Symposium

Illuminating the Diversity of Electrical Synapse Form and Function

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While the nervous system is organized by two main types of connections between neurons, electrical and chemical synapses, most studies focus upon the contribution of the molecularly complex and functionally diverse chemical synapse. Electrical synapses, in contrast, are perceived as simple, neuronal gap junction channels between contacts, often thought to be replaced in early development by more sophisticated chemical synapses. However, recent work and new tools push for the redefining of electrical synapse molecular architecture and function to allow for an updated interpretation of their ongoing contributions to neural circuit formation and function. Here, we describe fresh insight into neuronal gap junction structure, function, and molecular composition within invertebrate (nematodes, drosophila) and vertebrate (zebrafish, rabbit, rodent) model organisms across a variety of time periods and brain regions. Through molecular, genetic, connectomic, and functional explorations, our results argue for a new appreciation of electrical synapse heterogeneity. Based on these and other points of emerging new evidence, we argue for increased consideration of electrical synapses as complex and diverse structures which interact with chemical synapses to drive circuit formation and function.

Introduction

Neurons use two main mechanisms to pass information from one cell to another: chemical synapses, which feature unidirectional information flow through neurotransmitter release and reception across a synaptic cleft, and electrical synapses, which connect the cytoplasm of cells together via neuronal gap junction channels to allow for the usually bidirectional flow of ions and other small metabolites (Fig. 1). While chemical synapses are established as complex molecular structures with hundreds to thousands of proteins orchestrating their varied roles and diverse presentations (Collins et al., 2006; Bayés and Grant, 2009; Ryan and Grant, 2009; Dieterich and Kreutz, 2016), electrical synapses have traditionally been viewed as simple structures composed solely of the gap junction channel proteins, called Connexins in chordates or the evolutionarily distinct Innexins in prechordates (Goodenough and Paul, 2009). Non-neuronal gap junction channels, which use different Connexin/Innexin family members than their electrical synapse counterparts, are ubiquitous throughout tissues, and their trafficking, assembly, and regulation are well described, while the specific molecular mechanisms directing neuronal gap junction formation and function are less well appreciated.

The shape of gap junction contacts can vary from thin, lacey strings to thick, wide bands, but all electrical synapses contain groups of tens to thousands of channels that cluster into gap junction plaques (Nagy et al., 2018). Within these plaques, a single hexamer of Connexin/Innexin proteins, also known as a hemichannel, is contributed by one cell and docks with a hemichannel provided by the partner cell to form the intercellular gap junction channel connecting the cytoplasms of the two cells. Electrical synapses are not limited to somatic contacts but instead can connect all types of neuronal compartments (e.g., dendrodendritic, axoaxonic, dendrosomatic, etc.; Kosaka and Hama, 1985; Hamzei-Sichani et al., 2007; Nagy et al., 2018), and they

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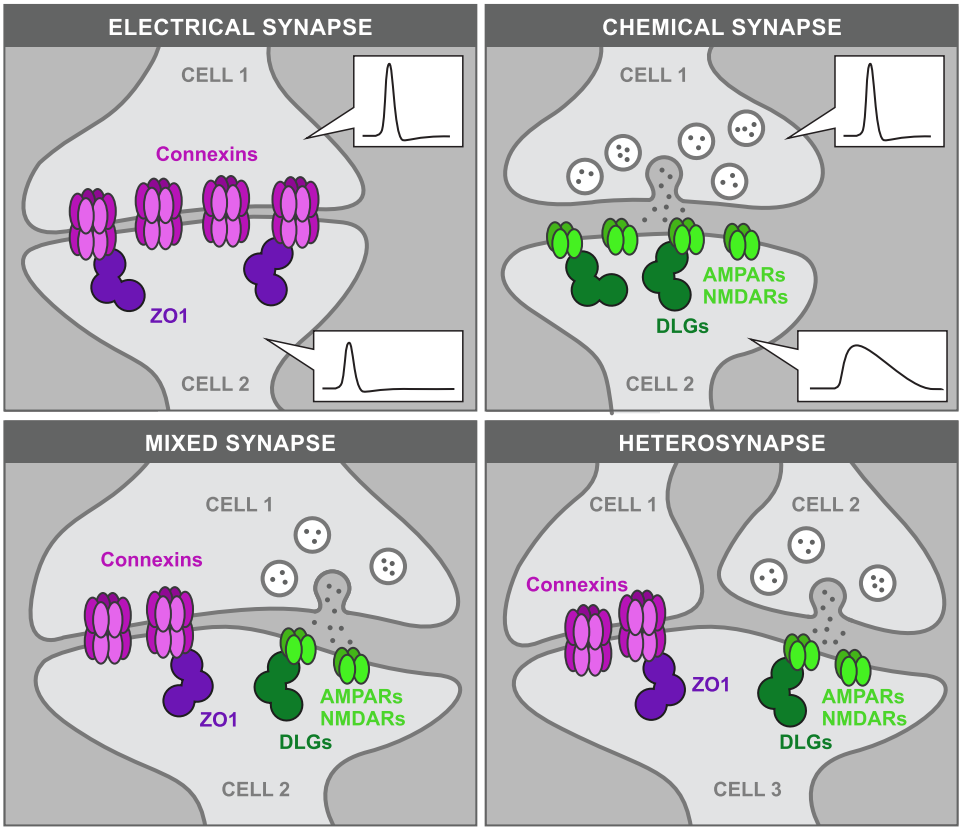


Figure 1. Electrical and chemical synapses are found in fully distinct locations as well as in mixed synaptic connections between two cells and heterosynaptic connections linking three cells. Both chemical and electrical synapses mediate transmission of the presynaptic current of Cell 1 to Cell 2 (callout boxes), albeit through distinct mechanisms that endow the resulting potentials with unique features. Gap junction Connexins in magenta, necessary electrical synapse scaffold ZO1 in purple, chemical synapse AMPA/NMDA neurotransmitter receptors in light green, assorted chemical synapse scaffolds DLGs in dark green.

are found across the brain from sensory to central to motor circuits in both developing and mature brains (Galarreta and Hestrin, 2001; Connors and Long, 2004; Nagy et al., 2018). The few published studies that have quantified electrical synapse number compared with that of chemical synapses in adult animals (*Caenorhabditis elegans*, rabbit retina, *Ciona*) find electrical synapses in some cell classes account for up to ~20–27% of visible connections (White et al., 1986; Anderson et al., 2011; Jarrell et al., 2012; Ryan et al., 2016; Cook et al., 2019). Although neuronal Connexin expression decreases with development, it does not disappear, and evidence now strongly supports the view that electrical synapses coordinate with chemical synapses across developing and mature neural circuits to dynamically shape brain function (Belluardo et al., 2000; Söhl et al., 2005, 2010; Wang and Belousov, 2011; Pereda, 2014; Song et al., 2016).

