Ghezzi et al., 2009), and receptors (Dahan et al., 2004; Lörinczi et al., 2012). Using VCF, auxiliary subunit modulation of VSDs has also been detected in ion channels, such as BK channels (Savalli et al., 2007) or Kv7.1 (Ruscic et al., 2013). However, VCF has only recently been adapted to investigate CaV channels (Pantazis et al., 2014).

In this study, by using VCF and a structurally relevant allosteric model of CaV1.2 activation, we show that the $\alpha_2\delta$-1 auxiliary subunit (a) facilitates the voltage-dependent activation of CaV1.2 VSDs I–III; (b) accelerates VSD I kinetics; and (c) increases the energetic contribu-
tion of VSDs I–III to pore opening. These results unravel the molecular mechanisms by which αδ-1 exerts its modulation on Cav1.2 channel activation, allowing for Ca\(^{2+}\) influx to occur in excitable cells at physiological membrane potentials.

**MATERIALS AND METHODS**

**Molecular biology**

Human α1C.77 subunits (GenBank accession no. CAA84346; Soldatov, 1992) of Cav1.2 channels, with a Cys substituted at an extracellular position in the S3–S4 linker of each VSD at a time, were used ((F231C, L614C, V994C, or S1324C for VSDs I–IV, respectively) as previously described (Pantazis et al., 2014). Single-point mutations were generated using the QuikChange Site-Directed Mutagenesis kit (Agilent Technologies) and confirmed by sequencing. Auxiliary subunits αβ (UniProt accession no. P13806) and β3 (UniProt accession no. P54286) were also coexpressed. The cRNA of the different subunits was transcribed in vitro (mMES SAGE MACHINE; Ambion) and injected into stage VI Xenopus laevis oocytes (50 nl at 0.1–0.5 µg/µl).

**VCF**

3–4 d after injection, oocytes were incubated with thiol-reactive fluorophores sensitive to environmental changes (10 µM tetramethylrhodamine-5-maleimide [TMRM] for VSD II and 20 µM 2-((5(6)-tetramethylrhodamine)carboxyamino)ethyl methanethiosulfonate [MTS-TAMRA] for VSD I, III, or IV) in a depolarizing solution (120 mM K-methanesulfonate [MES], 2 mM Ca(MES)\(_2\), and 10 mM HEPES, pH 7.0). Subsequently, oocytes were voltage clamped using the cut-open oocyte technique (Stefani and Bezanilla, 1998; Pantazis and Olcese, 2013). Fluorescence changes and ionic currents were acquired simultaneously from the same membrane area (Gandhi and Olcese, 2008; Pantazis and Olcese, 2013). External solution was 2 mM Ba(MES)\(_2\), 120 mM NaMES, and 10 HEPES, pH 7.0, supplemented with 0.1 ouabain to eliminate charge movement from Na/K ATPase (Neely et al., 1994). Internal solution was 120 mM K-glutamate and 10 mM HEPES, pH 7.0. Pipette solution was 2.7 M Na-MES, 10 mM NaCl, and 10 mM Na-HEPES, pH 7.0. Before experiments, oocytes were injected with 10 mM BAPTA•IK, pH 7.0, to prevent activation of native Ca\(^{2+}\)- and Ba\(^{2+}\)-dependent Cl\(^-\) channels (Barish, 1983).

**Data analysis**

The voltage dependence of ionic conductance (G(V), estimated from the peaks of the tail currents) and fluorescence changes (F(V)) were empirically characterized by fitting to one or two Boltzmann functions as \(f(V) = [1 + \exp(q \cdot (V_{half} - V_m) / F(RT))]^{-1}\), where \(q\) is the effective charge, \(V_{half}\) is the half-activation potential, \(V_m\) is the membrane potential, \(T\) is the absolute temperature, and \(F\) and \(R\) are the Faraday and Gas constants, respectively. F(V) curves can be satisfactorily described by single Boltzmann functions both the absence and the presence of the αδ-1 subunit (see Figs. 4 and 5).

The time course of fluorescence onset (VSD activation) was fitted in background-subtracted fluorescence traces to the sum of two exponential components:

\[ f(t) = B + \sum_{i=1}^{\infty} A_i \cdot \exp(-t/\tau_i), \]

where \(B\) is the baseline, \(A_i\) is the amplitude, \(t\) is time, and \(\tau\) is the time constant. Fitting was performed in Matlab (MathWorks) by least squares (Optimization Toolbox). Only traces with sufficient signal-to-noise ratio (S:N ≥2) were included in the kinetics statistics. S:N is defined as mean signal amplitude divided by the root mean square. Fractional amplitude–weighted time constants (\(\tau_{avg}\)) were calculated using

\[ \tau_{avg} = \sum_{i=1}^{\infty} \alpha_i \cdot \tau_i, \]

where \(\alpha_i = \frac{A_i}{A_1 + A_2}\).

Data are reported as mean ± SEM; statistical analysis was performed using Excel (Microsoft).

**Allosteric model**

Modeling Cav1.2 kinetics and activation curves through a five-particle allosteric scheme was performed as described previously (Pantazis et al., 2014). In brief, equilibrium states were determined from the values of five particle equilibrium constants (\(L, J_1–4\)) and four VSD–pore coupling constants \(D_1–4\) using the channel partition function:

\[ Z = \frac{1}{\prod_{i=1}^{\infty} (1 + J_i)(1 + J_i)(1 + J_i)(1 + J_i)(1 + J_i)(1 + J_i)(1 + J_i)(1 + J_i)(1 + J_i)(1 + J_i)}, \]

The pore particle (\(L\)) derives its voltage dependence through a gating charge displacement \(\Delta q_l\) and a characteristic midpoint voltage \(V_{1/2}\); for example, \(L = \exp(\Delta q_l(V - V_{1/2})/kT)\). Similar expressions applied to \(J_1–4\) were used to describe intrinsic VSD activation. The four allosteric factors \(D_1–4\) are related to VSD–pore interaction energies \(W_{1/4}\) through \(D_i = \exp(-W_i/kT)\). The equilibrium curves (\(k = \langle L_j \rangle\) for the five gating particles are easily derived from the partition function through the relations \(\langle L \rangle = \partial \ln Z / \partial \ln K\), which were used to fit the experimental conductance (G(V)) and fluorescence (F(V)) curves.

A kinetic model of channel activation that reduces to the thermodynamic model under equilibrium conditions was obtained by assigning two additional variables...
for each particle transition: a frequency factor $\nu$ and a fractional position $x$ of the transition barrier between resting and active states (Sigg, 2014). The forward and backward rate constants for the transition between a configuration $i$ and any of the accessible configurations $j$ after the activation of one of the five gating particles were expressed as

$$\alpha_{i,j} = \nu_k(Z_iZ_j)^{\beta}$$

and

$$\beta_{i,j} = \nu_A(Z_iZ_j)^{1-\beta},$$

where $k$ refers to the transitioning particle and $Z_i$ and $Z_j$ are the configuration-specific contributions to the overall partition function $Z$ (obtained by expanding the earlier expression of $Z$ into its 32 terms).

The channel kinetics were solved by integrating $p(t) = \tilde{p}(0) \cdot \exp(\tilde{Q}t)$, which describes state probabilities of all states ($p$). $Q$ is the standard rate matrix as described in Colquhoun and Hawkes (1981). The initial condition $p(0)$ for the holding potential was obtained from $\tilde{p}$ ($t \to \infty$). The time dependence of a quantity of interest $A$ (ionic current or fluorescence) was obtained from

$$\langle A(t) \rangle = N\sum p(t) a_i,$$

where $a_i$ is the value of the desired quantity at configuration $i$.

To find the set of parameters that best described the data, several approaches were used such as Marquadt–Levenberg, implemented in Berkeley Madonna, and Nedler–Mead, developed in Matlab, minimizing the error function “$\chi^2$” generated by the weighed sum of 10 error functions, five for steady-state voltage dependencies and five for time-dependent signals. Each individual error function corresponds to the sum of the squares of the difference between the experimental and simulated datasets normalized by $n$ and the square of the maximum value.