Electrical synapses are perhaps most known for their fast speed of information transfer and their well-defined roles in synchronizing neural networks to drive the earliest animal behaviors (Rekling et al., 2000; Saint-Amant and Drapeau, 2000; Marin-Burgin et al., 2006; Su et al., 2017), as well as mature circuit activity (Connors, 2017; Nagy et al., 2018). However, electrical synapses can also excite, inhibit, desynchronize, and coordinate with chemical synapses to drive diverse types of neural circuit processing (Connors, 2017; Vaughn and Haas, 2022). Disorders affecting various tissue types have been attributed to the loss of gap junctions (Laird, 2006). In the nervous system, studies of Cx36 knock-out mice show brainwide defects, such as the desynchronization of cortical interneurons (Zolnik and Connors, 2016). With knowledge growing as to electrical synapse molecular composition and diverse functionality, electrical synapses are becoming increasingly linked to neurological disorders, and their disruption is currently suggested to contribute to disorders such as epilepsy, myopia, and autism (Welsh et al., 2005; Cunningham et al., 2012; Kunceviciene et al., 2018). In this review, which is based on a mini-symposium presented at the 2026 meeting of the Society for Neuroscience, we examine recent evidence from both invertebrate and vertebrate systems that expand our current appreciation of the diversity of electrical synapse formation and function.

Electrical Synapses as Plastic, Asymmetric, and Complex Compartmentalized Structures

Evidence suggests that electrical synapses have a complex molecular organization with distinct pre- and postsynaptic sites that, in addition to gap junction proteins, require the contributions of additional cell–cell structures. This had been shown, for example, in the Mauthner cells, a pair of colossal reticulospinal neurons that, as a result of the spinal connectivity of their axons, mediate auditory-evoked escape responses in fish (Pereda and Faber, 2011). Auditory information is provided to the Mauthner cells by a group of large single contacts known as "Large Myelinated Club endings" (CEs; Bartelmez and Hoerr, 1933), where gap junctions and specializations for glutamatergic transmission coexist (Fig. 1). The large size of the Mauthner cell and CEs makes them ideal for structural-functional correlations, efforts that lead to the identification of the gap junction as the structural bases for electrical transmission (Robertson et al., 1963;

Furshpan, 1964). Investigations on these terminals exposed the ability of electrical transmission to undergo activity-dependent potentiation of electrical transmission, indicating that electrical synapses are capable of more sophisticated properties than generally recognized. The induction of these potentiations of electrical transmission occurs via interactions with its colocalized glutamatergic counterpart (Yang et al., 1990; Pereda and Faber, 1996; Pereda et al., 1998; Cachope et al., 2007).

More recent work has dealt with investigating the molecular organization of these synapses that is responsible for these plastic properties in larval zebrafish. Gap junctions at these terminals are asymmetrically organized: the intercellular channel is formed by a hemichannel made of Cx35, while the postsynaptic (Mauthner cell) hemichannel is formed by Cx34, both fish homologs of Cx36 the widespread neuronal Connexin present at most of electrical synapses in the mammalian brain (Rash et al., 2013; Miller et al., 2017; Lasseigne et al., 2021). We also found that electrical transmission at these gap junctions is supported by a molecular scaffold with properties that are similar to the postsynaptic density of glutamatergic synapses. One molecular component of this gap junction scaffold is ZO1, a protein that has a specialized function at neuronal gap junctions, and MAGUK family members like PSD95 and Sap102. The presence and interaction of ZO1 is hierarchically required for the surface expression of Connexins at CEs (Flores et al., 2008; Lasseigne et al., 2021). Further revealing the asymmetric organization of the gap junctions at these terminals, combined biochemical, genetic, and nanoscale analysis with expansion microscopy indicates that ZO1 is restricted to the scaffold of the postsynaptic hemiplaque (Lasseigne et al., 2021; Cárdenas-García et al., 2024). Expansion microscopy also revealed that electrical transmission is mediated by ∼35 gap junctions that occupy ∼80% of the contact's surface (Cárdenas-García et al., 2024). These junctions are interleaved with adherens junctions, cell–cell contacts that contain N-cadherins and beta-catenin, among other proteins (Cárdenas-García et al., 2024). Adherens junctions were proposed to play an important role in the delivery of channels to gap junctions (Shaw et al., 2007). That is, vesicles carrying undocked hemichannels are trafficked and delivered by microtubules to adherens junctions, and from there to the periphery of gap junctions where they dock with hemichannels in the apposed membrane forming new intercellular channels that will become part of the plaque. The turnover of gap junction channels is an intense process with a half-life of 2–5 h. We found evidence that this turnover occurs at gap junctions in CEs, in vivo, and that is required to maintain the strength of electrical transmission (Flores et al., 2012). Regulation of Connexin trafficking could therefore underlie changes in the strength of electrical transmission at these terminals. Thus, the CE electrical synapse shows a complex molecular organization with an asymmetric gap junction structure and a hierarchical role of its scaffold, as well the presence of additional structures that are likely to play an important functional role in the regulation of gap junction channels.

An Expanded Understanding of Electrical Synapse Molecular Composition Which Overlaps with Chemical Synapses

Electron microscopy data shows an electron dense area surrounding neuronal gap junction channels reminiscent of the well-recognized postsynaptic density of chemical synapses (Brightman and Reese, 1969; Kimmel et al., 1981). Coupled together with the ability of electrical synapses to undergo activity-dependent plasticity among other molecularly complex actions (Miller and Pereda, 2017; Alcamí and Pereda, 2019), these features hint at the necessity for electrical synapses to contain a specialized molecular architecture beyond Connexins/Innexins. To identify additional molecular players, we used Turbo-ID fused to a neural Connexin, Cx34, to biotinylate proteins in proximity to electrical synapses in larval and adult zebrafish (Michel et al., 2024). We identified Connexin-associated proteins via mass spectrometry revealing ∼150 proteins including those previously known to localize with Connexins at electrical synapses such as ZO1 and Neurobeachin (Miller et al., 2015; Marsh et al., 2017; Lasseigne et al., 2021; Martin et al., 2023). The remaining proteins identified include cell adhesion and trafficking proteins, cytoplasmic scaffolds and regulators, and canonical glutamatergic synapse proteins. While the significant overlap with chemical synapse molecules is perhaps surprising, electrical synapses can be found in close proximity to chemical synapses, such as at mixed electrical/glutamatergic contacts, and molecules have been found that are necessary to regulate the formation of both structures. For example, Neurobeachin interacts with ZO1 at electrical synapses and SAP102 at chemical synapses to regulate the formation of both synapse types (Lauks et al., 2012; Miller et al., 2015; Lasseigne et al., 2021; Martin et al., 2023).

To validate these Turbo-ID-identified candidates, we used the Mauthner circuit of larval zebrafish, which allows for access to electrical and chemical synapses at known contact points between known partner cells in early neural development. In particular, we examined the previously mentioned Club Ending (CE) mixed electrical/glutamatergic synapses which connect auditory afferents with Mauthner dendrites, and the M/CoLo electrical synapses connecting the Mauthner (M) axon to commissural local (CoLo) interneurons. We leveraged CRISPR methods to investigate (1) cell biological localization and (2) requirement in synapse formation, and in our unpublished data we find canonical chemical synapse proteins which both localize at the electrical synapse and regulate their formation (Michel et al., 2024). Further, we observe that candidate proteins can vary both in their localization patterns and their requirement for synapse formation at different Mauthner electrical synapse types, arguing for previously unknown heterogeneity between synapses (Michel et al., 2024). Overall, these results uncover a new understanding of electrical synapses as a complex molecular compartment and emphasize the need to investigate the overlapping synaptic players between electrical and chemical synapses to reveal the shared or differing molecular mechanisms a neuron uses to regulate the construction of these two synapse types.

The Heterochannel Nature of Electrical Synapses, an Additional Layer of Complexity

In addition to molecular diversity surrounding the gap junction channel, the channel proteins themselves (Connexins/Innexins) can be found in diverse arrangements to orchestrate specific synapse functions. Individual neurons in both vertebrates and invertebrates often express multiple Connexins/Innexins (Takeuchi et al., 2017; Bhattacharya et al., 2019; Taylor et al., 2021). In the model organism, nematode *C. elegans*, with a fully mapped nervous system connectome, 14 Innexin genes are expressed in a complex, combinatorial pattern that can also change in response to altered environmental conditions (Bhattacharya et al., 2019). The availability of a multitude of Connexins/Innexins allows gap junctions to form with diverse molecular

compositions, which may have distinct biophysical properties, including directionality of conductance, selective permeability of ions, second messengers, and metabolites (Veenstra, 1996; Weber et al., 2004; Söhl et al., 2005; Meşe et al., 2007; Miller and Pereda, 2017; Shui et al., 2020). Despite this functional diversity, the molecular mechanisms by which neurons utilize coexpressed Connexins/Innexins to establish selective connections with distinct synaptic partners have remained largely unknown. Insights into this organizational principle have emerged from studies of the Mauthner cell circuit in zebrafish, which mediates fast-acting escape responses. Of the two coexpressed Connexins in this circuit, Cx35 is used exclusively on the presynaptic side, while Cx34 is used on the postsynaptic side, forming heterotypic, rectifying electrical synapses (Miller et al., 2017). However, all gap junction channels present within a single electrical synaptic patch were thought to be molecularly identical. Contrary to this prevailing "one synapse, one channel type" paradigm, our recent work revealed that electrical synapses in the *C. elegans* nervous system can be formed by clustering molecularly distinct gap junction channel types (Vats et al., 2026). These channel types are composed of different Innexin proteins, thereby assigning a distinct combinatorial gap junction channel code to each synapse. Specifically, our work highlighted that in the posterior mechanosensory circuit of *C. elegans*, individual electrical synapses are formed by clustering homotypic gap junction channels composed of Innexin, INX-1, and reciprocally arranged heterotypic channels formed by two distinct Innexins, UNC-7 and UNC-9 (Vats et al., 2026). Within these "heterochannel electrical synapses," distinct channel types cooperate to regulate synaptic functions (Vats et al., 2026). Our unpublished, preliminary data suggest that this complex, higher-order heterochannel organizing principle may well be conserved in Purkinje cell synapses in the zebrafish nervous system. Furthermore, individual neurons often express multiple Innexin or Connexin splice isoforms, which may have distinct biophysical properties (Bhattacharya et al., 2019; Namikawa et al., 2019; Taylor et al., 2021). It can be hypothesized that expression of these splice isoforms further diversifies the combinatorial gap junction codes of each synapse, thereby contributing to functional diversity. Our recent work further demonstrated that the combinatorial gap junction code of individual synapses can independently change under altered environmental conditions that induce *C. elegans* larvae to molt into the dauer diapause stage (Vats et al., 2026), thereby introducing an added layer of synapse-level molecular and functional plasticity to electrical synapses.

Diversity in the Regulatory Mechanisms Allows the Formation or Modification of Heterochannel Code

Selective localization of distinct gap junction channels to heterochannel electrical synapses, as well as their modification during the dauer stage, indicates that different channel types may be regulated by channel-specific synaptic localization and turnover mechanisms. In the Mauthner cell circuit in zebrafish, the autism-associated protein Neurobeachin and the tight junction protein ZO1 function specifically on the postsynaptic side to regulate the localization of Cx34. (Martin et al., 2023). Within the motor circuit of *C. elegans*, localization of UNC-9 and thus the synaptic assembly of UNC-9-UNC-7 heterotypic channels are determined by cAMP levels, presumably through alterations in microtubule organization (Palumbos et al., 2021). The C-terminal amino acid sequence is shown to regulate the trafficking of Cx35 in fish (Flores et al., 2012). Our recent work revealed that within individual heterochannel electrical synapses of *C. elegans*, the synaptic trafficking of molecularly distinct gap junction channel types is independently regulated by specific and conserved kinesin motor proteins (Vats et al., 2026). In the posterior mechanosensory circuit, INX-1 channels are trafficked by two kinesins from two distinct conserved families: KLP-4/KIF13 and KIF14 (a kinesin-3 family protein), as well as KLP-3/KIFC2 and KIFC3 (a kinesin-14 family member). However, coexisting UNC-9-UNC-7 channels are trafficked by an independent, yet unidentified mechanism (Vats et al., 2026). Similarly, distinct pathways also regulate the turnover of these channels within the same heterochannel synapses. While the conserved retrograde kinesin KLP-15/KAAR-3 restricts the INX-1 channel localization, a pathway involving UNC-44/Ankyrin, UNC-33/CRMP, and VAB-8/Atypical kinesin regulates UNC-7-UNC-9 channel turnover (Meng et al., 2016; Vats et al., 2026). Yet the molecular and structural basis of this Innexin/Connexin selectivity remains to be fully understood. Notably, downregulation of KLP-4 expression in mechanosensory neurons in the dauer stage coincides with changes in the heterochannel gap junction code of specific synapses within this circuit, further emphasizing the need to understand the regulatory mechanisms of gap junction trafficking. Additionally, as chemical and electrical synaptic components can coexist in close proximity within mixed synapses (Cárdenas-García et al., 2024), it is essential to understand the cross talk between the cellular mechanisms governing these two types of synapses.