To test for the uniqueness of the solution and estimating the 95% credible interval of each parameter, we used a Bayesian approach using Markov chain Monte Carlo (MCMC) sampling as in Hines et al. (2014), but instead of using likelihood ratio to test for high posterior probabilities, we used

$$a = \min \left[1, \exp\left(\frac{\chi^2_{\text{calc}} - \chi^2_{\text{null}}}{2}\right)\right].$$

Then, transitions of the Markov chain were accepted with probability $a$, as described in Li (2012). To further constrain the solution space and take better advantage of the time-dependent data, we added a set of penalty functions as further explained in the supplementary figure legends.

Online supplemental material
Fig. S1 shows representative Ba$^{2+}$ current traces from Xenopus oocytes expressing CaV1.2 channel complexes formed by $\alpha_{1C}$ + $\beta_3$ subunits. Fig. S2 shows histograms of posterior distributions of 14 parameters obtained from a 100,000-trial MCMC run. Fig. S3 shows histograms of posterior distributions of parameters $x_i$ and $n_i$ ($i = 1, 1, 2, 3, 4$). Online supplemental material is available at http://www.jgp.org/cgi/content/full/jgp.201611586/DC1.

RESULTS
$\alpha_2\delta-1$ facilitates the voltage-dependent activation of human CaV1.2 channels
To understand the mechanism of $\alpha_2\delta-1$ subunits modulation of human CaV1.2 channels, we expressed in Xenopus oocytes CaV1.2 channels consisting of $\alpha_{1C}$ and $\beta_3$ subunits, in the presence or the absence of $\alpha_2\delta-1$ proteins. We voltage clamped the cells using the cut-open oocyte voltage clamp technique (Stefani and Bezanilla, 1998; Pantazis and Olcese, 2013) and recorded ionic currents (Fig. 2, A and B) using Ba$^{2+}$ as the charge carrier to prevent calcium-dependent inactivation (Peter–son et al., 1999; Qin et al., 1999). Because CaV1.2 channels lacking $\alpha_2\delta-1$ subunits activate slowly (Fig. 2 A), relatively longer depolarizations were necessary for ionic current to reach quasi–steady state (Fig. S1), a condition necessary to construct conductance versus voltage relationships (G(V)) from tail currents. We observed that $\alpha_2\delta-1$ coexpression strongly facilitated channel opening in human CaV1.2 channels by shifting the CaV1.2 half-activation potential ($V_{\text{half}}$) of the G(V) curves by ~50 mV toward more hyperpolarized potentials (Fig. 2 C), in agreement with data previously obtained from the rabbit isoforms (Felix et al., 1997; Platano et al., 2000; Bourdin et al., 2015). G(V) curves obtained from channels expressed with the full complement of auxiliary subunits were well described by the sum of two Boltzmann functions with distinct voltage-dependent properties ($V_{\text{half}} = -4.02 \pm 0.38 \text{ mV}$, $z_1 = 3.46 \pm 0.11 \text{ e}^0$, $G_1 = 57.56 \pm 4.82\%$; $V_{\text{half}} = 42.56 \pm 1.87 \text{ mV}$, $z_2 = 1.27 \pm 0.04 \text{ e}^0$, $G_2 = 42.44 \pm 4.82\%$; $n = 4$; Fig. 2 C), alluding to a complex voltage-dependent activation mechanism with more than one voltage-dependent opening transitions; in contrast, G(V) curves for channels lacking $\alpha_2\delta-1$ were well accounted for by a single Boltzmann distribution ($V_{\text{half}} = 68.01 \pm 1.32 \text{ mV}$ and $z = 1.19 \pm 0.01 \text{ e}^0$; $n = 7$), which is a tentative (yet tantalizing) indication that these channels gate in a two-state process. We further mechanistically evaluated this premise using an allosteric model of voltage-dependent CaV activation.

$\alpha_2\delta-1$ increases the rate of VSD I activation
Given the large difference (>50 mV) in the voltage dependence of CaV1.2 activation in the presence or ab-
sence of αδ-1 subunits (Fig. 2), we tested the hypothesis that αδ-1 association with α1C induces a functional re-modeling of one or more VSDs, altering their gating properties. We used the VCF technique (Mannuzzu et al., 1996; Cha and Bezanilla, 1997; Gandhi and Olcese, 2008) to track the molecular rearrangements of the individual VSDs of human CaV1.2 channels in the presence or absence of αδ-1. Briefly, this involves the introduction of a cysteine residue one at a time at a strategic and specific position extracellular to the S4 helix in each CaV1.2 VSD, as shown in our previous study (Pantazis et al., 2014). In voltage-gated ion channels, the S4 segment typically contains the positively charged amino acids effectively responsible for voltage sensing and undergoes structural rearrangements during depolarizations (Tombola et al., 2006; Bezanilla, 2008; Chanda and Bezanilla, 2008; Swartz, 2008; Catterall, 2010; Palovcak et al., 2014). CaV1.2 channels (α1C + β3 subunits) were expressed with or without αδ-1 subunits in Xenopus oocytes. After labeling of the cysteines with thiol-reactive fluorophores that are sensitive to the environment, we used VCF to simultaneously study the voltage-dependent activation of the pore (ionic current) and each of the four VSDs (fluorescence) in conducting CaV1.2 channels. The labeling positions and fluorophores used here were the same as in our previous work (Pantazis et al., 2014). The effect of αδ-1 subunits on the kinetics of VSD activation was quantified for 100-ms depolarizations to 20 mV (Table 1). The activation of VSD I was accelerated by approximately twofold by the αδ-1 subunit, increasing the fractional amplitude of the fast component of activation (Fig. 3 and Table 1). This result suggests that the acceleration of ionic current by the αδ-1 subunit (Fig. 2, A and B; Felix et al., 1997; Plata-

tano et al., 2000; Tuluc et al., 2007) may result from a faster VSD I, consistent with the findings of Nakai et al. (1994), who demonstrated a relevant role for VSD I in controlling CaV1.1 VSD I sequences into the corresponding location in CaV1.2. In contrast, the kinetics of VSDs II–IV were practically unaffected by αδ-1 (Fig. 3 and Table 1).

αδ-1 facilitates the voltage-dependent activation of VSDs I, II, and III

To assess how the voltage dependence of the individual VSDs was affected by αδ-1 subunit association, we constructed activation curves (F(V)) from the corresponding fluorescence intensity at the end of 100-ms pulses
over a wide range of membrane potentials (Fig. 4). The F(V) values of VSDs I–III were shifted to more negative potentials in the presence of the α2δ-1 subunit, whereas VSD IV activation was unaffected (Fig. 4, A–D). Moreover, VSDs I–III exhibited a steeper slope of voltage-dependent activation: the effective charge q increased by approximately twofold (Fig. 4, A–C; and Table 2). Overall, in the absence of the α2δ-1 subunit, the activation curves of the four VSDs were highly disparate, spread over a voltage range spanning ∼90 mV (Fig. 5 A and Table 2), while the variance of the Vhalf values was 1,300 mV². The association of α2δ-1 subunit narrowed the range of membrane potentials at which VSDs activated (∼50 mV; Table 2, Vhalf variance: 520 mV²) and shifted the G(V) such that VSD voltage dependence was closer to pore opening (Fig. 5 B). A separation of the half activation potential of VSD activation and channel opening can be interpreted as decreased coupling between VSD activation and pore gating (Sigg, 2014). Taken together, these results are consistent with the view that α2δ-1 is required to increase the coupling between VSDs I–III and the CaV1.2 pore. In addition, because the effective charge q is the summed displacements of residue charges across the membrane potential profile, we cannot exclude that α2δ-1 association also increases CaV1.2 voltage sensitivity by altering the shape of the profile (for example, making it steeper in the region of charge translation). To discriminate among these mechanisms (increased coupling, increased effective charge, or both), we modeled CaV1.2 activation with an allosteric model used previously (Pantazis et al., 2014).

α2δ-1 facilitates CaV1.2 activation by increasing the energetic contribution of VSDs I–III to pore opening

We analyzed the VCF data with the 32-state allosteric model for CaV1.2 activation (Pantazis et al., 2014), consisting of five gating elements (one pore, four VSDs; Fig. 6 A) and therefore relevant to CaV1.2 architecture. Pore and VSDs can exist in two states: closed-open and resting-active, each undergoing voltage-dependent transitions. Thus, in this model, the pore as well as the VSDs are intrinsically voltage dependent (half-activation potential V and charge displacement q). The activation of one or more VSDs stabilizes the open state of the pore through energy coupling with magnitude W_i (i = 1–4).