Mapping Cross-Modality Interaction in the Mammalian Retina: Mixed (Electrical + Chemical) Synapses and Other Emergent Circuit Topologies

Electrical synapses provide a powerful and dynamic alternative mode of signal transmission to chemical synapses within the nervous system. The specialized communication features of these two modalities make each well adapted for different aspects of signal processing. However, as mentioned above, chemical and electrical synapses often coexist within neural networks, and even within the same neuron, and the combination of these modalities can create complex, powerful signal integration and emergent functions within these cells and networks (Pereda, 2014; Vaughn and Haas, 2022). Heterosynaptic interactions between electrical and chemical synapses also provide additional mechanisms of synaptic plasticity (Cachope and Pereda, 2021; Fig. 1).

The ability to investigate cross-modality interactions has largely been thwarted by the ability to identify potential instances and define the networks in which they are embedded. The modern era of connectomics, ushered in by advances in computational infrastructure, now enable comprehensive mapping of network features in database-backed, large volumes of high-resolution imaged neural tissues (Sigulinsky et al., 2024). These datasets allow the identification and precise mapping of synapses, their spatial relationships, and classification of partner cells. In recent work, Retinal Connectome (RC1) was mined to investigate the potential for cross-modality interaction. RC1 is a high-resolution serial-section transmission electron microscopy (TEM) volume of adult, female rabbit retinal tissue (Anderson et al., 2011). Importantly, its native resolution of 2 nm/pixel reveals the morphology of gap junctions, which are the anatomical basis of electrical synapses, and archivable grids allow reimaging at higher magnification with goniometric tilt for validation when necessary. RC1 contains over 4,800 annotated candidate and confirmed gap junctions formed by a variety of retinal neurons.

In the mammalian retina, gap junctions are abundant and critical for signal transfer and shaping visual processing in diverse retinal networks. Cone bipolar cells are essential retinal interneurons mediating the transfer and initial processing of signals from light-sensing photoreceptors to brain-projecting ganglion cells. The seven classes of ON cone bipolar cells all form gap junctions in their axonal arbors and exhibit robust specificity in their partnerships, sizing, and spatial topologies (Sigulinsky et al., 2020). These gap junctions are also interspersed among a plethora of chemical synapses mediating excitatory output and inhibitory input of these same cells. In fact, preliminary findings indicate that nearly all ON cone bipolar cell gap junctions exist in close proximity (<2 μm) to multiple chemical synapses. Some of these gap junctions and chemical synapses exist in close proximity between the same two cells, forming classical mixed synapses, as previously described at Mauthner Club Ending (CE) synapses in fish. Glutamatergic excitatory motifs of these mixed synapses are common, but only a subset of ON cone bipolar cell classes participate and each exhibit class-specific partners. Interestingly, pure inhibitory motifs are absent. Rather, inhibitory chemical synapse input onto an ON cone bipolar cell arbor is only found with a gap junction between these same two cells when in combination with an excitatory chemical synapse, resulting in a rare, reciprocal-type of classical mixed synapse.

Another retinal cell, the transient ON direction-selective ganglion cell, also exhibits a high frequency of gap junctions interspersed among excitatory and inhibitory input within its dendritic arbor, including several instances of GABAergic inhibitory mixed synapses (Marc et al., 2018). Although additional two-cell mixed synapse motifs were absent, preliminary findings revealed this ganglion cell participates in highly stereotyped three-cell pairings involving gap junction coupling at the sites of excitatory input.

These two- and three-cell organizations of chemical and electrical synapses within the arbors of retinal cells likely facilitate cross-modality interactions with implications on signal integration, including the strength, timing, and plasticity of individual synapses. The mixed synapse motifs provide an additional potential mechanism for further diversifying ON cone bipolar output at individual synapses, which may underlie the formation of functionally distinct microcircuits or multiplexing across their arbors. The specificity of the observed motifs suggests they are essential components of the retinal network architecture. Furthermore, the organization of gap junctions in multiple distinct motifs and recent report of "silent but activatable" gap junctions (Ixmatlahua et al., 2020) in mixed synapse configurations underscores the importance of TEM-based ultrastructural connectomics to map gap junctions in neural networks and discover these important motifs to ultimately tie structure to function.

Connecting Electrical Synapse Formation and Function to Behavior in *Drosophila*

Both mammalian and invertebrate nervous systems contain densely packed neurons in stereotyped locations. While gap junctions are not formed promiscuously between random adjacent cells (Teubner et al., 2000; Rabinowitch et al., 2014), electrical synapses are well documented to be present within populations of a single neuron type, including the fly lamina, worm AVA interneurons, and mammalian inferior olive neurons (Bhattacharya et al., 2019; Turecek and Regehr, 2020; Ammer et al., 2022). Here we hypothesized that the formation of gap junction electrical synapses is required for the proper function of the population of individual neurons compromising a neuronal type. To investigate this, we used the well-characterized *Drosophila melanogaster* Moonwalker Descending Neuron (MDN) escape circuit, which commands backward walking (Bidaye et al., 2014). There are four MDNs which respond to environmental stimuli before initiating and maintaining backward walking (Sen et al., 2017, 2019; Feng et al., 2020; Lee and Doe, 2021; Israel et al., 2022; Cheong et al., 2025).

We assayed the widely expressed gap junction component Innexin 8 (Inx8; FlyBase: ShakB) (Hasegawa and Turnbull, 2014) and confirmed that Inx8 is detected in adult MDNs, specifically between MDN cell bodies and between primary neurites, with little at axon terminals and on dendrites. In control animals, the four MDN cell bodies are tightly clustered together. However, when Inx8 was knocked down in MDN, the cell bodies were no longer present in one tight cluster, instead broken into two or three aggregates. Additionally, when Inx8 was knocked down and MDNs were optogenetically activated, the flies did not walk backward (Fig. 2). This result begins to indicate that the location of the MDN cell bodies—and therefore the formation of the gap junctions between them—is required for the MDN