Kinetic and quasi-equilibrium data from the pore (ionic currents) and each VSD (fluorescence) were simultaneously fitted in the absence of α2δ-1 with no assumption or constraint. The model accurately accounts
for the voltage- and time-dependent properties of channels composed of $\alpha_{1C} + \beta_3$ subunits (Fig. 6, B and C). The most salient feature of the fitted quantities is that the energetic contribution to pore opening ($W$) of each VSD is small (<1 kT or 25 meV). A comparison of $W$ values with and without the $\alpha_2\delta^{-1}$ subunit demonstrates a doubling of the energetic contribution to pore opening by VSDs I and III ($W_1$ and $W_3$) and an approximately three-fold increase of $W_2$ in the presence of $\alpha_2\delta^{-1}$, whereas the contribution of VSD IV ($W_4$) was practically unchanged (Table 3; parameters with $\alpha_2\delta^{-1}$ are from Pantazis et al., 2014 and reported here for clarity). In addition to enhancing VSD I–III energetic contributions to pore opening, $\alpha_2\delta^{-1}$ increased the charge displacement ($q$) of VSDs I and II by $\sim 140\%$ and $\sim 200\%$, respectively (Table 3). In contrast, the intrinsic pore parameters $q_L$ and $V_L$ varied minimally with the addition of $\alpha_2\delta^{-1}$ to the channel. Thus, $\alpha_2\delta^{-1}$ modulates the CaV1.2 channel by exerting its effect on the VSDs (VSDs I–III) rather than on the pore.

### DISCUSSION

The $\alpha_3$ subunit of voltage-gated CaV channels is a modular, pseudotetrameric protein consisting of a central pore domain coupled to four homologous but not identical VSDs. Several auxiliary subunits, including $\alpha_2\delta$ and $\beta$, associate with the $\alpha_1$ subunit in a 1:1:1 ratio (Catterall, 2011; Wu et al., 2015) to regulate channel trafficking and biophysical properties (Fang and Colecraft, 2011; Dolphin, 2012; Neely and Hidalgo, 2014; Campiglio and Flucher, 2015). Using VCF to optically track the individual VSDs in a human CaV1.2 $\alpha_1 + \beta_3$ complex with and without the auxiliary $\alpha_2\delta^{-1}$ subunits, we gained a mechanistic understanding of $\alpha_2\delta^{-1}$–mediated facilitation of CaV1.2 activation. We found that $\alpha_2\delta^{-1}$ alters the biophysical properties of three VSDs (I–III). The association of $\alpha_2\delta^{-1}$ with $\alpha_1C$ increases the coupling of VSDs I–III to the channel pore, allowing the CaV1.2 channel to operate in the range of physiological membrane potentials found in excitable cells.

The physical nature of $\alpha_1C/\alpha_2\delta^{-1}$ association

The association of $\alpha_2\delta^{-1}$ with $\alpha_1C$ resulted in a substantial change in the intrinsic voltage-sensing properties of VSDs I–III (Fig. 4 and Table 3). This effect suggests either a direct physical or long-range allosteric interac-

### Table 2. Fitting parameters for the Boltzmann functions fitting the fluorescence data from each VSD (Fig. 4)

| VSD | Parameter | No $\alpha_2\delta$ | With $\alpha_2\delta$–I |
|-----|-----------|---------------------|-------------------------|
| VSD I | $q$ (e$^0$) | $1.6 \pm 0.1$ (n = 5) | $2.8 \pm 0.1$ (n = 5) |
|      | $V_{1/2}$ (mV) | $36.5 \pm 3.1$ | $61.1 \pm 1.3$ |
| VSD II | $q$ (e$^0$) | $1.2 \pm 0.1$ (n = 6) | $2.7 \pm 0.2$ (n = 5) |
|      | $V_{1/2}$ (mV) | $-6.7 \pm 1.8$ | $-50.8 \pm 3.9$ |
| VSD III | $q$ (e$^0$) | $0.9 \pm 0.1$ (n = 6) | $1.5 \pm 0.09$ (n = 5) |
|      | $V_{1/2}$ (mV) | $0.9 \pm 4.1$ | $-22.9 \pm 1.7$ |
| VSD IV | $q$ (e$^0$) | $0.9 \pm 0.04$ (n = 7) | $1.1 \pm 0.1$ (n = 4) |
|      | $V_{1/2}$ (mV) | $-51.4 \pm 4$ | $-48.5 \pm 2.5$ |