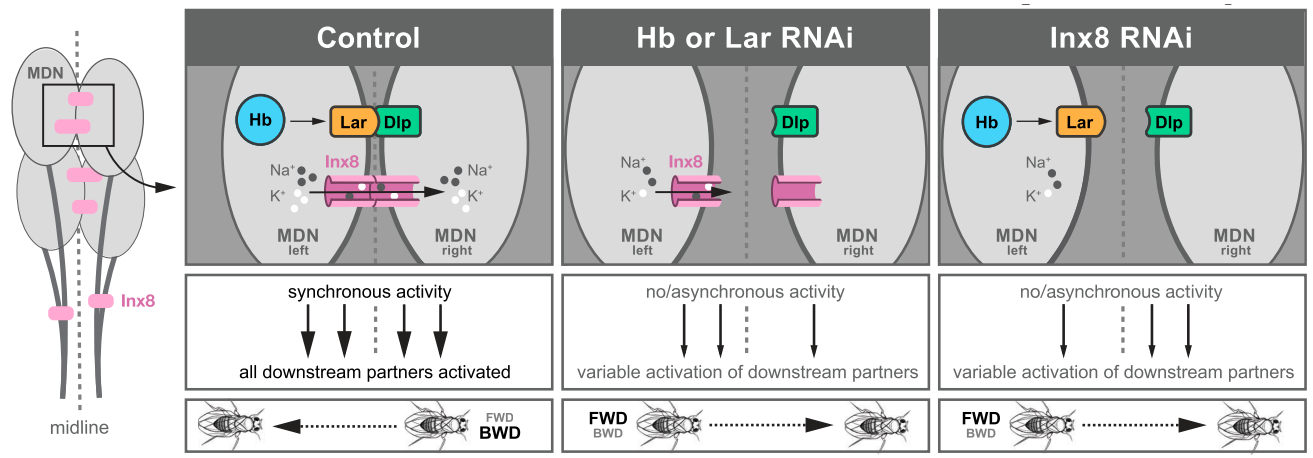


Figure 2. Gap junction (Inx8) and cell surface molecule (Lar and Dlp) expression in the *Drosophila* Moonwalker Descending Neurons (MDNs). In control animals, the transcription factor Hunchback activates the expression of Lar; Inx8 is required for the synchronous firing of all MDNs, which allow the fly to walk backward. This is disrupted when Hb, Lar, or Inx8 are knocked down, leading to a lower resting membrane potential, asynchronous activity, and a lack of backward walking.

circuit to function. To follow this up, we screened for additional genes that would disrupt MDN cell body location.

We found a single transcription factor, Hunchback, is required in the four MDNs for expression of the Lar–Dlp cell adhesion complex in the MDN cell bodies. Interestingly, Hunchback, Lar, and Dlp are required for the location of the MDN cell bodies—when any of these proteins were knocked down in MDN, the cell bodies were no longer present in one tight cluster. Furthermore, when Hunchback, Lar, or Dlp were knocked down and the adult MDNs were optogenetically activated, the flies did not walk backward. We also assayed chemical synapse number, axon and dendrite morphology, and neurotransmitter expression—knocking down Hunchback in MDNs did not significantly alter any of these neuronal properties. Strikingly, the simplest interpretation is that the backward walking circuit function relies on the four MDN cell bodies clustering together.

In follow-up studies, we found that Inx8 gap junction expression is independent of Hunchback, but both Hunchback and Lar are required for the size of the Inx8 plaques on MDN cell bodies. We also found that Inx8 puncta are associated with both Lar and Dlp on MDN cell body membranes. We conclude that Inx8, Lar, and Dlp are interacting on MDN cell bodies to promote MDN clustering, which is required for backward walking. These data also support the hypothesis that the transmembrane protein Lar is required for Inx8 organization at MDN-MDN contact sites.

We hypothesized that cell body clustering maintains MDN electrophysiological properties necessary for backward walking. Utilizing the genetically encoded voltage indicator ASAP5 (Hao et al., 2024), we assayed resting membrane potentials and synchronous activity in MDNs. We calculated the likelihood that two MDNs would have synchronous activity, finding that control MDNs were significantly more likely to have a synchronous voltage response compared with Hb and Inx8 knockdown animals. Additionally, we calculated the baseline fluorescence when the cells were inactive, analogous to their resting membrane potential. We found that Hb and Inx8 knockdown animals had a significantly lower baseline fluorescence compared with control, resulting in an approximate −20 mV difference. We believe that the decoupling of the MDN cell bodies, resulting in the lower resting membrane potential, make it less likely that the neurons will initiate an action potential. If they do have an action potential, it is unlikely that all four MDNs fire, resulting in uncoordinated activation of the downstream partners. Taken together, our findings highlight the importance of electrical synapse formation for neuronal circuit function.

Our data demonstrate an absolute requirement of electrical coupling within a neural network for behavior: without electrical coupling of the four MDNs, the fly is incapable of initiating backward walking. Given that fly Innexin gap junctions can function in vertebrate cells, and vertebrate Connexin gap junctions can function in invertebrates (Starich et al., 2009; Meng and Yan, 2020), these results are likely translatable to other systems.

Accessing the Functional Diversity of Electrical Synapses and Networks across Rodent Brain Regions: Photomapping Electrically Coupled Networks of Thalamus and Cortex

Our understanding of the roles electrical synapses and coupled networks play in the central nervous system is acutely limited by the tools currently available. The gold standard for measuring electrical synapses is through paired recordings, in which current is injected into one neuron and a hyperpolarizing deflection can be measured in a coupled neuron, and the ratio of those deflections indicates synapse strength (Furshpan and Potter, 1959; Loewenstein and Kanno, 1964; Bennett, 1977). While this method is the most direct, it is time-intensive, low-throughput, and limited to single synapses in vitro; measuring single synapses one by one, we risk missing the forest formed by the trees. Here we describe a new approach to electrical synapses by measuring the changes in latency of rheobase-driven spiking from one patched neuron driven by individual focal photostimulations of nearby neurons expressing the soma-targeted opsin st-ChroME (Vaughn et al., 2026). This method reduces the requirement for recording an electrical synapse to a single patched neuron and simultaneously widely expands the number of potential synapses that can be tested in one experiment. Further, this method allows us to map coupled networks.

We applied this new technique to neurons of the thalamic reticular nucleus, which was recently shown to be composed of distinct neuron subtypes—parvalbumin-expressing and somatostatin-expressing inhibitory neurons (Perez et al., 2017; Li et al., 2020; Martinez-Garcia et al., 2020). We earlier described in paired recordings that the two types appear to cross-couple (Vaughn et al., 2024). Because the two TRN neuron types appear to distinctly interact with either primary or higher-order thalamocortical projection neurons (Perez et al., 2017; Martinez-Garcia et al., 2020) but without reciprocal GABAergic connectivity in TRN (Hou et al., 2016), we speculate that electrical synapses between TRN neuron subtypes form the only intrathalamic link between distinct thalamocortical relay channels.

First, we validated a strong correspondence between the two methods for measuring coupling, using both traditional shared hyperpolarizations and rheobase–latency shifts during photoexcitation of coupled neurons in a set of paired recordings. We showed that latency changes are almost entirely abolished by Cx36 knockdown, compared with those measured in a Cx29 knockdown. Using photostimulation of coupled neighbors, we showed that electrical synapses couple neurons both within and between neuron subtypes in the TRN. We confirmed earlier findings that electrical synapse strength diminishes with intersomatic distances. We showed that, in contrast to previous work, electrical synapses link neurons across intersomatic distances exceeding 100 microns. Our results also indicated that a single TRN neuron is connected to only 1 to 4 other TRN neurons. From these results, we conclude that electrical coupling within TRN networks offers an intrathalamic bridge between parallel information relays.

We also applied this new technique to cortical inhibitory networks. In cortex, our results showed that electrical synapses also span hundreds of microns of intersomatic distance. In contrast to TRN, somatostatin-positive cortical neurons couple ∼50% more strongly in cortex than in TRN, while parvalbumin-positive neurons couple much less frequently than in TRN. Together, our results suggest that the frequency and strength of electrically coupled networks are tailored to each mature brain area in which they appear. In principle, photomapping of coupled networks can be easily applied anywhere in the brain and used to understand the unique configurations and roles of electrical synapses and networks across the brain.

Conclusion

To conclude, the field of electrical synapse biology is rapidly expanding. The longstanding molecular distinctions between electrical and chemical synapses are beginning to blur, while the functional contributions of electrical synapses to neural

circuits are becoming clearer. Overall, throughout our work we observe an expansion of our appreciation of the electrical synapse heterogeneity required to facilitate the different roles electrical synapses fulfill in development and adulthood. Continued investigations into this heterogeneity and the contributions of electrical synapses to neural circuit form and function will help us better understand how neural circuits are constructed and offer insight toward the etiology of neurological disorders.

References

Alcamí P, Pereda AE (2019) Beyond plasticity: the dynamic impact of electrical synapses on neural circuits. Nat Rev Neurosci 20:253–271.

Ammer G, Vieira RM, Fendl S, Borst A (2022) Anatomical distribution and functional roles of electrical synapses in *Drosophila*. Curr Biol 32:2022–2036.e4.

Anderson JR, et al. (2011) Exploring the retinal connectome. Mol Vis 17:355–379.

Bartelmez GW, Hoerr NL (1933) The vestibular club endings in ameiurus. Further evidence on the morphology of the synapse. J Comp Neurol 57:401–428.

Bayés A, Grant SGN (2009) Neuroproteomics: understanding the molecular organization and complexity of the brain. Nat Rev Neurosci 10:635–646.

Belluardo N, Mudò G, Trovato-Salinaro A, Le Gurun S, Charollais A, Serre-Beinier V, Amato G, Haefliger J-A, Meda P, Condorelli DF (2000) Expression of Connexin36 in the adult and developing rat brain. Brain Res 865:121–138.

Bennett MVL (1977) Electrical transmission: a functional analysis and comparison to chemical transmission. Compr Physiol 1977:357–416.

Bhattacharya A, Aghayeva U, Berghoff EG, Hobert O (2019) Plasticity of the electrical connectome of *C. elegans*. Cell 176:1174–1189.e16.

Bidaye SS, Machacek C, Wu Y, Dickson BJ (2014) Neuronal control of Drosophila walking direction. Science 344:97–101.

Brightman MW, Reese TS (1969) Junctions between intimately apposed cell membranes in the vertebrate brain. J Cell Biol 40:648–677.

Cachope R, Mackie K, Triller A, O'Brien J, Pereda AE (2007) Potentiation of electrical and chemical synaptic transmission mediated by endocannabinoids. Neuron 56:1034–1047.

Cachope R, Pereda AE (2021) Regulatory roles of metabotropic glutamate receptors on synaptic communication mediated by gap junctions. Neuroscience 456:85–94.

Cárdenas-García SP, Ijaz S, Pereda AE (2024) The components of an electrical synapse as revealed by expansion microscopy of a single synaptic contact. eLife 13:e91931.

Cheong HSJ, et al. (2025) Transforming descending input into motor output: an analysis of the Drosophila Male Adult Nerve Cord connectome. :2023.06.07.543976 Available at: https://www.biorxiv.org/content/10.1101/2023.06.07.543976v3 [Accessed July 13, 2026].

Collins MO, Husi H, Yu L, Brandon JM, Anderson CNG, Blackstock WP, Choudhary JS, Grant SGN (2006) Molecular characterization and comparison of the components and multiprotein complexes in the postsynaptic proteome. J Neurochem 97:16–23.

Connors BW (2017) Synchrony and so much more: diverse roles for electrical synapses in neural circuits. Dev Neurobiol 77:610–624.

Connors BW, Long MA (2004) Electrical synapses in the mammalian brain. Annu Rev Neurosci 27:393–418.

Cook SJ, et al. (2019) Whole-animal connectomes of both Caenorhabditis elegans sexes. Nature 571:63–71.

Cunningham MO, Roopun A, Schofield IS, Whittaker RG, Duncan R, Russell A, Jenkins A, Nicholson C, Whittington MA, Traub RD (2012) Glissandi: transient fast electrocorticographic oscillations of steadily increasing frequency, explained by temporally increasing gap junction conductance. Epilepsia 53:1205–1214.

Dieterich DC, Kreutz MR (2016) Proteomics of the synapse – a quantitative approach to neuronal plasticity *. Mol Cell Proteomics 15:368–381.

Feng K, Sen R, Minegishi R, Dübbert M, Bockemühl T, Büschges A, Dickson BJ (2020) Distributed control of motor circuits for backward walking in Drosophila. Nat Commun 11:6166.

Flores CE, Li X, Bennett MVL, Nagy JI, Pereda AE (2008) Interaction between connexin35 and zonula occludens-1 and its potential role in the regulation of electrical synapses. Proc Natl Acad Sci U S A 105:12545–12550.

Flores CE, Nannapaneni S, Davidson KGV, Yasumura T, Bennett MVL, Rash JE, Pereda AE (2012) Trafficking of gap junction channels at a vertebrate electrical synapse in vivo. Proc Natl Acad Sci U S A 109:E573–E582.

Furshpan EJ (1964) "Electrical transmission" at an excitatory synapse in a vertebrate brain. Science 144:878–880.

Furshpan EJ, Potter DD (1959) Transmission at the giant motor synapses of the crayfish. J Physiol 145:289–325.

Galarreta M, Hestrin S (2001) Electrical synapses between Gaba-releasing interneurons. Nat Rev Neurosci 2:425–433.

Goodenough DA, Paul DL (2009) Gap junctions. Cold Spring Harb Perspect Biol 1:a002576.

Hamzei-Sichani F, et al. (2007) Gap junctions on hippocampal mossy fiber axons demonstrated by thin-section electron microscopy and freeze–fracture replica immunogold labeling. Proc Natl Acad Sci U S A 104:12548–12553.

Hao YA, et al. (2024) A fast and responsive voltage indicator with enhanced sensitivity for unitary synaptic events. Neuron 112:3680–3696.e8.

Hasegawa DK, Turnbull MW (2014) Recent findings in evolution and function of insect innexins. FEBS Lett 588:1403–1410.

Hou G, Smith AG, Zhang Z-W (2016) Lack of intrinsic GABAergic connections in the thalamic reticular nucleus of the mouse. J Neurosci 36:7246–7252.