### Figure 4. The $\alpha_2\delta^{-1}$ subunit facilitates the voltage-dependent activation of CaV1.2 VSDs I–III, whereas VSD IV is unperturbed. (A–D) Mean voltage dependence of VSD activation constructed from experiments as in Fig. 3. The $\alpha_2\delta^{-1}$ subunit facilitated the activation of VSDs I–III, as revealed by a more hyperpolarized voltage dependence of VSD activation, although to a different extent, whereas VSD IV was unaffected. Boltzmann fitting parameters are reported in Table 2.
showing that the $\alpha_2\delta$-1 subunit interacts with the extracellular loops of repeats I–III (Wu et al., 2015), suggesting a common $\alpha_1/\alpha_2\delta$ topology in CaV1.1 ($\alpha_{1\delta}$) and CaV1.2 ($\alpha_{1C}$) channels. However, contribution of long-range allosteric interactions cannot be ruled out.

$\alpha_2\delta$-1 association alters the intrinsic voltage-sensing properties of VSDs I–III

The hyperpolarizing shifts in the F(V) values of VSDs I–III (Fig. 4) indicates that the active state of these voltage-sensors is favored in the presence of $\alpha_2\delta$-1 subunits. In VSD-gated channels, the activation of charge-bearing S4 segments is facilitated by the formation of salt bridges between positively charged S4 residues and negatively charged residues in adjacent VSD helices (Papazian et al., 1995; Wu et al., 2010; DeCaen et al., 2011; Tuluc et al., 2016). Association of the $\alpha_2\delta$-1 subunit may facilitate the formation of such bonds by physically remodeling the spatial organization of the transmembrane helices of VSDs, altering their relative positions. Interestingly, we found that the VSDs’ sensitivity to changes in the membrane potential, i.e., the effective charge or slope of the F(V) curves ($q$), is almost equal among the four VSDs in the absence of the $\alpha_2\delta$-1 subunit ($q \approx 1 \, e^0$), whereas $q$ increases by approximately twofold for VSDs I–III in the presence of $\alpha_2\delta$-1. The effective charge $q$ of a voltage-sensing residue is given by the product $ze^0$, where $z$ is the valence number and $e^0$ is the electrical distance or fraction of membrane potential traversed by the residue. Because it is very unlikely that $\alpha_2\delta$-1 association adds voltage-sensing charges, the increased apparent charge observed for VSD I–III suggests that their charged S4 helices move across a greater electrical distance, through either a larger spatial translation (e.g., moving at a steeper angle) or more concentrated electric field lines. Indeed, in VSD-endowed proteins, the local electric field can be tremendously enhanced by the existence of aqueous crevices separated by hydrophobic gaskets comprised of aromatic residue side chains (Asamoah et al., 2003; Starace and Bezanilla, 2004; Ahern and Horn, 2005; Chanda et al., 2005; Long et al., 2007; Tao et al., 2010; Lacroix and Bezanilla, 2011).

The $\alpha_2\delta$-1 subunit enhances the coupling of VSDs I–III to the pore

Do the observed changes in VSD voltage-sensing properties account for the facilitation of CaV1.2 activation by $\alpha_2\delta$-1 subunit? To answer this question, we used our allosteric model for CaV1.2 channel activation, which predicts the time- and voltage-dependent properties of each VSD and the pore (Pantazis et al., 2014). This model successfully accounted for the effects of $\alpha_2\delta$-1 binding by both increasing the energetic contributions of VSDs I–III to pore opening and increasing the effective charges of VSDs I and II (Fig. 6 and Table 3). Specifically, in channels lacking the $\alpha_2\delta$-1 subunit, VSDs I–III...
make a weak contribution to channel opening ($W > -20$ meV, equivalent to 0.8 kT or an allosteric factor of 2.2).

The striking outcome of this study is that the energetic contribution of the activation of VSDs II and III to pore opening in channels lacking $\alpha_2\delta$ is greatly reduced. This is in contrast to channels containing $\alpha_2\delta^{-1}$, where VSDs II and III contribute two to three times as much energy toward channel opening ($\sim -95$ meV, $\sim 3.7$ kT) or, in an alternative interpretation, their activation is obligatory for pore opening (Pantazis et al., 2014). The diminished VSD–pore conformational coupling in channels lacking $\alpha_2\delta$ is also supported by the good approximation of the $G(V)$ by a Boltzmann distribution (Fig. 2 C), which implies a single voltage-dependent opening transition without significant input from VSDs. Our previous work on $\alpha_2\delta$-containing CaV1.2 channels revealed a surprising disparity in the VSD voltage dependencies, greater than that observed in related pseudotetrameric NaV channels: VSD activations ($V_{\text{half}}$ values) spanned 50 mV. The functional heterogeneity of the four VSDs was attributed to (a) the different amino acid composition of each VSD and (b) the structural asymmetry of the channel complex arising from its 1:1:1 $\alpha_1/\beta/\alpha_2\delta$ subunit stoichiometry. Interestingly, in this work, we found that increasing CaV1.2 structural symmetry (by excluding $\alpha_2\delta^{-1}$ subunits) in fact made the VSD voltage dependencies even more disparate, spanning $\sim 90$ mV (Fig. 5). Our current model interpreted this finding as a result of direct modification of VSD voltage-sensing properties and reduced coupling of VSDs I–III to the pore by $\alpha_2\delta^{-1}$. Another possible explanation is that $\alpha_2\delta^{-1}$ acts as an allosteric center (in addition to the pore), increasing the coupling between voltage sensors. However, this possibility implies that $\alpha_2\delta^{-1}$ also undergoes conformational changes,
for which there is yet no experimental evidence. Perhaps future studies could explore the possibility.

Conclusions

In summary, we have used VCF to optically track the molecular rearrangements of the individual VSDs of a human CaV1.2 channel in the presence or absence of α2δ-1. VCF is now a well-established method to assess voltage-dependent conformational changes, allowing us to track the movement of individual VSDs and to resolve slow conformational changes (as those observed in VSD I) that are extremely difficult to capture by gating current measurements. In this work, we have not systematically recorded gating currents, as they could not reveal the individual contributions of each VSD to CaV1.2 activation. Perhaps the most important advantage of VCF is that all recordings could be performed in conducting channels, whereby ionic currents and VSD movements were sampled simultaneously, without the use of pore-blockers. We found that the α2δ-1 auxiliary subunit significantly alters the voltage dependence of VSDs I–III, facilitating their activation, but not that of VSD IV. A 32-state allosteric model, consistent with the CaV1.2 molecular architecture, predicts the major kinetic and steady-state features of the experimental data, revealing that the association of α2δ-1 with α1C (in the presence of β3) specifically increased the coupling energy of VSDs I–III to the pore, as well as effective gating charge in segments I and II. Without the enhanced gating properties brought about by α2δ-1 association, CaV1.2 channels could not operate at physiological membrane potentials.

Table 3. Fitting parameters for the model predictions in Fig. 6

| Parameter          | No α2δ | With α2δ-1 a |
|--------------------|--------|--------------|
| Pore               |        |              |
| qL (e0)            | 0.99   | 0.76         |
| VL (mV)            | 103    | 140          |
| xL                 | 1.0    | 0.49         |
| νL (s⁻¹)           | 1,356  | 670          |
| VSD I              |        |              |
| qL (e0)            | 1.4    | 2.0          |
| VL (mV)            | 37     | 8.5          |
| xL                 | 0.72   | 0.89         |
| νL (s⁻¹)           | 17     | 110          |
| VSD II             |        |              |
| qL (e0)            | 1.2    | 2.5          |
| VL (mV)            | -6.9   | -27          |
| xL                 | 0.78   | 0.99         |
| νL (s⁻¹)           | 35     | 44           |
| VSD III            |        |              |
| qL (e0)            | 0.83   | 1.0          |
| VL (mV)            | 3.7    | -11          |
| xL                 | 1.0    | 0.93         |
| νL (s⁻¹)           | 145    | 160          |
| VSD IV             |        |              |
| qL (e0)            | 0.92   | 1.1          |
| VL (mV)            | -54    | -52          |
| xL                 | 0.35   | 0.55         |
| νL (s⁻¹)           | 16     | 11           |
| Energetic interaction | W1 (meV) | -8.0 |-16 |
| W2 (meV)           | -16    | -50          |
| W3 (meV)           | -19    | -45          |
| W4 (meV)           | 4.1    | -0.87        |