Israel S, Rozenfeld E, Weber D, Huetteroth W, Parnas M (2022) Olfactory stimuli and moonwalker SEZ neurons can drive backward locomotion in Drosophila. Curr Biol 32:1131–1149.e7.

Ixmatlahua DJ, Vizcarra B, Gómez-Lira G, Romero-Maldonado I, Ortiz F, Rojas-Piloni G, Gutiérrez R (2020) Neuronal glutamatergic network electrically wired with silent but activatable gap junctions. J Neurosci 40:4661–4672.

Jarrell TA, Wang Y, Bloniarz AE, Brittin CA, Xu M, Thomson JN, Albertson DG, Hall DH, Emmons SW (2012) The connectome of a decision-making neural network. Science 337:437–444.

Jenkins VK, Larkin A, Thurmond J (2022) Using FlyBase: a database of Drosophila genes and genetics. In: *Drosophila: methods and protocols* (Dahmann C, ed.), pp 1–34. New York, NY: Springer US.

Kimmel CB, Sessions SK, Kimmel RJ (1981) Morphogenesis and synaptogenesis of the zebrafish Mauthner neuron. J Comp Neurol 198:101–120.

Kosaka T, Hama K (1985) Gap junctions between non-pyramidal cell dendrites in the rat hippocampus (CA1 and CA3 regions): a combined Golgi-electron microscopy study. J Comp Neurol 231:150–161.

Kunceviciene E, Sriubiene M, Liutkeviciene R, Miceikiene IT, Smalinskiene A (2018) Heritability of myopia and its relation with GDJ2 and RASGRF1 genes in Lithuania. BMC Ophthalmol 18:124.

Laird DW (2006) Life cycle of connexins in health and disease. Biochem J 394:527–543.

Lasseigne AM, Echeverry FA, Ijaz S, Michel JC, Martin EA, Marsh AJ, Trujillo E, Marsden KC, Pereda AE, Miller AC (2021) Electrical synaptic transmission requires a postsynaptic scaffolding protein. eLife 10:e66898.

Lauks J, Klemmer P, Farzana F, Karupothula R, Zalm R, Cooke NE, Li KW, Smit AB, Toonen R, Verhage M (2012) Synapse associated protein 102 (SAP102) binds the C-terminal part of the scaffolding protein neurobeachin. PLoS One 7:e39420.

Lee K, Doe CQ (2021) A locomotor neural circuit persists and functions similarly in larvae and adult Drosophila. eLife 10:e69767.

Li Y, et al. (2020) Distinct subnetworks of the thalamic reticular nucleus. Nature 583:819–824.

Loewenstein WR, Kanno Y (1964) Studies on an epithelial (gland) cell junction. J Cell Biol 22:565–586.

Marc RE, Sigulinsky CL, Pfeiffer RL, Emrich D, Anderson JR, Jones BW (2018) Heterocellular coupling between amacrine cells and ganglion cells. Front Neural Circuits 12:90.

Marin-Burgin A, Eisenhart FJ, Kristan WB, French KA (2006) Embryonic electrical connections appear to prefigure a behavioral circuit in the leech CNS. J Comp Physiol A 192:123–133.

Marsh AJ, Michel JC, Adke AP, Heckman EL, Miller AC (2017) Asymmetry of an intracellular scaffold at vertebrate electrical synapses. Curr Biol 27:3561–3567.e4.

Martin EA, Michel JC, Kissinger JS, Echeverry FA, Lin Y-P, O'Brien J, Pereda AE, Miller AC (2023) Neurobeachin controls the asymmetric subcellular distribution of electrical synapse proteins. Curr Biol 33:2063–2074.e4.

Martinez-Garcia RI, Voelcker B, Zaltsman JB, Patrick SL, Stevens TR, Connors BW, Cruikshank SJ (2020) Two dynamically distinct circuits drive inhibition in the sensory thalamus. Nature 583:813–818.

Meng L, Chen C, Yan D (2016) Regulation of gap junction dynamics by UNC-44/ankyrin and UNC-33/CRMP through VAB-8 in C. elegans neurons. PLoS Genet 12:e1005948.