aParameters in this column are from Pantazis et al., 2014.

Figure 7. The α2δ subunit covers ~3/4 of the CaV1.2 pore-forming subunit, excluding VSD IV. Side and top views of the CaV1.2 (α1C/α2δ/β) channel volume (pink and light blue) modified from cryo-EM data from Walsh et al. (2009a) are shown. The atomic structures of NaVAb (PDB accession no. 4EKW; Payandeh et al., 2012), with subunits arranged clockwise, and the atomic structure of AID-associated β3 subunit (PDB accession no. 1VYT; Chen et al., 2004) were manually positioned in the cryo-EM volume. Because VSD IV is not perturbed by α2δ-1, we propose that the resolved α1C volume not covered by α2 is occupied by VSD IV, whereas VSDs I–III are encompassed by the α2δ-1 subunit, which alters their biophysical properties and, in the case of VSDs II and III, enhances their coupling to the channel pore.

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SUPPLEMENTAL MATERIAL

Savalli et al., http://www.jgp.org/cgi/content/full/jgp.201611586/DC1

Figure S1. Representative Ba$^{2+}$ current traces from Xenopus oocytes expressing CaV1.2 channel complexes formed by $\alpha_{1C} + \beta_3$ subunits. Because in the absence of $\alpha_2\delta-1$ subunits the CaV1.2 channel is slow to activate, incrementally longer pulses were used to achieve quasi–steady state. The tail currents at −40 mV were used to construct the G(V) curves in Fig. 2 (black squares). The voltage protocol is reported above the current traces.

Figure S2. Histograms of posterior distributions of 14 parameters obtained from a 100,000-trial MCMC run. The first 6,000 values corresponding to the burn-in period that precedes the stationary phase were excluded. The orange vertical lines correspond to parameter values of the best fit that was used to generate the model predictions for fluorescence and ionic current (Fig. 6, black continuous lines). Horizontal blue bars show the 95% credible intervals that include 95% of the acceptable values visited during our statistical analysis. The bin size was scaled to maintain the most frequent value at $\sim$15,000 to highlight the overall similarity in shape. Histograms for the remaining 10 parameters ($x_i$ and $\nu_i$) are summarized in Fig. S3.
Figure S3. Histograms of posterior distributions of parameters $x_i$ and $\nu_i (i = L, 1, 2, 3, 4)$. As in Fig. S2, the orange vertical line corresponds to parameter values of the best fit, and the horizontal blue bars show the 95% credible interval. The bin size was scaled to maintain the most frequent value at $\sim 25,000$. To constrain the possible solutions, we added several constrains to the error function using the following equation:

$$C_{str}(i) = a_i \left[ \exp \left( \frac{|f(x) - f(c)|}{b_i} \right)^2 - 1 \right],$$

where $a_i$ is a scaling factor, $b_i$ sets the limits of the constrains, and $c_i$ corresponds to the optimum value. We constrained the time constants for the onset of ionic currents and fluorescence traces at 20 mV to the mean time constant obtained from single exponential fit ($\tau_{avg}$ in Table 1). We also penalized the time constant of the ionic tail current with a target value of 0.9 ms. The off relaxation of VSD II and VSD III fluorescence traces at 40 mV were also limited to $8.5 \pm 2$ and $8 \pm 2$ ms, which correspond to the time constants of the single exponential that best described the decay of the traces. Parameter sets that yielded ionic currents displaying inactivation (negative ratio between peak current and the amplitude at the end of the pulse) or with slope of $\ln(GV) - 160$ mV diverging from $q_i$ were also penalized.