Meng L, Yan D (2020) NLR-1/CASPR anchors F-actin to promote gap junction formation. Dev Cell 55:574–587.e3.
Meşe G, Richard G, White TW (2007) Gap junctions: basic structure and function. J Invest Dermatol 127:2516–2524.
Michel JC, Martin EA, Crow WE, Kissinger JS, Lukowicz-Bedford RM, Horrocks M, Branon TC, Ting AY, Miller AC (2024) Electrical synapse molecular diversity revealed by proximity-based proteomic discovery. :2024.11.22.624763 Available at: https://www.biorxiv.org/content/10.1101/2024.11.22.624763v1 [Accessed July 13, 2026].
Miller AC, Voelker LH, Shah AN, Moens CB (2015) Neurobeachin is required postsynaptically for electrical and chemical synapse formation. Curr Biol 25:16–28.
Miller AC, Whitebirch AC, Shah AN, Marsden KC, Granato M, O'Brien J, Moens CB (2017) A genetic basis for molecular asymmetry at vertebrate electrical synapses. eLife 6:e25364.
Miller AC, Pereda AE (2017) The electrical synapse – molecular complexities at the gap and beyond. Dev Neurobiol 77:562–574.
Nagy JI, Pereda AE, Rash JE (2018) Electrical synapses in mammalian CNS: past eras, present focus and future directions. Biochim Biophys Acta Biomembr 1860:102–123.
Namikawa K, Dorigo A, Zagrebelsky M, Russo G, Kirmann T, Fahr W, Dübel S, Korte M, Köster RW (2019) Modeling neurodegenerative spinocerebellar ataxia type 13 in zebrafish using a Purkinje neuron specific tunable coexpression system. J Neurosci 39:3948–3969.
Palumbos SD, Skelton R, McWhirter R, Mitchell A, Swann I, Heifner S, Von Stetina S, Miller DM (2021) cAMP controls a trafficking mechanism that maintains the neuron specificity and subcellular placement of electrical synapses. Dev Cell 56:3235–3249.e4.
Pereda AE, Bell TD, Chang BH, Czernik AJ, Nairn AC, Soderling TR, Faber DS (1998) Ca2+/calmodulin-dependent kinase II mediates simultaneous enhancement of gap-junctional conductance and glutamatergic transmission. Proc Natl Acad Sci U S A 95:13272–13277.
Pereda AE (2014) Electrical synapses and their functional interactions with chemical synapses. Nat Rev Neurosci 15:250–263.
Pereda AE, Faber DS (2011) Brain and nervous system | Physiology of the Mauthner cell: discovery and properties. In: *Encyclopedia of fish physiology* (Farrell AP, ed.), pp 66–72. San Diego: Academic Press. Available at: https://www.sciencedirect.com/science/article/pii/B9780123745538000071 [Accessed July 13, 2026].
Pereda A, Faber D (1996) Activity-dependent short-term enhancement of intercellular coupling. J Neurosci 16:983–992.
Perez AC, et al. (2017) Distinct thalamic reticular cell types differentially modulate normal and pathological cortical rhythms. Cell Rep 19:2130–2142.
Rabinowitch I, Chatzigeorgiou M, Zhao B, Treinin M, Schafer WR (2014) Rewiring neural circuits by the insertion of ectopic electrical synapses in transgenic C. elegans. Nat Commun 5:4442.
Rash JE, et al. (2013) Molecular and functional asymmetry at a vertebrate electrical synapse. Neuron 79:957–969.
Rekling JC, Shao XM, Feldman JL (2000) Electrical coupling and excitatory synaptic transmission between rhythmogenic respiratory neurons in the PreBötzinger complex. J Neurosci 20:RC113.
Robertson JD, Bodenheimer TS, Stage DE (1963) The ultrastructure of Mauthner cell synapses and nodes in goldfish brains. J Cell Biol 19:159–199.
Ryan K, Lu Z, Meinertzhagen IA (2016) The CNS connectome of a tadpole larva of Ciona intestinalis (L.) highlights sidedness in the brain of a chordate sibling. eLife 5:e16962.
Ryan TJ, Grant SGN (2009) The origin and evolution of synapses. Nat Rev Neurosci 10:701–712.
Saint-Amant L, Drapeau P (2000) Motoneuron activity patterns related to the earliest behavior of the zebrafish embryo. J Neurosci 20:3964–3972.
Sen R, Wu M, Branson K, Robie A, Rubin GM, Dickson BJ (2017) Moonwalker descending neurons mediate visually evoked retreat in *Drosophila*. Curr Biol 27:766–771.
Sen R, Wang K, Dickson BJ (2019) Two lumps ascending neurons mediate touch-evoked reversal of walking direction in *Drosophila*. Curr Biol 29:4337–4344.e5.
Shaw RM, Fay AJ, Puthenveedu MA, von Zastrow M, Jan Y-N, Jan LY (2007) Microtubule plus-end-tracking proteins target gap junctions directly from the cell interior to adherens junctions. Cell 128:547–560.
Shui Y, Liu P, Zhan H, Chen B, Wang Z-W (2020) Molecular basis of junctional current rectification at an electrical synapse. Sci Adv 6:eabb3076.
Sigulinsky CL, et al. (2020) Network architecture of gap junctional coupling among parallel processing channels in the mammalian retina. J Neurosci 40:4483–4511.
Sigulinsky CL, Pfeiffer RL, Jones BW (2024) Retinal connectomics: a review. Annu Rev Vis Sci 10:263–291.
Söhl G, Maxeiner S, Willecke K (2005) Expression and functions of neuronal gap junctions. Nat Rev Neurosci 6:191–200.
Söhl G, Joussen A, Kociok N, Willecke K (2010) Expression of connexin genes in the human retina. BMC Ophthalmol 10:27.
Song J, Ampatzis K, Björnfors ER, El Manira A (2016) Motor neurons control locomotor circuit function retrogradely via gap junctions. Nature 529:399–402.
Sprague J, Douglas E, Douglas S, Westerfield M (2001) The Zebrafish Information Network (ZFIN): a resource for genetic, genomic and developmental research. Nucleic Acids Res 29:87–90.
Starich TA, Xu J, Skerrett IM, Nicholson BJ, Shaw JE (2009) Interactions between innexins UNC-7 and UNC-9 mediate electrical synapse specificity in the Caenorhabditis elegans locomotory nervous system. Neural Dev 4:16.
Su X, et al. (2017) Neonatal CX26 removal impairs neocortical development and leads to elevated anxiety. Proc Natl Acad Sci U S A 114:3228–3233.
Takeuchi M, Yamaguchi S, Sakakibara Y, Hayashi T, Matsuda K, Hara Y, Tanegashima C, Shimizu T, Kuraku S, Hibi M (2017) Gene expression profiling of granule cells and Purkinje cells in the zebrafish cerebellum. J Comp Neurol 525:1558–1585.
Taylor SR, et al. (2021) Molecular topography of an entire nervous system. Cell 184:4329–4347.e23.
Teubner B, et al. (2000) Functional expression of the murine connexin 36 gene coding for a neuron-specific gap junctional protein. J Membr Biol 176:249–262.
Turecek J, Regehr WG (2020) Cerebellar and vestibular nuclear synapses in the inferior olive have distinct release kinetics and neurotransmitters. Elife 9:e61672.
Vats A, Sudhanand M, Bandyopadhyay A, Mascarenhas MX, Varma N, Koushika SP, Bhattacharya A (2026) The combinatorial innexin code of heterochannel electrical synapses governs synaptic function and is maintained by distinct cellular mechanisms. Proc Natl Acad Sci U S A 123:e2602614123.
Vaughn MJ, Laswick Z, Wang H, Haas JS (2024) Functionally distinct circuits are linked by heterocellular electrical synapses in the thalamic reticular nucleus. eNeuro 11:ENEURO.0269-23.2023.
Vaughn MJ, Uygun DS, Basheer R, Bender KJ, Haas JS (2026) Photomapping electrically coupled networks of the mature thalamus and cortex. bioRxiv 2025.03.06.641890.
Vaughn MJ, Haas JS (2022) On the diverse functions of electrical synapses. Front Cell Neurosci 16:910015.
Veenstra RD (1996) Size and selectivity of gap junction channels formed from different connexins. J Bioenerg Biomembr 28:327–337.
Wang Y, Belousov AB (2011) Deletion of neuronal gap junction protein connexin 36 impairs hippocampal LTP. Neurosci Lett 502:30–32.
Weber PA, Chang H-C, Spaeth KE, Nitsche JM, Nicholson BJ (2004) The permeability of gap junction channels to probes of different size is dependent on connexin composition and permeant-pore affinities. Biophys J 87:958–973.
Welsh JP, Ahn ES, Placantonakis DG (2005) Is autism due to brain desynchronization? Int J Dev Neurosci 23:253–263.
White JG, Southgate E, Thomson JN, Brenner S (1986) The structure of the nervous system of the nematode Caenorhabditis elegans. Philos Trans R Soc Lond B Biol Sci 314:1–340.
Yang X-D, Korn H, Faber DS (1990) Long-term potentiation of electrotonic coupling at mixed synapses. Nature 348:542–545.
Zolnik TA, Connors BW (2016) Electrical synapses and the development of inhibitory circuits in the thalamus. J Physiol 594:2579–2592.